Herpetological Review

Volume 39, Number 1 — March 2008

SSAR Officers (2008)

President
ROY McDIARMID
USGS Patuxent Wildlife Research Center
National Museum of Natural History
Washington, DC 20560, USA

President-elect
BRIAN CROTHER
Department of Biological Sciences
Southeastern Louisiana University
Hammond, Louisiana 70402, USA

Secretary
MARION R. PREEST
Joint Science Department
The Claremont Colleges
Claremont, California 91711, USA

Treasurer
KIRSTEN E. NICHOLSON
Department of Biology, Brooks 217
Central Michigan University
Mt. Pleasant, Michigan 48859, USA
e-mail: kirsten.nicholson@cmich.edu

Publications Secretary
BRECK BARTHOLOMEW
P.O. Box 58517
Salt Lake City, Utah 84158, USA
e-mail: ssar@herplit.com

Immediate Past President
ROBIN M. ANDREWS
Department of Biology
Virginia Polytechnic Institute
& State University
Blacksburg, Virginia 24061-0406, USA

Directors
RAFE BROWN (2008)
MEREDITH MAHONEY (2008)
JIM McGUIRE (2008)
RICHARD SHINE (2008)
PAUL CHIPPINDALE (2010)
TIFFANY DOAN (2010)
TRAVIS LaDUC (2010)
STEPHEN RICHTER (2010)

SSAR Editors

Journal of Herpetology
GEOFFREY R. SMITH, Editor
Department of Biology
Denison University
Granville, Ohio 43023, USA

Contributions to Herpetology
KRAIG ADLER, Editor
Department of Neurobiology & Behavior
Cornell University
Ithaca, New York 14853, USA

Facsimile Reprints in Herpetology
AARON M. BAUER, Editor
Department of Biology
Villanova University
Villanova, Pennsylvania 19085, USA

Herpetological Circulars
JOHN J. MORIARTY, Editor
3261 Victoria Street
Shoreview, Minnesota 55126, USA

Catalogue of American Amphibians and Reptiles
ANDREW H. PRICE, Editor
Texas Parks and Wildlife Department
Austin, Texas 78744, USA

Herpetological Conservation
ROBIN E. JUNG, Co-Editor
USGS Patuxent Wildlife Research Center
Laurel, Maryland 20708-4039, USA

JOSEPH C. MITCHELL, Co-Editor
Department of Biology
University of Richmond
Richmond, Virginia 23173, USA

HERPETOLOGICAL REVIEW

The Quarterly News-Journal of the Society for the Study of Amphibians and Reptiles

Editor
ROBERT W. HANSEN
16333 Deer Path Lane
Clovis, California 93619-9735, USA
rwh13@csufresno.edu

Managing Editor
THOMAS F. TYNING
Berkshire Community College
1350 West Street
Pittsfield, Massachusetts 01201, USA
ttyning@cc.berkshire.org

Associate Editors

ROBERT E. ESPINOZA
California State University, Northridge

CHRISTOPHER A. PHILLIPS
Illinois Natural History Survey

DEANNA H. OLSON
USDA Forestry Science Lab

ROBERT N. REED
USGS Fort Collins Science Center

MICHAEL S. GRACE
Florida Institute of Technology

R. BRENT THOMAS
Emporia State University

EMILY N. TAYLOR
California Polytechnic State University

GUNTHER KÖHLER
Forschungsinstitut und
Naturmuseum Senckenberg

MEREDITH J. MAHONEY
Illinois State Museum

Section Editors

Book Reviews
AARON M. BAUER
Department of Biology
Villanova University
Villanova, Pennsylvania 19085, USA
aaron.bauer@villanova.edu

Current Research
JOSHUA M. HALE
Department of Sciences
MuseumVictoria, GPO Box 666
Melbourne, Victoria 3001, Australia
jhale@museum.vic.gov.au

Current Research
BEN LOWE
Department of Biology
San Diego State University
San Diego, California 92182, USA
systematist@gmail.com

Geographic Distribution
ALAN M. RICHMOND
Biology Department, Morrill IV South
University of Massachusetts
611 North Pleasant Street
Amherst, Massachusetts 01003-9297, USA
alanr@bio.umass.edu

Geographic Distribution
INDRANEIL DAS
Institute of Biodiversity &
Environmental Conservation
Universiti Malaysia Sarawak
94300, Kota Samarahan, Sarawak, Malaysia
idas@ibec.unimas.my

Geographic Distribution
JERRY D. JOHNSON
Department of Biological Sciences
The University of Texas at El Paso
El Paso, Texas 79968, USA
jjohnson@utep.edu

Geographic Distribution
GUSTAVO J. SCROCCHI
Instituto de Herpetología
Fundación Miguel Lillo, Miguel Lillo 251
4000 Tucumán, Argentina
soniak@unt.edu.ar

Zoo View
JAMES B. MURPHY
Department of Herpetology
National Zoological Park
3001 Connecticut Ave., NW
Washington, D.C. 20008, USA
jbmurphy2@juno.com

Herpetological Husbandry
BRAD LOCK
Department of Herpetology
Zoo Atlanta
800 Cherokee Ave., S.E.
Atlanta, Georgia 30315, USA
block@zooatlanta.org

Natural History Notes
CHARLES W. PAINTER
New Mexico Dept. of Game & Fish
P.O. Box 25112
Santa Fe, New Mexico 87504, USA
charles.painter@state.nm.us

Natural History Notes
JAMES H. HARDING
MSU Museum
Michigan State University
East Lansing, Michigan 48824, USA
hardingj@msu.edu

Natural History Notes
ANDREW T. HOLYCROSS
School of Life Sciences
Arizona State University
Tempe, Arizona 85287-4701, USA
holycross@asu.edu

Copy Editors
BARBARA BANBURY
RAUL DIAZ
MICHAEL JORGENSEN
KYLE HESED

Natural History Notes
MARC P. HAYES
2636 59th Avenue NW
Olympia, Washington 98502-3449, USA
ranahayes@msn.com

SOCIETY FOR THE STUDY OF AMPHIBIANS AND REPTILES

www.ssarherps.org

The Society for the Study of Amphibians and Reptiles, the largest international herpetological society, is a not-for-profit organization established to advance research, conservation, and education concerning amphibians and reptiles. Founded in 1958, SSAR is widely recognized today as having the most diverse society-sponsored program of services and publications for herpetologists. Membership is open to anyone with an interest in herpetology—professionals and serious amateurs alike—who wish to join with us to advance the goals of the Society.

All members of the SSAR are entitled to vote by mail ballot for Society officers, which allows overseas members to participate in determining the Society's activities; also, many international members attend the annual meetings and serve on editorial boards and committees.

Annual Dues and Subscriptions: Annual membership dues for the year 2008 in the Society for the Study of Amphibians and Reptiles are as follows: **REGULAR** membership US$60 (Student $30)—includes *Journal of Herpetology* and *Herpetological Review*; **PLENARY** membership US$80 (Student $45)—includes *JH*, *HR*, and annual subscription to the *Catalogue of American Amphibians and Reptiles*; **INSTITUTIONAL SUBSCRIPTION** $115—includes JH and HR. Additional fee for airmail postage outside USA $35 for one year. Additional membership categories available on the SSAR webpage: http://www.ssarherps.org/pages/membership.html.

All members and institutions receive the Society's primary technical publication, the *Journal of Herpetology,* and its news-journal, *Herpetological Review*; both are published four times per year. Members also receive pre-publication discounts on other Society publications, which are advertised in *Herpetological Review*.

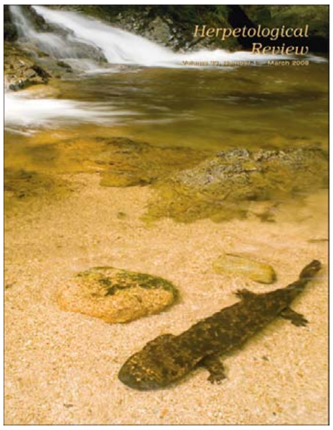
To join SSAR or to renew your membership, please visit the secure online Allen Press website:

http://timssnet.allenpress.com/ECOMSSAR/timssnet/common/tnt_frontpage.cfm

Future Annual Meetings

2008 — Montreal, Canada, 23–28 July (with ASIH, HL)
2009 — Portland, Oregon, 22–27 July (with ASIH, HL)
2010 — Providence, Rhode Island, 7–12 July (with ASIH, HL)
2011 — Minneapolis, Minnesota, 6–11 July (with ASIH, HL)

About Our Cover: *Andrias japonicus*



Clear, cold mountain streams drain the rugged mountains of Japan's Honshu, Shikoku, and Kyushu islands. At one site on Honshu Island, at an elevation > 500 m, the riverbed is rocky and water-swept. Oak and maple trees grow in the surrounding closed canopy forest along with clusters of bamboo. Cold temperatures and constant agitation of the water produce a highly oxygenated environment. Lurking beneath the waters of these clear mountain streams is the Japanese Giant Salamander, *Andrias japonicus*. *Andrias* contains two living species: *japonicus* and *davidianus* (from China). Together with the North American Hellbender (*Cryptobranchus alleganiensis*), these are the only living members of the ancient family Cryptobranchidae. This group is remarkable both for containing the largest living amphibians (the two species of *Andrias,* to ~ 1.5 m TL) and displaying a remarkable geography—two species in eastern Asia and one in eastern North America.

The breeding season for *A. japonicus* spans August to September when males compete to control breeding sites. The breeding sites consist of grottos and cubbies in the waterway that are protected enough for the male to guard eggs deposited there. Male competition for these sites can be fierce, even resulting in death. Males that retain control of the sites are referred to as "den masters." Females deposit clutches of 400–500 eggs within the dens, which the males guard until the larvae hatch and disperse. The larvae take 4 or 5 years to metamorphose into an adult form. Captive animals have lived > 50 yrs. Wild longevities are still not clearly understood.

Danté Fenolio was in Japan (with funding from the Javitch Foundation) visiting research biologist Sumio Okada (Ph.D. student at Shimane University). Okada has been monitoring several populations of *Andrias* for over 7 years with pit tags in roughly 650 individuals. This work is critical in developing an understanding of the population ecology and habitat requirements of these ancient animals. Fenolio met an elderly gentleman in one such village who has interacted with *A. japonicus* since the pre-World War II era. His descriptions of the sizes of animals he has encountered and their abundance, past and present, speaks to a slow but steady decline of the species. The salamander on our cover resides in a stream that is slated for a controversial hydroelectric dam. Public awareness about the plight of these salamanders is increasing and a television special featuring Sumio and his work recently aired on the National Geographic Channel.

Fenolio's photograph of our cover subject came about with the assistance of Sumio Okada, who led Danté to the mountain stream locality on Honshu Island where they encountered the salamander. Wildlife photographer Michael Ready shared a tip involving a flash method to create the "foggy" look in the waterfall while keeping the salamander in the foreground crisp and well lit. The photo was taken with a Konica-Minolta 5D, Minolta 24mm lens on a tripod, with a Sony twin-flash unit.

Danté Fenolio is a Ph.D. Candidate at the University of Miami and a recent hire by the Atlanta Botanical Garden for their amphibian conservation program. His research focuses on subterranean biodiversity and amphibian ecology/systematics. Mike Ready took this image of him at the Tottori Prefectural Museum, Japan, and it is used here with their permission. The preserved *Andrias japonicus* shown here was 143 cm long (TL) with a mass of 44.3 kg at the time of death.

SSAR BUSINESS

New Opportunities for Students at Montréal Meeting

There was much discussion at the 2007 St. Louis Annual Meeting regarding student involvement in SSAR and how to make the Annual Meeting more accessible to students and encourage their attendance. With this in mind, a Student Participation Committee was formed (Dawn Wilson, Chair) and plans are underway to change a number of aspects of the meeting. The 2008 Joint Meeting of Ichthyologists and Herpetologists will be held in Montréal, Canada, July 23–28. SSAR is planning two new events specifically for our student members. We will hold a lunch meeting (pizza provided) with the members of the Student Participation Committee, several members of the Board, and those students who want to take a more active role in the Society. Every year we look for students to serve on committees and editorial boards, organize events at our Annual Meeting, etc. If you have an interest in serving the Society in any capacity, the Board needs to know who you are! Additionally we are planning an evening students-only social immediately prior to the opening general reception (finger food plus drink tickets provided). Board members and a number of senior herpetologists will be in attendance and there will be ample opportunities to meet and talk with younger members of the Society.

We wish to issue a reminder that the Board Meeting (which will be held on July 22) is open to all members of the Society and everyone is encouraged to attend. Because we know that most members are not usually able to arrive a day early, we will be changing the format of the Business Meeting (usually held on the 4th day of the meeting) so that it will now more closely resemble a Board Meeting. Officers and Editors will be in attendance and will each present a brief Annual Report and a summary of upcoming plans. Audience participation will be expected.

We hope that these changes will encourage all of our members, especially students, to continue to support our Society and attend our Annual Meeting. SSAR is proud of its student roots and recognizes that the future of our organization rests on continued student involvement.

The Student Participation Committee welcomes feedback or suggestions from any interested member (e-mail: dwilson@amnh.org). We hope to see many of you in Montréal.

—Marion Preest, SSAR Secretary

SSAR Henri Seibert Awards for 2008

The Henri Seibert Awards were initiated in 1992 to provide recognition for the best student papers presented at the annual meeting of the SSAR. To be eligible, the presented paper must be the result of research by the individual making the presentation. The research must have been conducted while the student was enrolled in either an undergraduate or graduate degree program. Please refer to *Herpetological Review* 28(4):175 and the SSAR website (http://www.ssarherps.org/pages/seibert.php) for recommendations to students entering the Henri Seibert Competition. Students entering the competition must be members of SSAR. The presentations will be judged by the SSAR student prize committee. One Henri Seibert Award of US $200 may be given in each of the following **four** categories: **Systematics/Evolution**, **Ecology**, **Physiology/Morphology**, and **Conservation**. Students may win the award only one time. Please indicate the appropriate category for which you are applying on the abstract submission form. Announcement of winners will be made at the **SSAR Business Meeting**. All participants should be present at the business meeting. Contact Patrick Owen for further information: owen.133@osu.edu.

6th World Congress of Herpetology
Manaus, Amazonas, Brazil

We are preparing for a bumper crop of herpetologists from throughout the world to join us for the event of the millenia—the 6th World Congress of Herpetology, to be held jointly with SSAR, in Manaus, Amazonas, Brazil 17–22 August 2008. This will be first World Congress in the New World and it will be done with all of the flare and pomp of the First World Congress, with the added signature touch that people have appreciated in the SSAR-HL congresses I ran in Veracruz in 1987 and Manaus 2003. I will not try to outdo these, but come and see if I do! Remember my motto—no tickets, no lines, no limits!

Please see our website (www.worldcongressofherpetology.org). It is a dynamic site; as events and offers are solidifying they are being placed on the web, so visit us often.

It is our pleasure to host this meeting on the shores of the Rio Negro, in the heart of the Amazon Rainforest at the five-star Tropical Hotel. Since you are coming all this way, take some time before or after the meeting to go out and see the forest or take a river trip. Set up your own groups to go on river trips, many of the plenary speakers and symposium organizers will be accompanying post-meeting river trips, so you will have days to talk with some of the greats in herpetology on a one to one basis. Contact Vic Hutchison for his special Varig Airline Affiliated post meeting birding tours to the patanal.

Field trips along jungle rivers can be programmed aboard boats like our boat the *Enigma*, allowing total comfort and safety while fishing or observing wildlife.

On Sunday afternoon there will be a special session on "Amphibian Conservation: Moving from Global to Regional Planning" organized by Don Church of Conservation International, with an interval for the first plenary lecture, followed by a dinner and awards session for the invited members of the Amphibian Conservation session.

Each session of the meeting, from Monday–Friday, will begin with lectures by plenary speakers, followed by either symposia or contributed papers. Access to posters will be available all day, culminating with a happy hour session at the end of each afternoon when the poster authors will be available for discussions. There will be the traditional Monday night social icebreaker with regionally produced beverages, finger food, and regional dance show. Tuesday evening will be a special event—the Secretary General of the World Congress birthday party. Wednesday night will be a picnic (Churrasco) on the white sand beach of the Negro River complete with a Boi Boom Ba live dance show. Thursday night will be the traditional turtle group "lunch" as well as other optional choices such as trips to the Opera House, music and dance houses such as Remulos. Friday evening will be the final plenary presentation by Ian Swingland during the Banquet, followed by the AIHA sponsored live auction, and discotec until dawn.

Plenary Speakers

We have gathered a prestigious group of plenary speakers, known not only for the excellence of their science but also for their communication and entertainment skills. There will be no heads nodding in the aisles when these herpetologists are performing!

—William E. Magnusson, INPA. The evolution of herpetological studies in the Amazon. Sunday evening welcome address.
—Russell Mittermeier, President, Conservation International. Global priorities in reptile and amphibian Conservation. Keynote speaker.
—Darrel Frost, American Museum of Natural History, E.D. Cope Honorary Speaker. The past, present, and future of the science of amphibian systematics.
—Peter Narins, UCLA. Exploring frog communication behavior: continuing adventures of an expeditionary biologist.

—Michael Bull, Flinders University, Australia. Lizard social behavior and conservation: lessons from an Australian skink.
—Richard Shine, University of Sydney. Invasion biology and impact of cane toads in Australia—the way it really is!
—John M. Legler, Professor Emeritus, University of Utah. Turtles: a paradigm of versatility and venerability.
—Fred Allendorf, University of Montana. Genetic effects of habitat fragmentation.
—Jonathan Losos, Harvard University. Evolutionary diversification on islands and mainlands: historical contingency and adaptive radiation in *Anolis* lizards.
—Aaron Bauer, Villanova University. Spatial and temporal history of geckos of the world.
—Ian Swingland, Director of the First World Congress of Herpetology, from his world base in Kent. Peeling peaches in the pampas.
—James Parham, California Academy of Sciences. Human impact on the diversity, distribution, and systematics of turtles.
—Oscar Flores-Villela, UNAM. The making of a national herpetofauna, the herpetofauna of Mexico.
—Marvalee Wake, University of California, Berkeley. Evolution of amphibian viviparity and the origins of homoplasies.
—Celio Haddad, Universidade Estadual Paulista Júlio de Mesquita Filho (UNESP, Campus de Rio Claro). Reproductive behavior in anuran amphibians.

FEATURED SYMPOSIA

—Reproduction in Reptiles: From Genes to Ecology. Michael B. Thompson & Scott L. Parker, organizers.

—Invasive Reptiles and Amphibians: Global Perspectives and local solutions. Robert Reed, organizer.

—Sensory Ecology of Anuran Communication. T. Ulmar Grafe & Peter M. Narins, organizers.

—Evolutionary Transitions of Body Shape in Extant Amphibians and Reptiles: Call for Integrative Approaches. Tiana Kohlsdorf, organizer.

—Herpetofaunal Reintroductions, Translocations, and Supplementations. Jennifer M. Germano & Phillip J. Bishop, organizers.

—Disease and Amphibian Declines—Where Do We Go from Here? Jean-Marc Hero, Erin Muths, & Trenton W. J. Garner, organizers.

—The Biology and Management of Crocodilians. John Thorbjarnarson & Kent Vliet, organizers.

—Herpetological Conservation & Biology. Robert Brodman, Robert Murphy & Ben Bell, organizers.

—Biogeography of the South and South East Asian Herpetofauna. David J. Gower, organizer.

—Dendrobatoid Frog Biology. Taran Grant & Stefan Lötters, organizers.

—Ecophysiology of Reptiles. Denis Otávio Vieira de Andrade & Glenn Tattersall, organizers.

—Sequestered Defensive Compounds in Tetrapod Vertebrates and Defensive Mechanisms of Amphibians and Reptiles: Actual Knowledge and Future Perspectives. Alan H. Savitzky, Akira Mori, Deborah A. Hutchinson, & Luis Felipe Toledo, organizers.

—Status of South American Freshwater Turtles. Brian Bock, organizer.

—Development and Implementation of Recovery Programs for Endangered Chelonians. Rick Hudson, organizer.

—Conservation Genetics of Amphibians and Reptiles in the 21st Century. Robert Jehle & Trenton W. J. Garner, organizers.

All activities of the 6th World Congress of Herpetology 2008 will be held at the Hotel Tropical Conference Center, located 16 km from downtown Manaus, and 10 km from the airport, in a ritzy party area of Manaus known as Ponta Negra. The Ponta Negra Beach stretches along the high banks of the Negro River, which, at this point, enlarges to a width of 8 km. Outside the hotel complex an elevated riverside promenade with numerous bars and small restaurants provides an agreeable alternative for a late afternoon stroll in a safe atmosphere with local live music and live locals as well. You can share a privileged sight of the sunset over the Negro River.

There is a huge outdoor amphitheater where musical groups (boi, Manaus Philharmonic Orchestra, samba, pagode, etc.) perform free on Sunday afternoons and other special occasions. Because we are a special occasion, I am sure something will be happening there during your stay with us. It is too early to predict the program, but surely something lively will be happening.

The Hotel Tropical has 588 luxurious air-conditioned rooms with cable TV, minibar, telephone, and room service. The hotel has two swimming pools (one with a waterfall), a mini-shopping center, discotec (which will be open most nights), four restaurant-bars and two additional bars. The aviary, which used to occupy much of the lobby, has been transformed into a wonderful piano bar, a small zoo displaying some species of the local fauna, a botanical garden and orchid house, as well as access to the beach.

Several years ago, while on the beach at low water, a young girl walked by carrying a fresh meter-long anaconda. Frogs call from the trees, geckos scamper up walls. It is a huge, sprawling hotel with surrounding gardens and forest areas overlooking the Rio Negro! There is a security wall around the grounds, and security

Part of the local committee aboard the Enigma to welcome all herpetologists from around the world to the 6th WCH in Manaus.

guards at the access gates, so personal safety is guaranteed and the general public does not stroll onto the grounds by any means. So it is a safe place to turn your families loose. I personally recommend all people who do not speak Portuguese to stay at this hotel. A large percentage of the wonderfully friendly hotel staff speaks English.

The food is tremendous and you are not going to waste any time traveling back and forth to the paper sessions on buses or taxis. The hotel puts out a magnificent buffet breakfast with local fruits, juices, specialty Amazonian dishes, as well as standard eggs, cereals, meats, and dairy products. As the breakfast at all of the hotels comes with the room we will NOT have second breakfast being served at coffee breaks. Coffee breaks will be high on caffeine and low on carbohydrates and calories, but do not worry as there will be munchies!

Salads,vegetables, and pasta can be found in the menus offered at the hotel restaurants, as well as native fish, seafood, beef, and fowl, all in buffet style so it is not necessary to worry about something special for your palate or diet. Most of the fruit juices served are natural and the fruit collected locally. So if you want to eat only veggies or all red meat or only nibble a bit you are your own dietician and can fill and refill your plate to your taste, I would not try to lose weight while you are here. Remember, we are in the heart of the Amazon—not on the coast, so we will have a tremendous variety of local fishes prepared in traditional recipes to experiment with. Seafood is imported from the coast, expensive, frozen, and not to be expected as part of the local cuisine. We will have some surprisingly exotic food to sample both in the daily buffets as well as in the evening socials, anti-socials, picnics, and banquets.

There is a boat dock, helicopter pad, and pontoon plane hangar at the hotel with service to the surrounding jungle lodges. Most of these jungle lodges give daily boat taxi service to Hotel Tropical. The paper sessions are in the hotel complex. Most if not all of the picnics, banquets, social activities, dance presentations, etc., will be held at the hotel, or close by, so you will use your time much more efficiently and your stay will be much less hectic if you stay here.

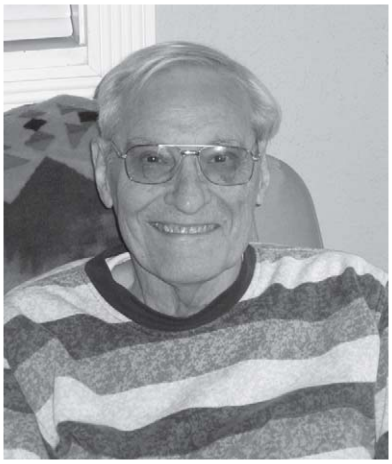
All other hotels are NOT within walking, running, or swimming distance from Hotel Tropical. If you prefer feeling the culture of Manaus, by all means stay at one of the smaller hotels downtown, but you will waste at least two hours a day doing so. Morning and evening shuttle buses will probably be provided, but you can also ride taxis back and forth, usually 4 people can ride for the price of the fare ($12US). There are inexpensive hotels downtown as well as youth hostels for people on a tight budget and we will run free shuttle buses to and from Hotel Tropical to downtown. Tap water is safe to drink, there is not a malaria problem at Hotel Tropical, the beer is cold, and the environment exciting to say the least. See our web site for registration material and hotel/airfare packages: www.Worldcongressofherpetology.org

See you in Manaus!

Richard C. Vogt, Director
Sixth World Congress of Herpetology-AIHA-SSAR Joint Meeting

NEWSNOTES

Herbert C. Dessauer Honored by LSU



On September 6, 2007 the Foundation for Louisiana State University Health Sciences Center added several positions to the Louisiana Board of Regents Endowed Professorship Program. Among these is the "Herbert C. Dessauer Professorship in Graduate Studies." Readers of HR will recognize Herb as a pioneer in the development of molecular systematics and the use of molecular techniques to analyze hybridization and its consequences. Herb was also the first graduate student to receive the Ph.D. from the LSU Health Sciences Center, in New Orleans, in 1952.

University of Illinois Museum of Natural History

The Illinois Natural History Survey (INHS) is pleased to announce that we are once again filling loan requests for the University of Illinois Museum of Natural History (UIMNH) Amphibian and Reptile Collection. This exceptional collection was moved to storage in 2002 because of space re-assignment by the University of Illinois. The INHS has just finished moving the entire collection to our facilities. Direct all inquiries to the curator, Chris Phillips, at chrisp@inhs.uiuc.edu. The collection is computerized and can be searched at the URL below. This search engine will return limited location data (county, state & country). Specific location information can be obtained by contacting the curator.

<www.inhs.uiuc.edu/cbd/collections/AmphReptColln/herp_links/uiherps.html>

Summer Field Course in Conservation Biology, with an Emphasis on Amphibians and Reptiles

A field course in Conservation Biology will be offered at the Lakeside Laboratory from 2–27 June 2008. Lakeside Lab is located in northwestern Iowa, along the intersection, from east to west, between the Eastern Deciduous Forest and Great Plains, and along the intersection, from north to south, between the recently glaciated Lakes Region and the older, better drained, and more variable stream systems associated with Missouri and Mississippi River uplands.

A combination laboratory and field course, Conservation Biology examines the history of the Upper Midwest from the retreat of the latest glaciers to the present day. Northwest Iowa is a landscape of lakes, wetlands, prairie, and oak savannah; it is also a place of intense agriculture, an area of concentrated summer tourism, and it hosts a wind farm. In this context, native ecosystems will be compared against altered ecosystems, and the processes of restoration will be measured against these extremes. Students will participate in a habitat restoration and should bring rugged clothing, sturdy boots, heavy canvas gloves, and a hard hat.

The herpetofauna of this area is well known. Highlights include the turn-of the-century (19th to 20th) commercial collections of 20 million leopard frogs/yr, the pioneering surveys of Frank Blanchard in the 1920s, and more recent findings that bear on the global problems of amphibian malformations and declines. We will visit the only known Prairie Rattlesnake populations in Iowa.

For information about Lakeside Lab see:

http://www.continuetolearn.uiowa.edu/lakesidelab/

For more information about the course, contact: Michael J. Lannoo, Indiana University School of Medicine - TH Holmstedt Hall, Rm 135, ISU Terre Haute, Indiana 47809, USA; e-mail: mlannoo@iupui.edu.

MEETINGS

Meetings Calendar

Meeting announcement information should be sent directly to the Editor (rwh13@csufresno.edu) well in advance of the event.

10–12 April 2008—55th Annual Meeting of the Southwestern Association of Naturalists, University of Memphis, Tennessee, USA. Information: http://www.biosurvey.ou.edu/swan/

11–13 April 2008—Fourth Annual Sonoran Desert Herpetological Symposium, Tucson, Arizona, USA. Information: http://www.arts.arizona.edu/herp/

5–8 June 2008—Joint Meeting of Texas Herpetological Society, SW PARC, Horned Lizard Conservation Society; University of Texas, Austin, Texas, USA. Information: gad.perry@ttu.edu

18–21 June 2008—32nd International Herpetological Symposium; Nashville, Tennessee, USA. Information: www.kingsnake.com/ihs/

23–27 June 2008—6th Symposium on Lacertids of the Mediterranean Basin, Lesvos Island, Greece. Information: http://www.elerpe.org/

23–28 July 2008—51st Annual Meeting, Society for the Study of Amphibians and Reptiles; 88th Annual Meeting, American Society of Ichthyologists and Herpetologists; 66th Annual Meeting, The Herpetologists' League. Montreal, Quebec, Canada. Information: http://www.dce.ksu.edu/jointmeeting/

17–20 August 2008—6th World Congress of Herpetology, Manaus, Brazil (meeting jointly with SSAR). Information: http://www.worldcongressofherpetology.org/index.php?section=51

24–29 November 2008—VIII Latin-American Congress of Herpetology (VIII Congreso Latinoamericano de Herpetologia), Topes de Collantes, Sancti Spiritus, Cuba. Information: Roberto Alonso Bosch (e-mail: 8voclah@fbio.uh.cu or ralonso@ecologia.cu).

CURRENT RESEARCH

The purpose of **Current Research** is to present brief summaries and citations for selected papers from journals other than those published by the American Society of Ichthyologists and Herpetologists, The Herpetologists' League, and the Society for the Study of Amphibians and Reptiles. Limited space prohibits comprehensive coverage of the literature, but an effort will be made to cover a variety of taxa and topics. To ensure that the coverage is as broad and current as possible, authors are invited to send reprints to the Current Research section editors, **Joshua Hale** or **Ben Lowe**; postal and e-mail addresses may be found on the inside front cover.

The current contents of various herpetological journals and other publications can be found at: http://www.herplit.com/contents.

Impacts of Invasion Stress on Invasive Cane Toads

The process of invading a novel environment may introduce new stresses to invasive species themselves, but these impacts have received little attention. While studying a Cane Toad (*Chaunus* [*Bufo*] *marinus*) invasion in the Northern Territory, Australia, the authors noticed frequent abnormalities in the spinal joints of toads at the invasion front. To determine whether the traits that facilitate successful invasion (e.g., larger body size and greater distance moved per day) are related to spinal arthritis, they examined toads from three invasive populations and established that arthritis only occurs in larger toads, and that the frequency of the condition in a population is correlated with the population's mean movement rate. Using pairs of size- and sex-matched toads that differed by spinal condition, both telemetry of free-ranging toads and locomotor performance tests showed that arthritic toads move as far and as fast as non-arthritic conspecifics. Bacterial cultures from arthritic and normal spinal joints showed that arthritic joints are infected by *Ochrobactrum anthropi*, a common soil bacterium. The authors conclude that the invasion process, which selects for larger body size and longer limb length (making animals more susceptible to degenerative disease), and a highly active lifestyle of almost constant movement (causing even greater stress on the spine), have rendered the cane toad susceptible to joint bacterial infection and spinal arthritis.

Brown, G. P., C. Shilton, B. L. Phillips, and R. Shine. 2007. Invasion, stress, and spinal arthritis in cane toads. Proceedings of the National Academy of Science 104:17698–17700.

Correspondence to: Richard Shine, School of Biological Science, University of Sydney, Sydney NSW 2006, Australia; and Department of Primary Industries, Fisheries, and Mines, Berrimah Veterinary Laboratories, Darwin NT 0801, Australia; e-mail: rics@bio.usyd.edu.au.

Scent Marks Indicate Competitive Ability in Lizards

Scent marks are typically thought to be signals of an individual's competitive ability, even in that individual's absence from its territory. However, in addition to conveying information about a potential competitor in an area, these marks may also signal the resource availability of the area. To determine the role of male scent marks in intrasexual communication in the Iberian Wall Lizard (*Podarcis hispanica*), and whether these roles change across the reproductive season, the authors captured lizards from near Valencia, Spain in the early (March) and late (June) breeding season. Ten males from each season were exposed to four experimental tests: scent marks from a stimulus male similar in size, scent marks from a larger male, scent marks from a smaller male, and no scent (control). These trials showed that early (but not late) in the breeding season, males were attracted to areas scent marked by males of similar or larger size (as measured by time spent in marked area) but not to areas marked by smaller males. These results indicate that *P. hispanica* males can obtain information on rival male size, shown to be an honest indicator of dominance in this species, from scent marks. Because males spent more time in areas marked by presumably dominant rivals, the authors suggest that scent marks may function as indirect measures of territory resource quality, assuming that dominant males defend superior territories. They also discuss potential explanations of the seasonal change in male response to scent marks.

Carazo, P., E. Font, and E. Desfilis. 2007. Chemosensory assessment of rival competitive ability and scent-mark function in a lizard, *Podarcis hispanica*. Animal Behaviour 74: 895–902.

Correspondence to: P. Carazo, Instituto Cavanilles de Biodiversidad y Biología Evolutiva, Universidad de Valencia, Apdo. 22085, 46071 Valencia, Spain; e-mail: pau.carazo@uv.es.

Discrimination of Sea Turtle Artifacts Using DRIFT Spectroscopy

Sea turtle (Family Cheloniidae) scutes have been widely used in the manufacture of historic artifacts such as jewelry and furniture. Keratins from these scutes closely resemble those from bovid horn sheaths, and it is of great importance to museum curators and law enforcement officers to correctly identify the origin of artifacts, particularly given the endangered status of all sea turtle species. The authors investigated the utility of diffuse reflectance infrared Fourier transform (DRIFT) spectroscopy to distinguish between sea turtle and bovid keratin. Using 35 bovid samples from 24 species and 24 sea turtle samples from 4 species, they found that examinations using only raw DRIFT spectra may result in spurious taxonomic assignment, but analyzing these data using discriminant analysis provided reliable identification of all standards used in this study. This technique can also be used to differentiate protein-based plastics (i.e., casein) from sea turtle carapace. The authors conclude that DRIFT spectroscopy combined with discriminant analysis provides a rapid, non-destructive method of distinguishing sea turtle keratin in artifacts and may be a powerful tool in the modern wildlife trade.

Espinoza, E. O., B. W. Baker, and C. A. Berry. 2007. The analysis of sea turtle and bovid keratin artifacts using drift spectroscopy and discriminant analysis. Archaeometry 49: 685–698.

Correspondence to: E. O. Espinoza, US National Fish & Wildlife Forensics Laboratory, 1490 E. Main St., Ashland, Oregon 97520, USA; e-mail: ed_espinoza@fws.gov.

Underestimation of Tropical Frog Diversity

Amphibians are rapidly declining, yet the number of amphibian species remains underestimated. Together, these observations reveal a pressing need for rapid evaluation of species richness. The authors used a combination of three methods—isolation by distance, phylogenetic analyses (to determine prevalence of paraphyletic species), and comparison of molecular distances (to determine patterns of intra- and interspecific distances)—to analyze previously published and new 16S mtDNA sequence data from 60 species of French Guianese frogs with the goal of estimating the number of undescribed species in this region. With these techniques, they calculated that the "true" number of species relative to the 60 study species is underestimated by 22–115%. They found that although geographically widespread species exist, most geographically distant populations could be considered new species, and that the many instances of paraphyletic species indicate the presence of undescribed cryptic species. From their results, the authors propose that a pairwise genetic distance of 0.03 is appropriate for distinguishing species in this group, and with this approach they identified 129 candidate species. This suggests that amphibian decline might be even more dramatic than previously thought, as both identified and unidentified species are being lost.

Fouquet, A., A. Gilles, M. Vences, C. Marty, M. Blanc, and N. J. Gemmell. 2007. Underestimation of species richness in neotropical frogs revealed by mtDNA analyses. PLoS ONE 2: e1109.

Correspondence to: Antoine Fouquet, Molecular Ecology Laboratory, School of Biological Sciences, University of Canterbury, Christchurch, New Zealand; and EA 3781 EGEE Evolution Genome Environment, Université de Provence, Centre St Charles, Marseille, France; e-mail: afo23@student.canterbury.ac.nz.

Hormones and Performance Capacity in Male Lizards

Males of many species advertise their quality with elaborate sexual signals, but the mechanism that links male signals and their fighting ability remains unknown. To test the hypothesis that signal size and fighting ability are related through circulating hormone levels, the authors examined adult male Green Anole lizards (*Anolis carolinensis*), a species in which dewlap (throat fan) size has been shown to predict bite force performance. They captured males during (N = 82) and after (N = 26) the breeding season in Louisiana, USA and measured their morphology, bite force, and concentrations of testosterone and corticosterone in blood. Males were classified into two previously described (presumably age-related) size classes: heavyweights and lightweights. Comparisons of the size classes revealed that heavyweights had higher testosterone levels in the breeding season than lightweights, but corticosterone levels did not differ between the groups. Also, for lightweights, but not heavyweights, testosterone levels were correlated with dewlap size and bite force, but this relationship is due to the influence of body size on all three traits. The authors conclude that these results suggest complex relationships among testosterone, morphology, and performance. They propose that smaller males suppress testosterone levels until they reach a competitive size, when higher testosterone then increases bite force and dewlap size.

Husak, J. F., D. J. Irschick, J. J. Meyers, S. P. Lailvaux, and I. T. Moore. 2007. Hormones, sexual signals, and performance of green anole lizards (*Anolis cristatellus*). Hormones and Behavior 52:360–367.

Correspondence to: Jerry F. Husak, Department of Biology, University of Massachusetts, Amherst, Massachusetts 01003, USA; e-mail: husak@bio.umass.edu.

Diversification and Convergence in Desert Night Lizards

Species in the night lizard genus *Xantusia* are characterized as having high site philopatry and specific habitat requirements (e.g., individuals may spend their lives under a single log), and previous work has shown that the group has an evolutionary history of deep divergences. To test species delimitations and to compare morphological and mitochondrial assessments of phylogenetic relationships, the authors used sequence data from the mitochondrial cytochrome *b* gene (1143 bp) of 508 lizards in the *X. vigilis* species complex from 156 localities. For subsets of these lizards, they also sequenced a second mitochondrial region (ND2) and two nuclear introns (α-enolase and GAPD). Using standard Bayesian, maximum parsimony, and maximum likelihood techniques with mtDNA data, they propose a novel phylogenetic hypothesis for the *X. vigilis* group, which is largely supported by the nuclear data, and which suggests that the recognized taxonomy for this group underestimates species diversity. Their results support the conclusion that multiple species have converged on a rock-crevice-dwelling morphology, and they infer a deep history of cryptic speciation. The authors propose that vicariance in the late Miocene drove the diversification of *Xantusia*, while climate change in the Pleistocene determined their distribution.

Leavitt, D. H., R. L. Bezy, K. A. Crandall, and J. W. Sites, Jr. 2007. Multi-locus DNA sequence data reveal a history of deep cryptic vicariance and habitat-driven convergence in the desert night lizard *Xantusia vigilis* species complex (Squamata: Xantusiidae). Molecular Ecology 16:4455–4481.

Correspondence to: Dean H. Leavitt, Department of Biology, San Diego State University, 5500 Campanile Drive, San Diego, California 92182-4614, USA; e-mail: dleavitt@rohan.sdsu.edu.

Environment Influences Developmental Trade-offs in Frogs

Fitness tradeoffs between size at and time to metamorphosis have been well-studied in amphibians and other taxa, and larval environmental conditions have been shown to influence these traits. However, multiple mechanisms may drive these tradeoffs. The authors tested the hypothesis that an individual's allocation of resources between growth and development in somatic versus reproductive structures may be driven by environmental conditions, a mechanistic explanation that has received little attention. To this end, they performed two experiments in which *Xenopus laevis* (African Clawed Frog) tadpoles were exposed to varying diets. In the first experiment, 5 groups of 5 tadpoles were reared communally in high or low food treatments, and one group per treatment were weighed and fixed for gonadal size measurement at approximately two week intervals. Results showed that food restriction reduced both somatic and gonadal growth rates, but not developmental rates. In the second experiment, 10 tadpoles were raised individually to metamorphosis on each of three food treatments (low, medium, and high), at which point they were weighed and fixed. These results showed that individuals that experienced food restriction had reduced somatic growth and development and smaller gonads, but the developmental stage of the male testes at metamorphosis was not affected. Together, these results suggest that the lower fecundity associated with small body size at metamorphosis may be at least partially due to gonadal size at metamorphosis, and not just reduced rates of somatic development. As habitats are continually degraded, the influence of environmental conditions on gonadal growth may be an important factor in population regulation.

McCoy, K. A., M. W. McCoy, A. Amick, L. J. Guillette Jr., and C. M. St. Mary. 2007. Tradeoffs between somatic and gonadal investments during development in the African clawed frog (*Xenopus laevis*). Journal of Experimental Zoology 307A:637–646.

Correspondence to: Krista A. McCoy, Department of Zoology, University of Florida, P. O. Box 118525, Gainesville, Florida 32611-8525, USA; e-mail: kristam@zoo.ufl.edu.

A Developmental Staging Series for *Anolis* Lizards

As the field of evolutionary developmental biology ("evo-devo") has emerged, the need for new model organisms for the study of morphological evolution has grown. Although there is no typical model squamate for developmental studies, lizards in the speciose genus *Anolis* have become a model system in ecology and evolution and may be useful in developmental studies as well. The relationship between morphology and ecology is well-known in this group, as many Caribbean species have independently evolved distinct morphological adaptations that allow them to exploit particular microhabitats. The authors propose that *Anolis* offer the opportunity to integrate evolutionary and developmental studies, and they present a generalized developmental staging series for the genus. They examined approximately 1000 embryos of 8 *Anolis* species, and determined *A. sagrei* to be the most appropriate model for the genus. They describe 19 morphologically distinct stages of *Anolis* embryo development from preoviposition to hatching, with close attention paid to the development of traits known to be important in anole biology (e.g., limb and toe lamellae development). The authors suggest that this staging series can provide a baseline for future studies of evo-devo in this genus.

Sanger, T. J., J. B. Losos, and J. J. Gibson-Brown. 2007. A developmental staging series for the lizard genus *Anolis*: a new system for the integration of evolution, development, and ecology. Journal of Morphology: Early View.

Correspondence to: Thomas J. Sanger, Department of Organismic and Evolutionary Biology, Biological Laboratories, 16 Divinity Avenue, Office 4110, Harvard University, Cambridge, Massachusetts 02138, USA; e-mail: TSanger@oeb.harvard.edu.

Fecundity and Female-Biased Size Dimorphism

Although there are many factors that may influence the evolution of sexual size dimorphism, fecundity is thought to be one of the most important selective forces on body size. The authors tested the hypothesis that female body size influences offspring number and size in the Mountain Dragon, *Rankinia* (*Tympanocryptis*) *diemensis*, an Australian agamid lizard with pronounced female-biased size dimorphism. Over four sampling years, they captured 93 gravid females in the field and housed them until oviposition, after which they measured clutch and individual offspring characteristics. Results showed a positive relationship between female size (and condition) and clutch size, but no correlation between female size or condition and average egg mass, or between number of eggs in a clutch and average egg mass. There were also no correlations between maternal size and hatching mass or hatching success. The authors infer that the only way for female *R. diemensis* to increase their reproductive output is to have a larger body size and therefore produce a larger number of offspring (as females did not vary individual egg size), a conclusion that contributes to explaining the female-biased size dimorphism in this species.

Stuart-Smith, J., R. Swain, R. D. Stuart-Smith, and E. Wapstra. 2007. Is fecundity the ultimate cause of female-biased size dimorphism in a dragon lizard? Journal of Zoology 273:266–272.

Correspondence to: E. Wapstra, School of Zoology, University of Tasmania, Locked bag 5, Hobart 7001 Tasmania, Australia; e-mail: Erik.Wapstra@utas.edu.au.

A New Phylogeny of African Cobras

Although many phylogeographic studies of widespread sub-Saharan African mammals exist, there are no such studies of reptiles. The authors examined African spitting cobras, a group of widespread elapid snakes that occur in savanna habitats and were once considered to be a single species (*Naja nigricollis*), to test phylogenetic and biogeographic hypotheses and to investigate the evolution of spitting. They obtained samples from 81 individuals of 48 taxa and sequenced fragments of two mitochondrial regions (ND4 and cytochrome *b*) totaling 1333 bp. Using standard phylogenetic analyses (maximum parsimony, maximum likelihood, and Bayesian inference), they propose a new phylogenetic hypothesis for the group that supports the monophyly of African spitting cobras, and of Asiatic cobras (both spitting and non-spitting). Biogeographic results indicate that the radiation of African spitting cobras likely predates the radiation of large African ungulates and the Pliocene expansion of the African savanna. The authors suggest that spitting evolved three times in cobras, but cannot exclude alternative hypotheses. They also discuss additional taxonomic implications of the new phylogeny.

Wüster, W., S. Crookes, I. Ineich, Y. Mané, C. E. Pook, J. Trape, and D. G. Broadley. 2007. The phylogeny of cobras inferred from mitochondrial DNA sequences: evolution of venom spitting and the phylogeography of the African spitting cobras (Serpentes: Elapidae: *Naja migricollis* complex). Molecular Phylogenetics and Evolution 45:437–453.

Correspondence to: Wolfgang Wüster, School of Biological Sciences, University of Wales, Bangor LL57 2UW, UK; e-mail: w.wuster@bangor.ac.uk.

Effectiveness of Coral Snake Antivenoms

Coralsnakes (Family Elapidae) occur from the southern U.S. to Argentina. Envenomations by coralsnakes are uncommon (< 2% of total snakebites), but their bites are considered serious medical emergencies, with antivenom as the main form of treatment. In the U.S., the two medically significant species of coralsnakes are *Micrurus fulvius* (Florida Coralsnake) and *M. tener* (Texas Coralsnake). In 2006, the lone producer of antivenom used for treatment of U.S. coralsnake envenomations announced that production would be discontinued, but that adequate supplies were available to meet expected needs into late 2008. In this study, Sánchez and colleagues evaluated the effectiveness of both the discontinued Wyeth antivenom (NACSA) and Coralmyn antivenom, produced by the Mexican company, Bioclon. Venom lethal doses (LD_{50}) and antivenom effective doses (ED_{50}) were determined in lab mice. Coralmyn antivenom was able to effectively neutralize LD_{50} doses from both *M. fulvius* and *M. tener*, while Wyeth antivenom only neutralized *M. fulvius* venom.

Sánchez, E. E., J. C. Lopez-Johnston, A. Rodríguez-Acosta, and J. C. Pérez. 2008. Neutralization of two North American coral snake venoms with United States and Mexican antivenoms. Toxicon 51(2):297–303.

Correspondence: Elda E. Sánchez, Natural Toxins Research Center, 975 W. Avenue B, MSC 158, Texas A&M University-Kingsville, Kingsville, Texas 78363, USA; e-mail: eldasanchez@tamuk.edu.

Fossil Anuran with South American Affinities Unearthed in Madagascar

Faunal connections between Madagascar and other Gondwanan land masses have long been a subject of interest for biogeographers. Comparisons of fossil dinosaur assemblages and molecular analyses on ratite birds, iguanian lizards, podocnemid turtles, and boid snakes all independently point to Madagascar having a late connection to the Indian subcontinent and South America (the later likely via Antarctica). The authors describe a large, highly ossified Late Cretaceous fossil anuran from Madagascar, rivaling in size the largest known extant (*Conraua goliath*) or extinct (*Calyptocephalella*) anurans. This anuran was placed in a newly erected genus: *Beelzebufo* (*Beel'zebul* [Greek], Devil, and *Bufo* [Latin], toad), and given the specific epithet *ampinga* ([Malagasy], shield). A suite of characters place *Beelzebufo* within the hyloid clade Ceratophryinae, a group whose extant and extinct members were previously known only from South America. This find represents the first Malagasy anuran (extant or extinct) belonging to a non-ranoid group with extant representatives. Because *Beelzebufo* appears to be nested within the clade, these results also point to an early diversification of extant Ceratophryinae (before 55 Mya) and cast doubt on the currently accepted estimates on the timing of the "main hyloid" (i.e., excluding Myobatrachidae and *Calyptocephalella*) radiation (65–55 Mya).

Evans, S. E., M. E. H. Jones, and D. W. Krause. 2008. A giant frog with South American affinities from the Late Cretaceous of Madagascar. Proceedings of the National Academy of Sciences 105: 2951–2956.

Correspondence to: Susan E. Evans, Research Department of Cell and Developmental Biology, Anatomy Building, UCL, University College London, Gower Street, London WC1E 6BT, United Kingdom; e-mail: ucgasue@ucl.ac.uk.

OBITUARIES

Herpetological Review, 2008, 39(1), 9–10.

José Miguel Cei (1918–2007)

ESTEBAN O. LAVILLA
Instituto de Herpetología, Fundación Miguel Lillo
Miguel Lillo 251, 4000 Tucumán, Argentina
e-mail: eolavilla@gmail.com

JOSÉ ALEJANDRO SCOLARO
Catedra Introducción a la Ecología, Universidad Nacional de la Patagonia
H. L. Jones 143, 9120 Puerto Madryn (Chubut), Argentina
e-mail: scolaro@cenpat.edu.ar

FERNANDO VIDELA
Instituto Argentino de Investigaciones en Zonas Áridas (CONICET)
C.C. 507, 5500 Mendoza, Argentina
e-mail: fvidela@lab.cricyt.edu.ar

and
KRAIG ADLER
Department of Neurobiology and Behavior, Cornell University
Ithaca, New York 14853–2702, USA
e-mail: kka4@cornell.edu

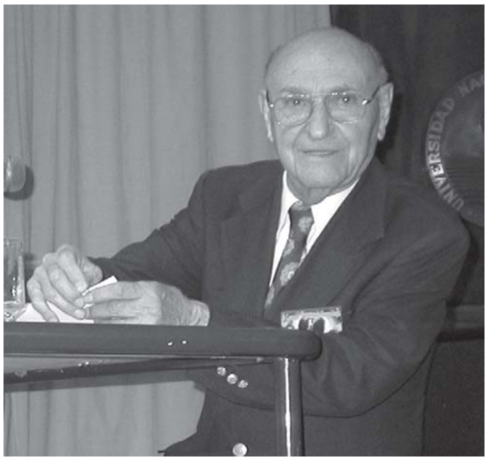


PHOTO BY ALEJANDRO SCOLERO

José M. Cei, author of landmark works on the herpetofauna of Argentina and Chile and a specialist on systematics, ecology, and biogeography, and on the chemistry of anuran skin secretions, died on 8 January 2007 in Mendoza, Argentina, at the age of 88. He was born Giuseppe Michele Alfredo Maria Cei on 23 March 1918 to a father whose early Tuscan ancestors were Greek immigrants from Ceo (Latin for Kéa or Keos, one of the Cyclades Islands in the Aegean Sea), hence the family name. His mother was a descendent of the famed 16th-century Italian, Pietro Andrea Mattioli, one of the greatest Renaissance botanists and herbalists. Cei's parents owned a business founded by his grandfather in the 1860s that produced seeds for agricultural purposes. During 1935–1939 Cei studied at the Università degli Studi di Firenze (Florence) with Nello Beccari, a comparative and neuroanatomist who specialized on amphibians and reptiles, but it was Cei's first expedition—to Ethiopia in northeastern Africa in 1939—that whetted his interest for exploration and ecology. He received his doctorate at the Università degli Studi di Pisa in 1940 and then became the equivalent of an assistant professor of zoology at Florence. His early work was on the comparative anatomy of mammals, respiration in cyclostome fishes, and sexual cycles in amphibians. It was his interest in amphibians that led him into a collegial relationship with Robert Mertens of Frankfurt am Main, then the leading European herpetologist, who became his mentor.

By 1940 World War II was well underway and Cei was mobilized by the Italian Army, first as an artillery officer in North Africa and then as an officer in the topographical service. After the Italian armistice in September 1943, Cei was held briefly by British forces, but on his release he joined Italian partisans fighting German troops in northern Italy. Eventually he was captured by the Germans. During his interrogation by a German officer, Cei learned that the officer was from Frankfurt and that, in fact, he knew of Professor Mertens. By chance, Cei still had in his pocket his last letter from Mertens, which he showed to the officer. Cei was soon released. By July 1944, Cei was working with the United States occupation forces and in 1945 returned to Florence as a teacher in courses for American soldiers organized by the American Army in Europe. Later, he decided to emigrate and, after a short research visit to Senegal, in November 1947 he arrived in Argentina where he would spend most of the rest of his life.

Cei was hired as a professor of biology at the Universidad Nacional de Tucumán in northwest Argentina and became a citizen in 1952. During this period he held several important posts, including director of the Institute of Biology and, later, of the Institute of General Biology and Experimental Embryology. In 1950 he was one of the founders of the university's School of Medicine. In 1955, he moved south to the Universidad Nacional de Cuyo in the state of Mendoza, on the eastern slopes of the Andes, where he served as professor in the School of Medicine and director of the Institute of Animal Biology until he retired in 1980. Throughout this period and even after retirement, Cei held visiting professorships in several countries including Angola, Chile, Italy, Portugal, and the USA. He maintained a research relationship with the Museo Regionale di Scienze Naturali in Torino (Turin) during 1986–1998 and served in several important governmental advisory posts at the state and national levels in Argentina. He was a member of the executive committee that organized the First World Congress of Herpetology, held in the UK in 1989. He became an honorary professor in the national universities at Río Cuarto in 1981, at Tucumán in 1990, and at Cuyo in 1995. In 2005, he was awarded an honorary doctorate by Argentina's Universidad de San Luis and in 2006 was honored for his life's work by the Legislature of his home state of Mendoza with the "Libertador General San Martin" prize.

His first publication (1940) was on the ferns of the Kaffa primary forest in Ethiopia. What followed was a steady stream of nearly 370 titles on fishes, amphibians, reptiles, and mammals covering diverse topics including sexual cycles, ecology, physiology, biochemistry, systematics, evolution, and biogeography. His works appeared every year, except for the war year of 1945, and he became a regular contributor to European, South American, and North American serials including *Journal of Herpetology*. For three decades he collaborated with the Italian pharmacologist, Vittorio Erspamer, at the Università degli Studia di Roma "La Sapienza" in Rome, to investigate biogenic substances isolated from the skins of frogs. Cei's use of various biochemical techniques allowed him to distinguish sibling species which led him to work in systematics beginning in the 1950s. He described nearly 50 new taxa of frogs (in the genera *Atelognathus, Leptodactylus, Odontophrynus, Syrrhophus* [now in *Phrynopus*], *Telmatobius*), snakes (*Atractus, Elapomorphus*), and lizards (*Cnemidophorus,* *Ctenoblepharis, Diplolaemus, Homonota, Phymaturus, Pristidactylus, Tropidurus, Vilcunia*, and especially *Liolaemus*). Cei was particularly interested in the natural history and systematics of the reptiles of arid and semi-arid environments in South America.

He authored several books, including college-level texts on animal biogeography (1946) and general biology (1951) and a Darwin anthology (1947). He is best remembered by herpetologists for his masterful monographs on the herpetofaunas of Argentina and of Chile. His "Batracios de Chile" (1962) is the companion to Roberto Donoso-Barros's volume on Chilean reptiles (1966). "Amphibians of Argentina" was issued in 1980, with supplements in 1987 and 2001, the latter co-authored with Esteban Lavilla. Cei's massive volumes on Argentina's reptiles covered the central and southern (1986) and northern and eastern regions (1993) of the country. In 1993, he co-authored a revision of the Neotropical colubrid genus *Chironius* with two herpetologists in the United States, James Dixon and John Wiest. Cei's systematic research involved some 25 years of work with his wife, Sylvana Silvi Cei, who died in 1998 (see *Herpetological Review*, 30:202, 1999). He had travelled some 350,000 km in the field with her and, as a skilled artist, she illustrated many of his books and papers.

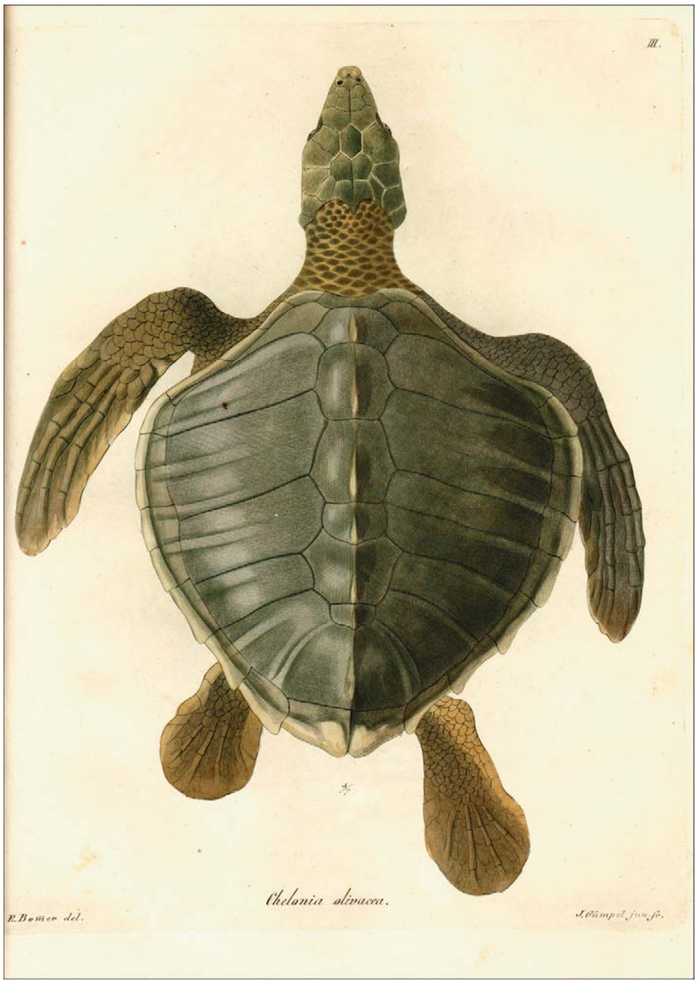
José Cei was a dynamic teacher and research colleague who had a broad knowledge of modern biology, which he integrated in innovative ways in his research. He trained several doctoral students, co-workers, and disciples in Argentina (Beatriz Alvarez, Francisco D. Barbieri, Francisco Bertini, Victor Bottari, Rosa Graciela Cohen, Alejandra Hernando, Nora Ibarguengoytía, Arnaldo H. Legname, A. E. Paz, Dora Rengel, José Alejandro Scolaro, Maria E. Tedesco, Fernando Videla, Jorge D. Williams) and in Portugal (Eduardo G. Crespo). Cei's enthusiastic personality and contagious love of science and life will be greatly missed by his colleagues throughout the world.

ZOO VIEW

RIDLEY SEA TURTLE

> Many men do little else than supply the markets with the meat and eggs. According to an expert in these matters, Richard M. Kemp, turtlers most often make their captures by means of the peg. Bits of the grass floating above the grazing turtles betray their whereabout and if they are not startled it is not difficult to fasten the peg in the shell. A peg is an iron instrument like a blunt nail, to which a line is attached; and which, when driven into the shell, easily slips out of the end of the long pole in which it is placed. By means of the line the turtle is drawn on board the boat . . . If a turtler going his rounds in the morning finds the tracks made on the way to and from the nest, he takes a short stick and carefully thrusts it down here and there in the trampled space until pushed through the egg, the yolk upon the probe discovers the nest. Fourteen nights later he is on the ground waiting til the creature comes ashore, when he turns her on her back, and in the morning carries her to the markets. Persons in the business claim that there is no diminution in numbers, which, in view of the wholesale destruction, seems hardly possible.
>
> —Samuel Garman, *On the Reptiles and Batrachians*, 1883

Kemp's Ridley Turtle (*Thalassochelys Kempii*, now *Lepidochelys kempii*) was described by Samuel Garman in 1880 (Bull. Mus. Comp. Zool. 6[6]:123–126) using two specimens sent by Richard M. Kemp of Florida. At that time, this turtle, found in the Gulf of Mexico, was called "Bastard," because it was thought to be a cross between the Green and Loggerhead turtle. Director



The Olive Ridley Sea Turtle (*Chelonia olivacea*, now *Lepidochelys olivacea*) was described in Johann Friedrich Eschscholtz's *Atlas,* issued in five parts with lovely hand-colored plates. In this work, he described new species of amphibians and reptiles, including the sea turtle pictured here.

Eschscholtz, J. F. 1829–1833. *Zoologischer Atlas: enthaltend Abbildungen und Beschreibungen neuer Thierarten, während des Flottcapitains v. Kotzebue zweiter Reise um die Welt, auf der russisch-kaiserlichen Kriegsschlupp Predpriaetië in den Jahren 1823-1826 / / beobachtet von Dr. Friedr. Eschscholtz . . .*

Imprint: Berlin, Gedruckt und verlegt bei G. Reimer.

Reprint of all the herpetological part from the *Atlas*; only parts I, II, and V contain material on amphibians and reptiles. Facsimile Reprints in Herpetology, No. 15. Ohio Herpetological Society, Athens, Ohio. 1966.

Credit: Courtesy of Emily Becker, Dana Fisher, and James Hanken, collections of Ernst Mayr Library, Museum of Comparative Zoology, Harvard University.

Patrick M. Burchfield from the Gladys Porter Zoo in Brownsville, Texas, has been involved in recovery efforts of this endangered sea turtle for the past 25 years. During that time, Burchfield has been a vital force in coordinating a joint program with the United States Fish and Wildlife Service (USFWS) and the Mexican government. He leads the U.S. portion of the Programa Binacional de la Tortuga Lora, the Binational Project by focusing on logistics, nest protection protocols, new technologies, and legal matters. He was a recipient of the USFWS Endangered Species Program's 2006 National Recovery Champion award, a prestigious honor indeed.

In a new book, *Biology and Conservation of Ridley Sea Turtles*, edited by Pamela T. Plotkin (2007, Johns Hopkins Press, Baltimore MD; ISBN-13: 978-0-8018-8611-9, ISBN-10: 0-8018-8611-2), Burchfield and two coauthors nicely summarize the recovery efforts in the chapter called "Kemp's ridley recovery: how far have we come, and where are we headed?"

Research Associate Jack Frazier from the Smithsonian's National Zoological Park Conservation and Research Center in Front Royal, Virginia, is senior author of another chapter entitled "Human-turtle interactions at sea."

Two Views of the Value of Zoos and Aquariums

For much of their history, zoos have affirmed only an imperial mastery over Nature. When operated with intelligence and compassion, however, zoos can be a most effective conservation tool. How can they best achieve this? By becoming conservation centers, by placing Nature preservation at the center of all their efforts, by reaching for the highest standards in all their projects and activities, by seeking to awake, enthrall, and educate, by articulating the wonderful benefits of conserving biological diversity, and by making strategic alliances with other cultural and natural history institutions. Zoos have the marvelous potential to develop a concerned, aware, energized, enthusiastic, caring, and sympathetic citizenry. Zoos can encourage gentleness toward all other animals and compassion for the well-being of wild places. Zoos can cultivate environmental sensitivity among their hundreds of millions of patrons. Such a populace might then want to live more lightly on the land, be more careful about using the world's natural resources, and actually to vote for politicians who care about the wild inhabitants of the earth and the health of the wild places that remain. To help save all wildlife, to work toward a healthier planet, to encourage a more sensitive populace: these are the goals for the new zoos."

—David Hancocks, *A Different Nature. The Paradoxical Work of Zoos and Their Uncertain Future*, 2001.

The zoo today is a cultural fossil, a barren evolutionary branch in the leafing of postmodernism. But it was not always so. Early on, a weak case could be made that here indeed was actual Nature, if not in the raw at least very close. Earlier zoo-goers saw the lions' fearsome teeth, felt the water buffalo's power. There was awe when the caged vulture spread its wings. Such attributes seemed real, the animals their embodiment. All it took was naïveté and a misdirected imagination after the manner of Flaubert's fictional characters Bouvard and Pécuchet. Today's zoo occupies a netherworld between real Nature, which can never be known, and a leaky bucket of cinematic "realism" that retains no secrets at all. Zoos claim to represent the first while competing actively with the second. Grasping at both, they fail to be either.

—Stephen Spotte, *Zoos and Postmodernism. Signs and Simulation*, 2006

Professionals in zoos and aquariums are faced with a multitude of critics who often ask challenging questions, the most serious of which is whether these institutions should exist at all. And if they deserve to continue, what form should they take? When these questions are generated by their colleagues with years of professional experience, it behooves the serious zoo worker to pay attention to their concerns, arguments, and conclusions.

An architect by training, David Hancocks was the director of several zoos: Woodland Park Zoo in Seattle, Washington; Arizona-Sonora Desert Museum in Tucson; and Werribee Open Range Zoo in Australia. In his writings, he has stressed the importance of naturalistic exhibits and the critical need for zoo workers to design aesthetic enclosures that appeal to both the visitor and the animals which live in these places. His book (2001; University of California Press; Berkeley; ISBN 0-520-21879-5) follows the historical evolution of zoos and aquariums throughout the world: the good, the bad, and the ugly. Overall, he stresses that much work needs to be done today to address the issues of animal welfare and conservation. Zoos and aquariums can become major forces only if enlightened and positive changes can be integrated into their missions.

Marine biologist Stephen Spotte has been director of the Aquarium of Niagara Falls, curator of the New York Aquarium, and director of Mystic Aquarium. His book (2006; Fairleigh Dickinson Press; Madison, Teaneck, New Jersey; ISBN 0-8386-4094-X) goes a step beyond Hancocks' book, for questioning whether zoos and aquariums should exist at all in their present form. His argument is unsettling: the dual purposes of education and conservation are not met in these institutions. He puts his conclusion this way, "Postmodernism is driven by images and other simulations of reality that make the exhibition of living animals passé. In shopping malls and theme parks, captive animals are transformed into their own images, becoming symbols of indirect commodification, objects of visual *flaneur*. Window shopping and a visit to the zoo have much in common."

It is easier to dismiss the views of critics outside the zoo profession but when the criticisms are generated from within our community, these harsh evaluations deserve careful scrutiny.

Bibliomania

Belin. Well; we live in a marvellous book-collecting and book-reading age—yet a word more :

Alman. I crave your pardon, Belinda ; but I have a thought which must be now imparted, or the consequences may be serious.

Lysand. I wait both your commands.

Alman. My thought—or rather the subject which now occupies my mind—is this :You have told us of the symptoms of the *Disease of Book Madness*, now pray inform us, as a tender-hearted physician, what are the *means of its cure*?

Belin. The very question I was about to put to our bibliomaniacal physician. Pray inform us what are the means of cure in this disorder?

Lysand. You should say **Probable Means of Cure**; as I verily believe that there are no certain and correct remedies.

Belin. Well, Sir ; *probable* means—if it must be so. Discourse largely and distinctly upon these.

Lysand. Briefly, and perspicuously, if you please : and thus we begin.

—Reverend Thomas Frognall Dibdin, 1811

Herpetologists throughout the ages have struggled with a seemingly incurable malady: bibliomania. Families have been torn apart, weeks spent in used bookstores, houses remodeled to accommodate more and more bookcases, and finances devastated. The condition is so severe and pervasive that a book was written about it by the Reverend Thomas Frognall Dibdin in 1811—*Bibliomania, or, Book Madness: A Bibliographical Romance, in Six Parts: Illustrated with Cuts* (Printed for the author, by J. M'Creery ..., and sold by Messrs. Longman, Hurst, Rees, Orme, and Brown).

The University of Michigan has digitized this book and it is available at http://mdp.lib.umich.edu/cgi/pt?id=39015005643682.

A lasting cure has still not been found to this day. Read *A Gentle Madness: Bibliophiles, Bibliomanes, and the Eternal Passion for Books,* by Nicholas A. Basbanes (New York, H. Holt and Co., 1995).

A Chelonian Armageddon

On 25–28 July 2007, I traveled to Atlanta, Georgia to attend the 5th Annual Symposium on Conservation and Biology of Freshwater Turtles and Tortoises, a joint annual meeting sponsored by the IUCN Turtle Survival Alliance (TSA) and IUCN Tortoise and Freshwater Turtle Specialist Group. Although I knew that the news about the continued survival of many of these creatures was problematical, I was not prepared for the constant stream of troubling papers by workers documenting pressures throughout the world, with many showing graphic images detailing the causes: habitat destruction and alteration, poaching and stockpiling of turtles and tortoises in range countries for eventual slaughter to satisfy the demand for pâté in Asia, mounds of hundreds of chelonians held in food markets, disease outbreaks, commercial turtle farming, trade in plastrons and shells for curios, and collection for the commercial pet trade. To get a sense of the scope of this catastrophe, consult IUCN Red List of Threatened Species 2006 <http://www.iucn.org/themes/ssc/redlist2006/redlist2006.htm>.

Each evening, many delegates gravitated to the nearest saloon to try and make sense about we had heard and what strategies could be employed to reverse the trend. By the end of the conference, I was nearly in shock even though organizer Rick Hudson tried to lift spirits by saying that the mission of TSA was to ensure that no chelonian became extinct during this century. It was gratifying to see that so many committed people, battling heroically against exploiters, developers, and bureaucrats, cared about the fate of terrapins, turtles, and tortoises; their efforts deserve support.

For more information on TSA activities, see 2004 *Herpetological Review* 35[2]:110 and contact Dwight Lawson (e-mail: dlawson@zooatlanta.org) or Rick Hudson (e-mail: rhudson@fortworthzoo.org), or visit the TSA website at www.turtlesurvival.org.

Biographies

Kraig Adler has written two important historical biographies of deceased biologists and their contributions to the science of herpetology: *Herpetologists of the Past, Part 1* (1989) and *Herpetologists of the Past, Part 2* (2007). In Adler's spectacular volumes, there are many references to zoos and aquariums—names of importance to the zoo community, details of the careers of people working in these places or authors writing about these institutions (Table 1). Each biography gives a sketch of the person's life with special emphasis on herpetology, references to other biographical information, the person's signature, and in almost every case their portrait. To obtain a copy of vol. 2, contact SSAR Publications Secretary Breck Bartholomew (details located inside front cover of this issue). Vol. 1 is now out of print but available on the web (www.ssarherps.org) [click on "Publications"]. Additional information may be found in Card and Murphy (2000. SSAR Herpetol. Circ. No. 27, 44 pp.) and Murphy (2007. Herpetological History of the Zoo and Aquarium World. Krieger Publ. Co., Malabar, Florida).

Table 1. Names of zoo-affiliated herpetologists profiled in *Contributions to the History of Herpetology*.

Volume 1 (1989)	Volume 2 (2007)
Raymond L. Ditmars *, **	Ensil Ross Allen **
Afrânio do Amaral	Miguel Alvarez del Toro **
Archie F. Carr Jr.	Walter Auffenberg Jr.
Doris M. Cochran	William Bartram
Georges Cuvier **	Johann Matthaeus Bechstein **
André-Marie-Constant Duméril **	John L. Behler **
Auguste-Henri André Duméril **	Edward G. Boulenger **
Leopold J. F. J. Fitzinger **	Alfred E. Brehm **
Carl F. Kauffeld *, **	Arthur E. Brown *, **
Vivian F. M. FitzSimons **	Mark Catesby
Emilio Augusto Goeldi **	Roger Conant *, **
Laurence M. Klauber *, **	Charles R. Darwin **
Wilhelm K. A. Klingelhöffer **	Edward R. Elkan **
B.-G.-É. Lacepède **	Johann von Fischer **
Arthur Loveridge	Frederick W. FitzSimons **
Robert F. Mertens **	Vivian F. M. FitzSimons **
Charles R. S. Pitman **	Stanley Smyth Flower **
Clifford H. Pope	Heini Hediger **
Raymond Rollinat	Catherine G. Hopley **
Hermann Schlegel **	James A. Oliver *, **
Karl P. Schmidt	Richard Owen
Léon-Louis Vaillant **	C. B. "Si" Perkins *, **
Zdenek Vogel **	Joan Beauchamp Procter **
Franz Werner **	Heinz H. Reichenbach-Klinke **
	Charles E. Shaw *, **
	Eric F. Worrell **

* *Lineages of zoo herpetologists in the United States.*
** *Herpetological History of the Zoo and Aquarium World.*

In the paper following, Hans-Werner Herrmann describes his exciting project in Cameroon which includes a study of the endangered Goliath Frog (*Conraua goliath*). This anuran is rarely kept in zoo collections because it is generally a delicate captive. An adult individual was successfully maintained at the Chicago Zoological Park (Brookfield Zoo) for 4 years, 4 months, and 7 days in a large semi-aquatic exhibit (Snider and Bowler 1992. SSAR Herpetol. Circ. No. 21, 40 pp.). In the mid-1960s at the Ft. Worth Zoo in Texas, late curator John Mehrtens hung wide, flexible strips from the top to the bottom of the enclosure to cover walls and exhibit glass; he said this arrangement prevented the jumping frogs from injury. From the Duisburg Zoological Garden in Germany, Gewalt (1977. Zool. Gart. [N.F.], Jena 47:161–192) covered catching, transport and keeping these frogs. Gillespie et al. (1988/1989. Proc. Amer. Assoc. Zoo Vet., p. 62) sexed Goliath Frogs by laparoscopy at the Cincinnati Zoo.

The American Association of Zoological Parks and Aquariums (AAZPA) was founded in 1924. Seventy years later, the name was changed to the Association of Zoos and Aquariums (AZA). Some of the organizational purposes of AZA are 1) to promote discussions and cooperation among zoo and aquarium professionals; 2) to set high standards for its accredited member institutions, now over 200; and 3) to support *in situ* and *ex situ* conservation initiatives. In the article below, Shelly Grow and Ruth Allard highlight the ambitious initiative by these institutions to address the issue of amphibian declines and the role that zoos and aquariums will play in 2008 to reverse this trend.

Herpetological Review, 2008, 39(1), 13–15.

Zoo-based In-situ Herpetology: Setting Baselines in Western Cameroon

HANS-WERNER HERRMANN
Arizona Research Laboratories &
School of Natural Resources (Wildlife Conservation and Management)
University of Arizona, 1657 E. Helen Street (BIO5)
Tucson, Arizona 85721, USA
e-mail: hwh@u.arizona.edu

In 1998, when I was the curator of Herpetology, Ichthyology and Invertebrates at Cologne Zoo in Germany, I was contacted by representatives of the German branch of the American Linen Supply Company (ALSCO). The company wanted to contribute to the conservation of species and biodiversity. ALSCO Germany felt that their corporate identity would be enhanced by chameleons and that they would like to sponsor a chameleon enclosure at Cologne Zoo. After further correspondence, I realized that this could extend far beyond the common zoo practice of sponsored enclosures. Subsequently, we developed an outline for a conservation initiative sponsored by ALSCO Germany and implemented by Cologne Zoo. Chameleons were to be the flagship species for this project which in reality encompassed multiple species of amphibians and reptiles.

We eventually set our sights on Cameroon. Southwest Cameroon is one of the chameleon diversity hotspots in Africa, in particular the mountain chain known as "Dorsale Camerounaise" contains numerous montane species. We focused our attention on two areas: 1) Mt. Nlonako, a tropical rainforest mountain that reaches 1800 m in elevation situated approximately in the center of the north–south extending "Dorsale Camerounaise" and 2) Tchabal Mbabo, a mountain range at the northern limit of this mountain chain representing a humid island with cloud forests in a tropical savanna landscape.

Compared to other tropical African countries, Cameroon has received a considerable amount of herpetological attention, especially owing to the works by Swiss herpetologist J.-L. Perret and French herpetologist J.-L. Amiet, who both described a large number of new species from this area over the last 50 years. However, complete herpetofaunal inventories, quantitative surveys, and ecological studies on amphibians and reptiles are rare or lacking for large parts of Africa, including Cameroon. Baseline data from such studies constitutes the foundation on which conservation and management decisions should be based.

We started herpetofaunal inventories in 1998 at both locations, Mt. Nlonako and Tchabal Mbabo—the latter posing remarkable logistic challenges due to its remoteness and inaccessibility (Herrmann et al. 1999, 2000; Herrmann and Herrmann 1999, 2002; Kolter et al. 2001). The cooperate funding during the initial phase of the project permitted us to invite herpetologists from the Zoological Research Institute and Museum Alexander Koenig (ZFMK) in Bonn. Wolfgang Böhme and his students Oliver Euskirchen and Andreas Schmitz, who had just finished their graduate work at nearby Mt. Kupe, joined us for field work. At this time I was adjunct faculty at Philipps University in Marburg, where as a part of my teaching commitment, I gave herpetological lecture series and seminars. Some of the graduate students showed interest in fieldwork in Cameroon and joined us during several of our field trips. Later two of them, Mirco Plath and Markus Solbach, decided to conduct their graduate research on anuran communities at Mt. Nlonako (Plath 2003; Plath et al. 2004; Solbach 2003).

In 2002 I joined the Zoological Society of San Diego's Center for Reproduction of Endangered Species for a three-year field study on Goliath Frogs (*Conraua goliath)* in Cameroon. The species' distribution is limited to Cameroon and Equatorial Guinea. Goliath Frogs are under severe pressure from hunting and habitat degradation. During many long nights in rapids and around waterfalls of Cameroon's rivers, the preferred habitat of Goliath Frogs, we processed a large number of frogs, tadpoles, and eggs—which are deposited in nests on gravel banks. We used capture-recapture techniques and radiotelemetry (Figs. 1, 2) to gain insight into the species' ecology and behavior. As the number of Goliath Frogs in zoological collections is very limited, we performed morphological and anatomical studies on sexual diversity, reproductive status, stomach contents, and diet. Additionally, a large number of tissue samples were collected. These tissues are now being used in DNA-based studies on the phylogeography, population genetics, and mating systems of Goliath Frogs.

In 2003, Taylor Edwards from the University of Arizona joined us for seasonal field work. During his stay we discovered a novel type of locomotion in Goliath Frogs—"skittering"—which had previously been seen only in much smaller frogs (Herrmann and Edwards 2006). On 23 February 2003 at 0200 h, we found a pair of Goliath Frogs mating in a nest. The male measured 330 mm SVL, with a mass of 3220 g—which makes it the largest frog of this or any other living species ever documented (Fig. 3). This size is rivaled only by the recently described fossil frog *Beelzebufo ampinga* (Evans et al. 2008) which, extrapolated from skull bone fragments, may have reached SVL of > 400 mm. Does this indicate the functional size limit of the anuran body plan?

As for chameleons, which initially caught ALSCO's attention, we studied chameleon spatial distribution at Mt. Nlonako (Harbort

FIG. 1. Hans-Werner Herrmann and Taylor Edwards (right) implant a radio transmitter under field conditions into an adult Goliath Frog (*Conraua goliath*).

FIG. 3. A male Goliath Frog captured on 23 February 2003 at 0200 h with a snout–vent length of 330 mm and a mass of 3220 g. Note the casting net that is used to capture the frogs.

and Herrmann 2002) and aspects of the reproductive biology of Mountain Chameleons (*Chamaeleo montium*) (Herrmann and Herrmann 2005). Chameleons are a characteristic and relatively abundant herpetofaunal element of SW Cameroon. They are particularly attractive lizards and are in high demand in the international reptile pet trade. Large numbers are collected and exported from Cameroon. In this situation, baseline data and research on the biology of chameleons with the goal of establishing meaningful management plans is an immediate need.

FIG. 2. Field assistant Solomon Echalle crossing a river during a Goliath Frog radio tracking session.

A specimen of Sundevall's Worm Snake (*Leptotyphlops sundewalli gestri*) from the lower slopes of Mt. Kupe proved to be the second specimen recorded from Cameroon, and is a significant taxon range extension (Herrmann et al. 2004).

At the end of 2004 we concluded the detailed inventories of amphibians and reptiles at Mt. Nlonako (Herrmann, Böhme, Euskirchen et al. 2005; Herrmann, Böhme, Herrmann et al. 2005) and Tchabal Mbabo (Herrmann et al. 2007). Mt. Nlonako was established as the center for species richness for a single locality in Africa, with 93 amphibian and 63 snake species. Indeed, with regard to snake species diversity, Mt. Nlonako ranks as one of the most species-rich places in the world. Two new frog species, *Cardioglossa alsco* from Tchabal Mbabo (Herrmann et al. 2004) and *Phrynobatrachus nlonakoensis* from Mt. Nlonako (Plath et al. 2006), were discovered during our survey work.

An extensive collection of herpetological vouchers is now deposited at the Zoological Research Institute and Museum Alexander Koenig, Bonn (ZFMK), and the National Museum of Natural History, Smithsonian Institution, Washington, D.C. (USNM).

Acknowledgments.—I wish to thank Mr. Horst Nobis of ALSCO Germany for his substantial financial contribution which initiated and facilitated this endeavor. I am grateful to Cologne Zoo and the San Diego Zoological Society for additional financial support. I would like to thank the local experts, mostly Cameroonian villagers, without whom only a fraction of the amphibians and reptiles would have been found. Special thanks to my Cameroonian project field assistants Otto Mesumbe, Solomon Echalle, Bartholomew Tchiengue, and Dansala Djire. Dansala died tragically in December 2004. Taylor Edwards helped tremendously, both in the field and in the lab. Numerous discussions with Taylor inspired a multitude of new ideas. Roger Repp edited the manuscript. The Cameroonian Ministries of Scientific and Technical Research (MINREST) and Environment and Forestry (MINEF) issued research, collecting, and export permits.

LITERATURE CITED

EVANS, S. E., M. E. H. JONES, AND D. W. KRAUSE. 2008. A giant frog with South American affinities from the Late Cretaceous of Madagascar. PNAS 105(8):2951–2956.

HARBORT, J., AND H.-W. HERRMANN. 2002. Untersuchungen an ausgewählten Chamäleon-Populationen am Mt. Nlonako/Kamerun mit Bemerkungen zu einem *Chamaeleo camerunensis*. Herpetofauna, Weinstadt 24(140):25–34.

HERRMANN, H.-W., W. BÖHME, O. EUSKIRCHEN, P. A. HERRMANN, AND A. SCHMITZ. 2005. African biodiversity hotspots: The reptiles of Mt Nlonako, Cameroon. Revue suisse de Zoologie 112:1045–1069.

———, ———, AND P. HERRMANN. 2000. The ALSCO Cameroon expedition 1998: The sampling of a mountain rainforest. Bonn. zool. Monogr. 46:95–103.

———, ———, P. A. HERRMANN, M. PLATH, A. SCHMITZ, AND M. SOLBACH. 2005. African biodiversity hotspots: the amphibians of Mt Nlonako, Cameroon. Salamandra 41:61–81.

———, AND T. EDWARDS. 2006. *Conraua goliath* (Goliath frog). Skittering locomotion. Herpetol. Rev. 37(2):202–203.

———, AND P. A. HERRMANN. 2002. Herpetological conservation at the Cologne Zoo. Herpetol. Rev. 33(3):168–169.

———, ———, AND W. BÖHME. 1999. Die ALSCO-Kamerun-Expedition I: Amphibien und Reptilien vom Mt. Nlonako. Z. Kölner Zoo 42(4):181–197.

———, ———, A. SCHMITZ, AND W. BÖHME. 2004. A new frog species of the genus *Cardioglossa* from the Tchabal Mbabo Mtns, Cameroon. Herpetozoa 17(3/4):119–125.

———, ———, AND V. WALLACH. 2004. *Leptotyphlops sundewalli gestri* (Sundevall's Worm Snake). Distribution. Herpetol. Rev. 35(4):411.

———, A. SCHMITZ, P. A. HERRMANN, AND W. BÖHME. 2007. Amphibians and reptiles of the Tchabal Mbabo Mtns, Adamaoua Plateau, Cameroon. Bonner zool. Beitr. 55(1):27–35.

HERRMANN, P., AND H.-W. HERRMANN. 1999. The ALSCO Cameroon chameleon expedition: A rainforest survey on Mount Nlonako. Sonoran Herpetologist 12(9):94–97.

———, AND ———. 2005. Egg and Clutch Characteristics of the Mountain Chameleon, *Chamaeleo montium*, in Southwestern Cameroon. J. Herpetol. 39(1):154–157.

KOLTER, L., W. KAUMANNS, AND H.-W. HERRMANN. 2001. Forschung im Zoologischen Garten Köln. Z. Kölner Zoo 44 (2):83–96.

PLATH, M. 2003. Räumliche Verteilungsmuster der Anuren-Gesellschaft eines Fließgewässers im Tiefland-Regenwald am Mt. Nlonako/Kamerun unter Berücksichtigung populationsökologischer Aspekte. Unpublished MSc Thesis, Philipps University, Marburg. 166 pp.

———, H.-W. HERRMANN, AND W. BÖHME. 2006. New frog species of the genus *Phrynobatrachus* (Anura: Phrynobatrachidae) from Mt Nlonako, Cameroon. J. Herpetol. 40(4):486–495.

———, M. SOLBACH, AND H.-W. HERRMANN. 2004. Anuran habitat selection and temporal partitioning in a montane and submontane rainforest in Southwestern Cameroon—first results. Salamandra 40(3/4):239–260.

SOLBACH, M. 2003. Systematik und Ökologie der Batrachofauna einer montanen Regenwaldregion am Mt. Nlonako in Westkamerun. Unpublished MSc thesis, Philipps University, Marburg. 241 pp.

Herpetological Review, 2008, 39(1), 15–22.

The Zoological Community Celebrates 2008: The Year of the Frog

SHELLY GROW
and
RUTH ALLARD
Association of Zoos & Aquariums, 8403 Colesville Road, Suite 710
Silver Spring, Maryland 20910, USA
e-mail: SGrow@aza.org

YEAR OF THE FROG: ADDING THE *EX SITU* COMMUNITY'S STRENGTHS TO CURB AMPHIBIAN EXTINCTIONS

Since its formation in 1990 and until its merger with other World Conservation Union (IUCN) amphibian conservation groups into the IUCN/Species Survival Commission (SSC) Amphibian Specialist Group (ASG) in 2006, the Declining Amphibian Population Task Force (DAPTF) led the charge to verify anecdotal reports beginning in the 1970s of drastic and global amphibian population declines and disappearances, identify target populations and regions meriting immediate attention, gather evidence concerning the causal factors contributing to amphibian population declines, set up infrastructure to facilitate long-term data collection, and to disseminate information throughout the scientific community and to the public (Heyer and Murphy 2005). In September 2005, the IUCN and Conservation International convened an Amphibian Conservation Summit in Washington, DC to devise a global strategy of action to arrest amphibian declines and extinctions (Mendelson et al. 2006). A key outcome of this meeting was the *Amphibian Conservation Action Plan* (ACAP), which was published in July 2007 (Gascon et al. 2007). Even before its final publication, researchers, land managers, zoological community professionals, and others had already started tackling the pieces for which they were responsible. Momentum continues to build as awareness about the amphibian conservation crisis grows.

The ACAP charges the global zoological community with leading *ex situ* conservation efforts, creating assurance populations for those species that would go extinct before the threats facing them could be addressed in the wild (Lips et al. 2005; Mendelson et al. 2006; Pounds et al. 2007; Young et al. 2007). *Ex situ* conservation is only one component of the much larger holistic approach of the ACAP and species that are candidates for *ex situ* conservation primarily consist of those listed as Endangered and Critically Endangered by the Global Amphibian Assessment (GAA) (IUCN et al. 2006). The *ex situ* programs are implemented by regional zoological associations: the Association of Zoos and Aquariums (AZA) in the United States, the European Association of Zoos and Aquariums (EAZA), the Australasian Regional Association of Zoological Parks and Aquaria (ARAZPA), etc. To facilitate these efforts at the international level, the IUCN's Conservation Breeding and Amphibian Specialist Groups (CBSG and ASG), and the World Association of Zoos and Aquariums (WAZA) formed the Amphibian Ark (www.amphibianark.org) (Zippel 2007a).

An additional function of Amphibian Ark is to develop and deploy a public awareness and fundraising campaign to support the *ex situ* conservation community's efforts. Effectively addressing the amphibian conservation crisis requires rapid expansion of zoos' and aquariums' capacity to meet immediate needs and funding to support these projects into the future. Amphibian Ark has designated 2008: Year of the Frog as the umbrella for this global effort. The goal of this campaign is to ensure that the general public will understand the critical issues facing amphibian populations and support the zoological community's efforts. The Amphibian Ark coordinates the international communication and networking associated with Year of the Frog, while regional zoological associations implement the campaign and related efforts in their respective regions.

Demonstrating a Growing Commitment to Amphibian Conservation and Public Outreach: A Regional Example

AZA's Year of the Frog is an integrated campaign approved by the AZA Board of Directors and developed by a task force representing AZA's Amphibian Taxon Advisory Group (ATAG; amphibian experts in the AZA community that guide collection planning, provide strategic conservation and research direction, and strive to improve amphibian husbandry throughout the AZA community); Conservation Education (CEC), Public Relations, and Marketing committees; and AZA executive office communications and conservation department staff.

While the emphasis of AZA's Year of the Frog is on challenging zoos and aquariums to expand or develop *ex situ* conservation initiatives, the campaign also encourages zoos and aquariums to support complementary *in situ* conservation efforts, develop capacity internationally, address local amphibian conservation through monitoring, surveying, research, partnering with local government agencies, advocacy, and to engage their 157 million visitors to build popular support for amphibian conservation.

The AZA Year of the Frog task force facilitates public engagement opportunities. The CEC has gathered, reviewed, culled and edited existing resources, and has created new amphibian education and outreach materials that focus on encouraging positive conservation impacts. They have also worked with the ATAG to develop fact sheets to inform visitors, docents, and staff about amphibians, population declines, the zoological community's response, and what visitors can do to help amphibians. The task force has developed campaign graphics and other items for widespread distribution, including a public service announcement that can be personalized for each participating zoo or aquarium's market. These materials, customized for a North American audience but also consistent with the global Amphibian Ark effort, are available free of charge to AZA members and the general public through the AZA Web site (www.aza.org/yearofthefrog).

The task force is also responsible for publicizing AZA members' amphibian conservation efforts within the zoological community and to a larger public audience. Articles for zoo and aquarium member magazines regarding amphibian declines and the zoological community's response have been generated, press releases have been circulated, and communication continues with well-known popular media outlets. For the campaign year, each monthly edition of the AZA member magazine *Connect* features an amphibian conservation-related story and the March 2008 edition will be devoted entirely to amphibian conservation. The general public can visit the AZA Web site and become "Friends of Frogs" by joining a grassroots advocacy network that will be contacted regarding regional and national opportunities for action. AZA zoos and aquariums will launch Year of the Frog on Leap Day, 29 February 2008, will use amphibian themes at their annual Party for the Planet on and/or around Earth Day (22 April 2008), and will also incorporate amphibian themes in their year-round education programming.

As amphibian experts for the AZA community, the ATAG provides strategic direction and shares expertise. In July 2007, the Fort Worth Zoo hosted a meeting where the ATAG identified amphibian species in Canada, the United States, Mexico, and the Caribbean whose populations would most benefit from *ex situ* conservation programs. Meeting participants included members of the ATAG steering committee and its advisory committee, staff from Amphibian Ark, and university scientists. The group utilized a prioritization tool developed by the Amphibian Ark that incorporates the species' conservation status, threats, the potential for mitigating these threats in the wild, phylogenetic uniqueness, social and scientific value, and other key variables. The group examined over 200 species listed by the GAA as Critically Endangered, Endangered, Data Deficient, or listed under the US Endangered Species Act. The resulting priority rankings are subject to change as new information and management processes are implemented, but in the meantime, provide much-needed direction for next steps. Programs to protect species outside of the US will be developed in collaboration with local experts, international zoological colleagues, and the Amphibian Ark, while programs within the US will complement state and federal priorities. The priorities, a description of the process used, and an analysis of current AZA amphibian collections and capacity (staffing, space, etc.) are included in the *Action Plan for* Ex Situ *Amphibian Conservation in the AZA Community* report (Crump and Grow 2007). The Action Plan was modeled deliberately after the *ARAZPA Amphibian Action Plan* published in June 2007 (Gillespie et al. 2007), so that zoological community capacity can be compared and evaluated internationally.

In addition to the Action Plan, the ATAG published the *Amphibian Conservation Resource Manual* in September 2007 with AZA staff support (Grow and Poole 2007). The Resource Manual is designed to help zoos and aquariums incorporate amphibian conservation into their institutional collection and master plans, create holistic *in situ* and *ex situ* conservation and research projects, build biosecure amphibian holding facilities, identify funding resources to support amphibian efforts, and develop programs for those species that would most benefit from *ex situ* conservation programs. The ATAG has also produced an *Amphibian Husbandry Resource Guide* so that AZA members can expand their staff skillsets and can respond appropriately to the conservation crisis (Poole and Grow 2008). When working with endangered species, there is little room for error, so all of these tools are designed to build capacity and expertise.

Perhaps the most important component of the Year of the Frog campaign is the infusion of AZA's significant and growing amphibian conservation efforts with a sense of urgency. AZA recognizes that the rapid and widespread declines in amphibian populations present a conservation challenge unlike any other faced by humans (Stuart et al. 2004). The Association is responding by dedicating more resources and attention to amphibians now than it has ever committed to any one taxonomic group in the organization's history. For example, the Board of Directors created a conservation biologist position at the AZA office dedicated exclusively to amphibian conservation. Since 2004, the AZA Board of Regents has held an annual training course in *Amphibian Biology and Management*. In 2007, the Board of Regents offered this course twice to meet unprecedented student demand, thanks in large part to the generosity of volunteer instructors and the Toledo Zoo, which hosted both sessions. Because the April 2008 course filled by late 2007, it is likely that arrangements will be made to offer the course twice in 2008 as well. Tight budgets have been stretched to fund travel expenses for key meetings to expand amphibian conservation partnerships and every department at the AZA office has been engaged in Year of the Frog efforts.

The AZA community is expanding Year of the Frog beyond the zoological sector by engaging appropriate government entities in the campaign as well. At the national level, AZA's government affairs department is working with U.S. Congressional staff to support amphibian conservation, and at the local level, AZA members are engaging their public in local development projects affecting amphibian habitat and inviting civic officials to help launch and participate in Year of the Frog. Members are also reaching out to other local partners, such as parks, museums, and schools, to celebrate the Year of the Frog.

Year of the Frog Expectations

2008: Year of the Frog activities will raise the profile and the level of public support for amphibian conservation efforts. The Year of the Frog will also feature significant fundraising efforts. Already, the AZA has developed an amphibian-specific funding process that has received significant contributions from private individuals and companies. International and collaborative fundraising efforts, such as those led by the Emerging Wildlife Conservation Leaders program (EWCL; a collaborative effort sponsored by Defenders of Wildlife, U.S. Fish and Wildlife Service (FWS), and White Oak Conservation Center/Howard Gilman Foundation), the ATAG small grants program, Project Golden Frog/Atelopus Conservation Trust (Zippel 2002), and Amphibian Ark are also underway.

Conservation success requires a long-term commitment and, as demonstrated by long-term participation in amphibian Species Survival Plans® (see next section for more detail), AZA facilities are dedicated to these efforts. 2008 will see the expansion of existing amphibian programs to both increase population sizes and include additional facilities. It will also welcome the laying of groundwork for additional programs that may not come online until after 2008. The Year of the Frog conservation goals are to lay the foundation for long-term amphibian conservation action and to garner institutional commitments so that these programs can grow over the next several years. Successful programs will not sprout immediately, but many new efforts will begin to take shape in 2008.

Partnerships are a key element to amphibian conservation success. The first 2008 edition of FWS' *Endangered Species Bulletin* periodical will be dedicated to amphibian issues, and approximately half of the articles will come from AZA-accredited zoos and aquariums. AZA members are increasingly engaging with Partners in Amphibian and Reptile Conservation (PARC), and each regional PARC network has been challenged to develop collaborative activities with local zoos and aquariums and to celebrate Year of the Frog at its 2008 meetings and events. Three AZA-accredited zoos and aquariums, the Columbus Zoo and Aquarium, Phoenix Zoo, and Houston Zoo, supported PARC's recent symposium, "Amphibian Decline and Chytridiomycosis: Translating Science into Urgent Action." Staff from fifteen zoos and aquariums and the AZA executive office participated in the meeting. The EWCL program has committed to raise funds so that AZA member Africam Safari in Mexico can expand its amphibian conservation work. AZA is helping EWCL with its fundraising and public outreach efforts. AZA is also growing its international partnerships, working with the Amphibian Ark and the Zoo Conservation Outreach Group (ZCOG) to train and support the work of international colleagues, utilizing internationally consistent prioritization processes, and sharing resources, information, and Year of the Frog campaign strategies.

Collaborative Conservation: A History of Commitment

AZA-accredited zoos and aquariums are already an active force in amphibian conservation (Wiese and Hutchins 1994; Zippel 2007b). Since 2000, AZA members have spent more than $1.1 million on amphibian conservation and research projects in more than 20 countries around the world. Two amphibian species, the Puerto Rican Crested Toad (*Peltophryne lemur*) and Wyoming Toad (*Anaxyrus baxteri*)[1] are already part of AZA's Species Survival Plan® (SSP) program (Fig. 1). The first amphibian SSP program was approved in 1984, focusing on the Puerto Rican Crested Toad (Johnson 1994). In 2007, more than 20 AZA-accredited zoos and aquariums continue to hold, breed, and provide animals for reintroduction at three sites in Puerto Rico. The program is far larger than its *ex situ* components: AZA members have developed extensive education and outreach programs that incorporate local folklore, passions, and interests, have helped create and maintain new toad habitat and have developed partnerships with government, academic, and other stakeholders throughout Puerto Rico. The second amphibian flagship program, the Wyoming Toad SSP, was approved in 1996. Listed by the GAA as Extinct in the Wild, nine AZA facilities continued to hold, breed, and reintroduce animals in 2007, in collaboration with other breeding facilities, the Wyoming Game and Fish Department, and the FWS. Causes of the population crash in the mid-1970s were unknown for decades, but the proximate cause of decline has now been attributed to the fungus *Batrachochytrium dendrobatidis* (*Bd*), complicating the rearing and reintroduction program (Johnson 1994; Odum and Corn 2005; Stebbins and Cohen 1995). Both of these SSP programs are fairly well known, in part due to their longevity and collaborative nature; however, other amphibian conservation efforts at AZA zoos and aquariums may be less familiar. The following program descriptions offer a sampling of what AZA institutions are currently doing and demonstrate the expertise and enthusiasm AZA institutions have for long-term amphibian conservation action.

Zoo staff were among the key players in discovering the novel pathogen, *Bd*, and the effect it was having on amphibians (Nichols 2003). In 1991, Dr. Donald K. Nichols took on a position as a pathologist at the National Zoological Park (NZP), following his experience as a resident there and as a pathologist at the National Institute of Health. He had consulted with zoos and universities throughout the country and in 1991 received the bodies of three

Fig. 1. The first amphibian AZA Species Survival Plan was approved for the Puerto Rican Crested Toad (*Peltophryne lemur*) in 1984. Photo courtesy of John Kast, Fort Worth Zoo.

Fig. 2. The Toledo Zoo and Bronx Zoo (Wildlife Conservation Society) house the only known surviving Kihansi Spray Toads (*Nectophrynoides asperginis)* and are developing plans in collaboration with international colleagues and the Tanzanian government to reintroduce this species to Tanzania in the future. Photo courtesy of R. Andrew Odum, Toledo Zoo.

preserved California Arroyo Toads (*Anaxyrus californicus*)^2. The toads came from a captive colony whose population had declined 60% in just a few months; an unknown skin disease was responsible for their demise. Dr. Nichols reviewed amphibian pathology files from the NZP collection and saw evidence of the same disease. Over the next several years, clues surfaced indicating that a chytrid fungus might be involved; however, no chytrids had ever been recognized as pathogens in vertebrates at that time. In 1996, a new outbreak of the disease affected the National Zoo's collection. The zoo's resident pathologist at the time, Dr. Allan Pessier, and Dr. Nichols were able to locate and send fresh skin samples to one of the few chytrid experts in the world, Dr. Joyce Longcore. Dr. Longcore identified the fungus as not just a new chytrid species, but also as a previously unknown genus. The organism was eventually named *Batrachochytrium dendrobatidis*, meaning "frog chytrid of dendrobatids" (Longcore et al. 1999). While the organism was being identified, researchers in Central America and Australia began reporting identical situations in the field. While unable to culture the organisms, they had also identified the organisms as chytrids and Longcore later confirmed them as the same species (Berger et al. 1998; Nichols 2003). Zoological facilities continue to have an important role to play in efforts to prevent *Bd* from spreading between captive facilities and in trade, monitoring the disease, participating in captive-breeding and reintroduction programs, and limiting the impact of the disease on wild amphibian populations (Young et al. 2007).

Combining *in situ* and *ex situ* work, in 2000 the Bronx Zoo brought 500 Kihansi Spray Toads (*Nectophrynoides asperginis*) from Tanzania to US zoos when their habitat came under threat from the development of a dam (Krajick 2006) (Fig. 2). This transfer turned out to be extremely fortunate as the entire population collapsed in 2003. The timing of the crash coincided with the completion of the dam's construction and related changes to the ecosystem, the brief opening of floodgates to flush sediments, and the first observation of the fungus *Bd* in the habitat (Krajick 2006). Today, the sole surviving Kihansi Spray Toads live at the Toledo and Bronx Zoos. In May 2007, AZA members from the Bronx, Toledo, and San Diego zoos met with government and World Bank officials, facilitators from the IUCN's Conservation Breeding Specialist Group (CBSG), and other scientists to complete a Population and Habitat Viability Assessment and develop a long-range strategy to reintroduce the spray toad back to Tanzania.

Based on predictions made by Lips et al. (2006) on the rate that *Bd* was moving through Panama and its potential impacts, biologists from Zoo Atlanta and the Atlanta Botanical Garden knew that the devastating fungus *Bd* was rapidly approaching a hotspot of amphibian diversity in that country's central region (Norris 2007). Based on its location, elevation, and species assemblages (Lips et al. 2005), *Bd* was expected to take a high toll on its amphibian populations. Working with others in Panama and building on partnerships developed through Project Golden Frog (Zippel 2002), these researchers spearheaded an effort in 2005 to evacuate hundreds of frogs representing 35 species from central Panama in an effort to protect them from this devastating disease (Mendelson and Gagliardo 2005; Gagliardo et al., *in press*). The Houston Zoo and El Nispero Zoo (Panama) began constructing the first in-country, dedicated facility for maintaining critically threatened amphibians locally (Fig. 3). More than two dozen AZA facilities have supported the construction, training, and operations costs of this facility. The *Bd* fungus reached El Valle in early 2006 and wild amphibian populations began to plummet. While held temporarily at the Hotel Campestre as building construction was completed, today many of the species decimated by the arrival of *Bd* can still be found in assurance populations in the US and at El Valle Amphibian Conservation Center (Fig. 4). In 2007, the Houston Zoo was awarded a grant from the AZA Conservation Endowment Fund (CEF) to continue to support this Center and expand its outreach efforts in each of the partnering zoos and aquariums in North America.

FIG. 3. Jorge Garcia from Panama's Autoridad Nacional del Ambiente (ANAM) helps transfer Panamanian Golden Frogs (*Atelopus zeteki*) from temporary quarters to the El Valle Amphibian Conservation Center. Photo courtesy of Bill Konstant, Houston Zoo.

In the southwestern United States, AZA institutions participate in government-led ranid conservation projects. In the mountains of California, the Critically Endangered Mountain Yellow-legged Frog (*Rana muscosa*) has a limited and disjointed range (Fig. 5). Introduced trout have long been a threat to the eggs and tadpoles of this species in parts of its range and the removal of the trout has led to growth in populations of the frogs (Knapp and Matthews 2000; Vredenburg 2004). As a result, the U.S. Geological Survey (USGS) and the FWS developed a plan to remove these predators from the Angeles National Forest north of Los Angeles and zookeepers from the Los Angeles Zoo and Fresno Chaffee Zoo provided assistance in the field. The San Diego Zoo's center for Conservation and Research for Endangered Species (CRES) is currently safeguarding frogs rescued by government agencies from drying streambed pools in the San Jacinto Mountains. Once drought conditions have passed, zoo staff will work with state and federal agencies to return these individuals to their natural habitat (Bradley, *in press*).

The Santa Barbara Zoo works with the U.S. Forest Service (USFS) to survey streams in the Los Padres National Forest for California Red-legged Frogs (*Rana draytonii*) and the California Arroyo Toad (*Anaxyrus californicus*). In addition to surveying and recording ambient conditions and developmental stages when egg masses are located, zoo staff place exclosures around the mass to prevent aquatic predators, especially the introduced Bullfrogs (*Lithobates catesbeianus*)[3] and Mosquitofish (*Gambusia affinis*), from consuming them (Foster et al. 2007). The Zoo is currently developing an exhibit that will feature reptiles and amphibians native to the Los Padres National Forest, and estimate that up to 450,000 visitors annually will learn about local herpetofaunal con-

FIG. 4. Spiny-headed Treefrog (*Anotheca spinosa*) is one of several imperiled species reproducing at the El Valle Amphibian Conservation Center (Gagliardo et al., *in press*). Photo courtesy of John Kast, Fort Worth Zoo.

FIG. 5. Several zoos and aquariums are supporting government-led recovery efforts on behalf of the Mountain Yellow-legged Frog (*Rana muscosa*) in the western United States. Photo courtesy of Jeff Lemm, San Diego Zoo's Center for Conservation and Research for Endangered Species (CRES).

servation through this exhibit, in addition to visitors that hear about these efforts through zookeeper talks and presentations, signage at the Zoo, and through the Zoo's newsletter (Foster et al. 2007).

In Arizona and New Mexico, ranid conservation at AZA facilities includes leopard and Tarahumara frogs. The Phoenix Zoo has been part of the Ramsey Canyon Leopard Frog Conservation Team (RCLFCT), a partnership of state and federal agencies and private groups for ten years. The Arizona-Sonora Desert Museum (ASDM) is also part of the Team. The Team's goal is to halt population declines of the Ramsey Canyon Leopard Frog (*Rana chiricahuensis*)[4]. Because of high mortality rates in the wild for eggs and small tadpoles, the team decided to rear egg masses collected in the wild at the zoo and museum, and delayed release until they had developed into large tadpoles or small frogs, life stages with higher survival rates. As a result of these reintroductions and translocations, the number of ponds where Ramsey Canyon Leopard Frogs have become or are becoming established has increased four-fold (Sprankle, *in press*). The Fort Worth Zoo headstarts Chiricahua Leopard Frogs (*R. chiricahuensis* from locales other than Ramsey Canyon) in close collaboration with the FWS, New Mexico Game and Fish Department, Arizona Game and Fish Department, and Turner Endangered Species Fund (Garrett 2006). Adult populations are brought to the Zoo for breeding purposes and the resulting tadpoles are returned to the field for repatriation.

ASDM also works with Tarahumara Frogs (*Lithobates tarahumarae*)[5]. The Tarahumara Frog's historical range included southern Arizona and northwest Mexico, but the last US Tarahumara Frog was found dead in Arizona in 1983; at least three of the last local extirpations were a result of *Bd* (Hale et al. 2005). Collaborative efforts involving two nations, two states, numerous state and federal agencies, universities, private landowners, and interested citizens have led to the collection of founder individuals in Mexico, captive rearing of these founders at ASDM, surveys of historical habitat, assessment of population responses to *Bd,* and in 2004–2006, reintroductions of the Tarahumara Frog in Arizona. While re-establishment of the species in the United States continues to face many challenges, so far, the animals are persisting and breeding in Arizona once again (Johnson 2007).

AZA efforts are not focused exclusively on frogs and toads. In the central and eastern United States, survey evidence suggests that Hellbender salamander populations of both subspecies (*Cryptobranchus alleganiensis alleganiensis* and *C. a. bishopi*) are declining throughout their ranges in the U.S. Southeast and Appalachian regions. Surveys from Missouri and Ohio suggest population declines of ca. 80% over the past few decades and a skewing of populations towards older and larger animals, indicating low recruitment into the populations (Briggler et al. 2007; Lipps 2007). Staff and volunteers from zoos and aquariums throughout the salamander's range have begun surveying and monitoring local populations in the wild. The North Carolina Zoo works with the state's Wildlife Resource Commission to monitor populations and collect water quality and population data, and in 2007 received CEF funds to expand this effort. In Ohio, AZA member The Wilds is creating a stream on-grounds to mimic Hellbender habitat with the hope of eventually housing Hellbenders and studying the reasons behind their decline. While captive animals have not yet reproduced in captivity, in October 2007, a female Hellbender laid eggs at an indoor facility at the Saint Louis Zoo. The males did not fertilize the clutch, but this landmark event was significant to those developing protocols for rearing this species in captivity. The Columbus Zoo and Aquarium, Fort Worth Zoo, Lincoln Park Zoo, Louisville Zoo, Nashville Zoo, National Aquarium in Baltimore, Oglebay's Good Zoo, Omaha's Henry Doorly Zoo, Pittsburgh Zoo and PPG Aquarium, Smithsonian National Zoological Park, and Wonders of Wildlife are all involved in Hellbender conservation as well.

Further north in Canada, the Toronto Zoo has taken an ecosystem approach to amphibian conservation. In partnership with Environment Canada, the Toronto Zoo developed an Adopt-A-Pond program in 1991 (Sealock 2005). The goal of this program is to address declining amphibian populations by providing teachers, students, and community groups with informational resources and educational opportunities to conserve, restore, and create wetland habitats. Through its FrogWatch program, people of all ages are taught to monitor local frog populations by listening for their calls and reporting their observational data to a central database. Their publication *The Urban Outback* (http://www.torontozoo.com/adoptapond/) provides technical information to homeowners for small-scale wetland restoration and includes sections dedicated to amphibians. Full curricula and complementary resource networks, such as the Zoo's Pond Guardian initiative, have been developed around these successful programs. The Toronto Zoo is a leader in promoting backyard conservation and putting international conservation issues into a familiar and local context.

Finally, throughout the AZA community, facilities are committing millions of dollars to expand their amphibian conservation capacity. The largest of these efforts will take place at the Fort Worth Zoo, where leaders have committed $18 million to build a new herpetarium. The new facility, dubbed Museum of Living Art (MOLA), will include spacious, humidity-controlled exhibits and state-of-the-art conservation tools, such as quarantine rooms and hibernacula that support the breeding of rare, endangered, and critically endangered amphibians. Both the Jacksonville Zoo and Toledo Zoo are utilizing existing space at their facilities to create additional amphibian conservation and exhibit space, while Omaha's Henry Doorly Zoo is increasing their amount of biosecure space to accommodate increasing numbers of endangered and critically endangered amphibian species.

GREATER THAN THE SUM OF ITS PARTS: AN UNPRECEDENTED CAMPAIGN

2008: Year of the Frog is a campaign unlike any other in the zoological community. Participation in the campaign is global. The campaign responds directly to an urgent conservation need. It focuses on a taxonomic group not generally given the spotlight at zoological facilities. It is fully integrated to include herpetologists, zoological veterinarians and epidemiologists, nutritionists and invertebrate specialists, small population biologists, educators, public relations departments, and marketers. It promises to raise public awareness so that 600 million annual zoo visitors worldwide understand and support amphibian conservation. The Year of the Frog campaign will raise money and attention so these programs can continue to grow in the future, even if the spotlight may shift to other taxonomic groups. And it promises to fulfill the *ex situ* recommendations outlined in the IUCN's *Amphibian Conservation Action Plan* (ACAP). The implementation of the campaign will also provide support and participation to other components of the ACAP, making this campaign vital to the overall success of the ACAP.

This campaign is larger than the zoological community, and appropriately so. To halt amphibian declines, commitments and action must also be larger than the zoological community or any other single organization or interest group. Join us in this campaign and work together with your local zoos and aquariums to help this effort be unprecedented in its integration and ultimately, unprecedented in its success.

Guide to Acronyms

ACAP – Amphibian Conservation Action Plan
ARAZPA – Australasian Regional Association of Zoological Parks and Aquaria
ASG – Amphibian Specialist Group
ASDM – Arizona-Sonora Desert Museum
ATAG – Amphibian Taxon Advisory Group
AZA – Association of Zoos and Aquariums
Bd - Batrachochytrium dendrobatidis
CBSG – Conservation Breeding Specialist Group
CEC – Conservation Education Committee
CEF – Conservation Endowment Fund
CRES – Conservation and Research for Endangered Species
DAPTF – Declining Amphibian Populations Task Force
EAZA – European Association of Zoos and Aquariums
EWCL –Emerging Wildlife Conservation Leaders
FWS – U.S. Fish and Wildlife Service
GAA – Global Amphibian Assessment
IUCN – World Conservation Union
MOLA – Museum of Living Art
NZP – National Zoological Park
PARC – Partners in Amphibian and Reptile Conservation
RCLFCT – Ramsey Canyon Leopard Frog Conservation Team
SSC – Species Survival Commission
SSP – Species Survival Plan
USFS – U.S. Forest Service
USGS – U.S. Geological Survey
WAZA – World Association of Zoos and Aquariums
ZCOG – Zoo Conservation Outreach Group

Referenced and Related Web Sites

Amphibian Ark: www.amphibianark.org
AmphibiaWeb: http://amphibiaweb.org/index.html
AZA: www.aza.org; www.aza.org/yearofthefrog
Global Amphibian Assessment: www.globalamphibians.org
IUCN Red List of Threatened Species: http://www.iucnredlist.org/
Toronto Zoo: http://www.torontozoo.com/adoptapond/

Acknowledgments.—We wish to thank Diane Barber (Fort Worth Zoo), Lesley Dickie (Amphibian Ark/Zoological Society of London), Kris Vehrs, Paul Boyle, and Steve Feldman (AZA), and James Murphy (Smithsonian National Zoological Park) for their assistance with the development of this manuscript. We would also like to thank John Kast (Fort Worth Zoo), Bill Konstant (Houston Zoo), Jeff Lemm (San Diego Zoo's CRES), and R. Andrew Odum (Toledo Zoo) for the use of their photos and Jennifer Pramuk (Bronx Zoo) for her help with taxonomic questions. Finally, our gratitude goes out to all of the global zoos and aquariums involved in amphibian conservation, including the many facilities whose efforts were not mentioned specifically in this article.

Literature Cited

Berger, L., R. Speare, P. Daszak, D. Green, A. Cunningham, C. Goggin, R. Slocombe, M. Ragan, A. Hyatt, K. McDonald, H. Hines, K. Lips, G. Marantelli, and H. Parkes. 1998. Chytridiomycosis causes amphibian mortality associated with population declines in the rain forests of Australia and Central America. Proc. Natl. Acad. Sci. USA 95:9031–9036.

Bradley, A. *In press*. One Giant Leap: Reviving the Mountain Yellow-legged Frog. U.S. Fish Wildlife Serv. Endangered Species Bull.

Briggler, J., M. Wanner, and C. Schuette. 2007. Hellbender declines and recovery actions in Missouri, with emphasis on chytridiomycosis. Presented at the "Amphibian Decline and Chytridiomycosis: Translating Science into Urgent Action" symposium. (6 November). Tempe, Arizona.

Crump, P. and S. Grow. 2007. Action plan for *ex situ* amphibian conservation in the AZA community. Association of Zoos and Aquariums. Silver Spring, Maryland. 46 pp. http://www.aza.org.

Dubois, A. 2006. New proposals for naming lower-ranked taxa within the frame of the International Code of Zoological Nomenclature. C. R. Biol. 329:823–840.

Foster, C. D., J. Traverse, P. Martin, A. Varsik, and E. Sandhaus. 2007. Anuran conservation through collaborations: Santa Barbara Zoo teams up with the U.S. Forest Service. Herpetol. Rev. 38:141–142.

Frost, D. R. 2007. Amphibian Species of the World: an online reference. Version 5.1 (9 October 2007). American Museum of Natural History, New York, New York. Electronic Database accessible at http://research.amnh.org/herpetology/amphibia/index.php.

Gagliardo, R., P. Crump, E. Griffith, J. Mendelson, H. Ross, and K. Zippel. *In press*. The principles of rapid response for amphibian conservation, using the programmes in Panama as an example. Inter. Zoo. Yearb.

Garrett, C. M. 2006. Herpetology at the Fort Worth Zoo: a 45-year history. Herpetol. Rev. 37:264–268.

Gascon, C., J. P. Collins, R. D. Moore, D. R. Church, J. E. McKay, and J. R. Mendelson III (eds.). 2007. Amphibian Conservation Action Plan. IUCN/SSC Amphibian Specialist Group. Gland, Switzerland and Cambridge UK. 64 pp.

Gillespie, G., R. Traher, and C. Banks. 2007. ARAZPA Amphibian Action Plan. Australasian Regional Association of Zoological Parks and Aquaria. 33 pp.

Goldberg, C. S., K. J. Field, and M. J. Sredl. 2004. Mitochondrial DNA sequences do not support species status of the Ramsey Canyon leopard frog (*Rana subaquavocalis*). J. Herpetol. 38:313–319.

Grow, S., and V. Poole (Eds.). 2007. Amphibian Conservation Resource Manual. Association of Zoos and Aquariums. Silver Spring, Maryland. 208 pp. http://www.aza.org.

Hale, S. F., P. C. Rosen, J. L. Jarchow, and G. A. Bradley. 2005. Effects of the chytrid fungus on the Tarahumara frog (*Rana tarahumarae*) in Arizona and Sonora, Mexico. *In* G. J. Gottfried, B. S. Gebow, L. G. Eskew, and C. B. Edminster (eds.), Connecting Mountain Islands and Desert Seas: Biodiversity and Management of the Madrean Archipelago II, pp. 407–441. Proc. RMRS-P-36. Fort Collins, Colorado, U.S. Department of Agriculture, Forest Service, Rocky Mountain Research Station.

Heyer, W. R., and J. B. Murphy. 2005. Declining Amphibian Populations Task Force. *In* M. J. Lannoo (ed.), Amphibian Declines: The Conservation Status of United States Species, pp. 17–21. University of California Press, Berkeley and Los Angeles, California.

Hillis, D. M., and T. P. Wilcox. 2005. Phylogeny of New World true frogs (*Rana*). Mol. Phylogen. Evol. 34:299–314.

IUCN, Conservation International, and NatureServe. 2006. Global Amphibian Assessment. http://www.globalamphibians.org.

Johnson, J. P. 2007. Herpetology at the Arizona-Sonora Desert Museum. Herpetol. Rev. 38:7–12.

Johnson, R. R. 1994. Model programs for reproduction and management: *ex situ* and *in situ* conservation of toads of the family Bufonidae. *In* J. B. Murphy, K. Adler and J. T. Collins (eds.), Captive Management and Conservation of Amphibians and Reptiles, pp. 243–254. SSAR Contrib. Herpetol. Vol. 11, Ithaca, New York.

Knapp, R. A., and K. R. Matthews. 2000. Non-native fish introductions and the decline of the mountain yellow-legged frog from within protected areas. Conserv. Biol. 14:428–438.

Krajick, K. 2006. The lost world of the Kihansi toad. Science 311:1230–1232.

Lipps, G. 2007. Unpublished data presented at the "Ohio Zoo & Aquarium Meeting" at Columbus Zoo. (13 November). Columbus, Ohio.

Lips, K. R., P. A. Burrowes, J. R. Mendelson, and G. Parra-Olea. 2005. Amphibian population declines in Latin America: A synthesis. Biotropica 37:222–226.

———, F. Brem, R. Brenes, J. D. Reeve, R. A. Alford, J. Voyles, C. Carey, L. Livo, A. P. Pessier, and J. P. Collins. 2006. From the cover: Emerging infectious disease and the loss of biodiversity in a Neotropical amphibian community. Proc. Natl. Acad. Sci. USA 103:3165–3170.

Longcore, J. E., A. P. Pessier, and D. K. Nichols. 1999. *Batrachochytrium dendrobatidis* gen. et. sp. nov., a chytrid pathogenic to amphibians. Mycologia 91:219–227.

Mendelson, J. R., III, and R. W. Gagliardo. 2005. Atlanta institutions lead the way to save amphibians. Wild Times, Zoo Atlanta, Atlanta, Georgia. Fall 2005:3–5.

———, K. R. Lips, R. W. Gagliardo, G. B. Rabb, J. P. Collins, J. E. Diffendorfer, P. Daszak, R.Ibáñez, K. C. Zippel, D. P. Lawson, K. M. Wright, S. N. Stuart, C. Gascon, H. R. da Silva, P. A. Burrowes, R. L. Joglar, E. La Marca, S. Lotters, L. H. du Preez, C. Weldon, A. Hyatt, J. V. Rodriguez-Mahecha, S. Hunt, H. Robertson, B. Lock, C. J. Raxworthy, D. R. Frost, R. C. Lacy, R. A. Alford, J. A. Campbell, G. Parra-Olea, F. Bolaños, J. J. Calvo Domingo, T. Halliday, J. B. Murphy, M. H. Wake, L. A. Columa, S. L. Kuzmin, M. S. Price, K. M. Howell, M. Lau, R. Pethiyagoda, M. Boone, M. J. Lannoo, A. R. Blaustein, A. Dobson, R. A. Griffiths, M. L. Crump, D. B. Wake, and E. D. Brodie. 2006. Confronting amphibian declines and extinctions. Science. 313:48.

Nichols, D. K. 2003. Tracking down the killer chytrid of amphibians. Herpetol. Rev. 34:101–104.

Norris, S. 2007. Ghosts in our midst: Coming to terms with amphibian extinctions. BioScience 57:311–316.

Odum, R. A., and P. S. Corn. 2005 *Bufo baxteri* Porter, 1968. *In* M. J. Lannoo (ed.), Amphibian Declines: The Conservation Status of United States Species, pp. 390–392. University of California Press, Berkeley, California.

Poole, V. A., and S. Grow (Eds.). 2007. Amphibian Husbandry Resource Guide. Association of Zoos and Aquariums. Silver Spring, Maryland. 86 pp. http://www.aza.org.

Pounds, J. A., A. C. Carnaval, R. Puschendorf, C. F. B. Haddad, and K. L. Masters. 2007. Action on amphibian extinctions: going beyond the reductive. Science (E-Letter, 28 August 2007) http://www.sciencemag.org/cgi/eletters/313/5783/48#9870.

Platz, J. E. 1993. *Rana subaquavocalis*, a remarkable new species of leopard frog (*Rana pipiens* complex) from southeastern Arizona that calls under water. J. Herpetol. 27:154–162.

Sealock, L. 2005. Protecting urban biodiversity: The Toronto Zoo's Adopt-A-Pond program. AZA Communiqué, Association of Zoos and Aquariums. May:16

Sprankle, T. *In press*. Giving native leopard frogs a head-start on survival. U.S. Fish Wildlife Serv. Endangered Species Bull.

Stebbins, R. C., and N. W. Cohen. 1995. A Natural History of Amphibians. Princeton University Press, Princeton, New Jersey.

Stuart, S. N., J. S. Chanson, N. A. Cox, B. E. Young, A. S. L. Rodrigues, D. L. Fischman, and R. W. Waller. 2004. Status and trends of amphibian declines and extinctions worldwide. Science 306:1783–1786.

U.S. Fish and Wildlife Service. 2007. Chiricahua Leopard Frog (*Rana chiricahuensis*) Recovery Plan. U.S. Fish and Wildlife Service, Southwest Region, Albuquerque, New Mexico. 149 pp. + appendices A–M.

Vredenburg, V. T. 2004. Reversing introduced species effects: Experimental removal of introduced fish leads to rapid recovery of a declining frog. Proc. Natl. Acad. Sci. USA 101:7646–7650.

Wiese, R. J., and M. Hutchins. 1994. The role of zoos and aquariums in amphibian and reptilian conservation. *In* J. B. Murphy, K. Adler, and J. T. Collins (eds.), Captive Management and Conservation of Amphibians and Reptiles, pp. 37–45. SSAR Contrib. Herpetol. Vol. 11, Ithaca, New York.

Young, S., L. Berger, and R. Speare. 2007. Amphibian chytridiomycosis: strategies for captive management and conservation. Inter. Zoo Yearb. 41:85–95.

Zippel, K. 2002. Conserving the Panamanian golden frog: Proyecto Rana Dorada. Herpetol. Rev. 33:11–12.

———. 2007a. Why do we need an Amphibian Ark? Actionbioscience. http://www.actionbioscience.org/biodiversity/zippel.html.

———. 2007b. Zoos play a vital role in amphibian conservation. Berkeley, California: AmphibiaWeb. http://amphibiaweb.org.

[1] The taxonomy of *Bufo baxteri* has been updated to *Anaxyrus baxteri*, as reflected in Frost (2007).

[2] The taxonomy of *Bufo californicus* has been updated to *Anaxyrus californicus,* as reflected in Frost (2007).

[3] The taxonomy of *Rana catesbeiana* has been updated to *Lithobates catesbeianus* as a result of the work of Dubois (2006), as reflected in Frost (2007).

[4] The Ramsey Canyon Leopard Frog was described as *Rana subaquavocalis* by Platz (1993), however, the U.S. Fish and Wildlife Service has adopted the taxonomy of *R. chiricahuensis* due to genetic work by Goldberg et al. (2004) and Hillis and Wilcox (2005) that supports subsuming *R. subaquavocalis* into *R. chiricahuensis* (U.S. Fish and Wildlife Service 2007).

[5] The taxonomy of *Rana tarahumarae* has been updated to *Lithobates tarahumarae* as a result of the work of Dubois (2006), as reflected in Frost (2007).

LETTERS TO THE EDITOR

Herpetological Review, 2008, 39(1), 23–29.

The Amphibian Extinction Crisis: A Call To Action

KEVIN C. ZIPPEL
Amphibian Ark/CBSG, 12101 Johnny Cake Ridge Road
Apple Valley, Minnesota 55124, USA
e-mail: KevinZ@AmphibianArk.org

and

JOSEPH R. MENDELSON III
Zoo Atlanta, 800 Cherokee Avenue
Atlanta, Georgia 30315, USA
e-mail: jmendelson@zooatlanta.org

The Global Crisis

Addressing the amphibian extinction crisis curently represents one of the greatest species conservation challenge in the history of humanity. The Global Amphibian Assessment (Stuart et al. 2004) revealed that one third to one half of the world's 6000 amphibian species are threatened with extinction. This is significantly more than any other group of organisms: by comparison, 12% of bird species and 23% of mammal species are threatened. As many as 165 amphibian species have already disappeared in recent years. This qualifies as a mass extinction event, and deserves special attention for reasons of ethics and also because amphibians deliver substantial goods and services to their ecosystems and to humans (e.g., biomedicines). The amphibian crisis has taught us a valuable lesson in that 20 years of monitoring, research, and conservation action have advanced our state of knowledge of the situation, but these efforts have failed to prevent numerous extinctions in places such as Australia, Puerto Rico, Costa Rica, and Ecuador. This sends the message that "business as usual" in terms of research and conservation, while remaining crucial, are insufficient by themselves to confront this crisis (Mendelson et al. 2006).

The Amphibian Conservation Action Plan

The myriad intertwined factors likely affecting amphibian declines and extinctions require novel, cross-disciplinary approaches to research and conservation. In September 2005, the IUCN and Conservation International convened an Amphibian Conservation Summit in Washington, DC. The world's amphibian authorities from academia, zoos, government, veterinary medicine, and other diverse disciplines gathered to conceptualize a global Amphibian Conservation Action Plan (ACAP, Gascon et al. 2007), outlining general responses required in the fields of research, assessment, conservation, and rapid responses to stem widespread global amphibian extinctions. The entire ACAP may be downloaded from the website of the IUCN-SSC Amphibian Specialist Group (www.amphibians.org).

The ACAP emphasizes basic research targeted at the underlying causes of amphibian declines, specifically highlighting diseases, climate change, and environmental contamination. The ACAP calls for aggressive field efforts to refine our knowledge of amphibian diversity (phylogeny and taxonomy), distributions, and conservation status, in order to continuously update the Global Amphibian Assessment (www.globalamphibians.org). Conservation programs must address important long-term objectives in habitat protection, while also managing the difficult arenas of reintroductions and control of harvest. Finally, the ACAP delegation agreed that there are a number of examples in the wild where rapid-response interventions might be necessary. Such programs are to include capacity for responding to impending crises related to habitat loss, contamination, or disease outbreak, and also establishing a globally organized program to maintain captive survival assurance populations. Captive populations may represent short- or long-

The Lemur Leaf Frog *Hylomantis* (= *Phyllomedusa*) *lemur* is listed by IUCN as Endangered across its entire range in Costa Rica, Panama, and Colombia. Its current status in the wild appears to continue to decline as populations in Panama are being sequentially impacted by epidemics of amphibian chytridiomycosis. Following guidelines laid out by the Amphibian Conservation Action Plan and the Amphibian Ark, this species is currently well established in ex-situ populations in USA, UK, Costa Rica, and Panama and in-situ efforts are ongoing in Costa Rica. Progeny from the captive colonies are now forming the basis for basic academic studies on chytridiomycosis. Photo by Ron Holt/Courtesy Atlanta Botanical Garden.

term temporary solutions to prevent extinctions, while necessary research and conservation programs work to address the primary threats. They also should represent phylogenetically diverse groups for important research programs (e.g., evolution of resistance to emerging infectious diseases). These captive programs fall under the auspices of the newly launched Amphibian Ark (Pavajeau et al., *in press*; see below).

Despite substantial progress in recent years in understanding the underlying causes of the amphibian crisis, significant research gaps remain. Examples include:

- what are the effects of long-term sub-lethal exposure to environmental contaminants?
- are there direct cause-effect linkages between climate and disease?
- how exactly does chytridiomycosis kill amphibians?
- how and why are some species more affected by certain threats than are others?
- how is chytridiomycosis spread, and does the pathogen have a resting stage?
- can we design bioreserves to protect against invasive species, emerging infectious diseases, contaminants, and climate change?

These projects, among many others, will require significant time, effort, and resources before their results can be applied in any meaningful way to conserve amphibians. These questions will also require truly cross-disciplinary approaches, if we are to have any hope of identifying and responding to the potential synergistic effects between the various documented threats to amphibians. Using the ACAP as an outline, the specific role of the IUCN/SSC Amphibian Specialist Group is to support a holistic approach to the amphibian crisis that includes complementary efforts among *in situ* and *ex situ* conservation programs, basic research, and related issues of policy, law, ethics, and public education.

A Call to Action for Conservation Research

Academic institutions are the centers for much of the basic research necessary to inform conservation programs. Never has the connection between basic research and conservation been more imperative than in the present amphibian crisis. Mendelson et al. (2006) opined that the status quo approach of programs of research-as-usual and conservation-as-usual are insufficient to confront this crisis. The basic problem in this case is that conservationists, land-managers, and other stakeholders literally do not know how to implement conservation programs in the face of threats as global and pervasive as emerging infectious disease and climate change. Thus, synthetic programs must be coordinated among the research institutions globally, in order to discover the science underlying the current threats to amphibian biodiversity. This research must take place against a backdrop of continued vigilance of programs to protect habitat, prevent overexploitation, and other "familiar" threats to biodiversity. However, basic research takes time, and recent history has demonstrated that the passage of time correlates with additional amphibian extinctions. Thus, while researchers work to answer basic questions relevant to amphibian conservation (e.g., "Does amphibian chytrid fungus persist in the environment, in the absence of amphibians?") their results may come too late to save some species. Thus the clarion call was sounded to bolster such research programs contemporaneously with ambitious conservation programs (Gascon et al. 2007; Mendelson et al. 2006).

There have been a number of world-class amphibian research programs and facilities at universities and museums (e.g., Kluge 1996; Myers 2000; Trueb and Duellman 1996) that have framed our understanding of amphibian biology, and also our fundamental skills for maintaining and breeding certain species in captivity (e.g., Cullum and Justus 1973; Nace 1968; Nace et al. 1974). Nevertheless, these programs historically have tended to function in isolation from ongoing conservation programs—in fact, the focus was never on conservation. Fortunately, however, the recent call for integrated research and conservation programs comes as some such programs already exist (see below).

Model Programs

Environmental Contaminants

MICHELLE BOONE
Miami University, e-mail: boonemd@muohio.edu

Contaminants are both accidentally and purposefully released into the environment and can move through local, regional, and global cycles that result in widespread chemical exposure in both protected and human-dominated landscapes. Therefore, understanding how contaminants can influence population dynamics and community regulation is of great importance, particularly in light of worldwide amphibian population declines. Additionally, contaminants could also play a key role in amphibian declines by interacting with other stressors present in the environment, such as disease pathogens. While traditionally, toxicology has focused on lethal concentrations in simple systems in the laboratory, it is critical to integrate effects found in the laboratory with those in the field to gain better predictive power for population and community level consequences of contaminants in nature.

A group of scientists working in the field of ecotoxicology has been examining the effects of contaminants from the laboratory to field mesocosms to artificial wetlands to the natural environment, which has allowed us to explore the ecological ramifications of chemical exposure to the aquatic and terrestrial environment (reviewed in Boone and James 2005). For instance, our work with expected environmental concentrations of the insecticide carbaryl has allowed us to examine how carbaryl's toxicity was influenced by competition, predation, and pond hydroperiod across experimental venues (reviewed in Boone and Bridges 2003; Boone et al. 2004). These studies are among the first to examine how natural environmental factors can alter toxicity. From initial studies, it became clear that environmental factors influence and modify the effect of a contaminant on amphibians, that species are differentially susceptible to contaminants, and that the indirect effects of the contaminants (e.g., altering predator communities and food resources) are potentially more important than direct effects (Boone and Bridges 2003; Mills and Semlitsch 2004). We have also manipulated factors associated with chemical application, such as multiple contaminant exposures through larval development (reviewed in Boone and Bridges 2003) and exposure to chemical mixtures (reviewed in Boone and James 2005; Boone et al. 2007). These studies suggest that mimicking contaminant application through multiple exposures during larval development can stimulate larval development and lead to precocious metamorphosis.

Additionally, multiple contaminants can result in interactions that are not predictable from single factor studies. In this respect, these studies have directly addressed the question of "multiple stressors" and set the stage for evaluating ecological questions related to contaminants in natural environments that other researchers have and could follow.

The benefit of an approach evaluating effects of select contaminants in complex experimental environments (rather than broader testing of different contaminants in the simple laboratory or field conditions) is that it addresses relevant ecological questions and evaluates how contaminant effects may change or manifest themselves outside of traditional types of tests used for federal regulation. Currently, we do not understand the ecological effects of the major classes of chemical contaminants and we are only beginning to explore how other factors mediate the contaminants' effects. Although there is evidence suggesting that contaminants may play a role in amphibian population declines (e.g., Davidson et al. 2002), the mechanism is not clear. There is accumulating evidence indicating that contaminants play a role in the incidence of limb malformation through increased susceptibility to trematodes (Johnson and Chase 2004) and can decrease immune response (Christin et al. 2003; Gilbertson et al. 2003). Such studies suggest that presence of contaminants in combination with other factors could contribute to declines. Contaminants are present in the environment, and for this reason evaluating their effects, which can alter community structure and population dynamics, is of paramount importance.

Diseases

KAREN R. LIPS
Southern Illinois University, e-mail: klips@zoology.siu.edu

JOSEPH R. MENDELSON III
Zoo Atlanta, e-mail: jmendelson@zooatlanta.org

Infectious disease is now acknowledged as a major threat to amphibian populations and species at a growing number of sites around the world (Daszak et al. 2003; Stuart et al. 2004; Lips et al. 2006; Ron 2005). While amphibian chytridiomycosis has been the focus of the majority of investigations in recent years, other pathogens (e.g., ranaviruses) must also be considered (see review by Daszak et al. 2003). Along with studies related to climate change (see below), it appears that the relative novelty of disease as a direct threat to biodiversity has effectively instigated a number of innovative collaborative projects that represent basic research related to disease or pathogens, but within an explicit framework of amphibian conservation (e.g., Carey et al. 2006; Harris et al. 2006; Kriger and Hero 2007; Lips et al. 2006; Rachowicz and Vredenburg 2004; Rollins-Smith et al. 2002; Wilcox 2006; Woodhams et al. 2007). Furthermore, cross-disciplinary research and conservation programs are coming to exist, such as the Endangered Frog Research Centre at Griffith University, the Amphibian Diseases Research Group at James Cook University (both in Queensland, Australia), the Integrated Research and Challenges in Environmental Biology amphibian disease group (based at Arizona State University, USA), the Latin American RANA organization, and the recently formed international Amphibian Ecotoxicology Consortium. While many more such projects and programs are needed, the evident synthesis of research and conservation priorities does bode well for amphibian biodiversity.

A key to the innovative nature and ultimate productivity of such cross-disciplinary programs is their culture of meeting and collaborating, rather than the more traditional approach of research labs around the world working independently and thus competing with one another, whether it be implicitly or explicitly so. Within such networks graduate students, for example, may have open access to expertise that that is beyond the scope of their base institution or advisory committee. This type of access to advice and training effectively ensures future generations of researchers with a broad array of skills, professional experience, and key contacts across disciplines. Similarly, international collaborations build a culture of trust that can simplify layers of bureaucracy, to allow better focus on the science at hand and improve diplomatic relations among academics across regions.

Climate Change

SANTIAGO R. RON
Museo de Zoología, Pontificia Universidad Católica del Ecuador
e-mail: srron@puce.edu.ec

An increasing number of studies show a correlation between amphibian population declines and climate change (e.g., Blaustein et al. 2003; Pounds et al. 2006). Climate change could produce population declines either directly (e.g., by reducing the area of distribution of the species' fundamental niche; Thomas et al. 2004) or indirectly by interacting with other risk factors. The most widely studied hypothesis for the role of climate change is known as the climate-linked epidemic hypothesis (Harvell et al. 2002; Pounds et al. 2006), which posits that declines are a consequence of synergisms between pathogens and novel climate conditions. A recently proposed variant is the chytrid thermal optimum hypothesis, which posits a link between climate change and population declines attributable to chytridiomycosis (Bosch et al. 2006; Pounds et al. 2006).

The available evidence suggests that research on the effects of climate change could be crucial to develop realistic conservation programs for amphibians. Future efforts should be directed to explore the link, if any, between climate change and population declines in a more diverse array of taxa and on a larger number of regions with emphasis in biodiversity hotspots. Because the association between climate change and disease does not necessarily imply causation, there is an urgent need to test the climate-linked epidemic hypothesis by acquiring a better understanding of how climate influences the immune response of amphibians and the survival rates, virulence, and transmission of pathogens. Climate change has been linked to disease outbreaks involving more than one pathogen at a single population (Di Rosa et al. 2007) suggesting that research should test interactions with a broad array of potential pathogenic agents.

Although there are numerous studies documenting distributional shifts as a the result of climate change (for a review see Parmesan 2006), there are surprisingly few published reports for amphibians (e.g., Seimon et al. 2007). It is expected that species with restricted ranges inhabiting polar regions and mountaintops will experience severe range contractions, population declines and extinctions (Parmesan 2006). Thus, future efforts should also focus on assessing how climate change is impacting distribution ranges, especially in tropical mountains, the regions of greatest amphibian diversity.

Global warming is unlikely to weaken in the short term. A more complete baseline dataset on how climate influences disease spread and outbreaks will allow development of predictive models to forecast regions and taxa that are likely to become threatened by climate change.

The Amphibian Ark

Although the only place to truly conserve wildlife is in the wild, *in-situ* threats sometimes cannot be mitigated quickly enough to prevent extirpation or extinction. Even with timely protection from threats under our immediate control, such as habitat destruction and over-collection, we cannot protect wild populations from threats whose effects would continue long after the immediate cause has ceased, e.g., introduced diseases, climate change, and pollution. In cases where *in-situ* threats cannot be mitigated quickly, as appears to be the case for many of amphibian species, *ex-situ* intervention is the only option to prevent immediate extinction. The IUCN has urged that "All Critically Endangered and Extinct in the Wild taxa should be subject to *ex situ* management to ensure recovery of wild populations." (IUCN 2002). Comparable calls to action are included in the Global Amphibian Assessment, the ACAP, and other IUCN documents. In response to this challenge, the World Association of Zoos and Aquariums, the IUCN/SSC Conservation Breeding Specialist Group, and the IUCN/SSC Amphibian Specialist Group have jointly formed the Amphibian Ark (www.AmphibianArk.org, Pavajeau et al. in press), to guide short-term *ex situ* management to help ensure long-term survival in nature of amphibian taxa for which adequate protection in the wild is not currently possible. The AArk will facilitate coordination of *ex situ* programs implemented by partners around the world, with the first emphasis on programs within the range countries of the species, and with a constant attention to our obligation to link *ex situ* conservation measures with complementary *in situ* programs. The AArk will lead a 2008 globally coordinated, zoo-based awareness and capital campaign, 'Year of the Frog' (www.2008YearoftheFrog.org) to help zoos raise the funds to support rapidly developing programs for *ex situ* rescue efforts, *in situ* conservation, research, and communication about the amphibian extinction crisis. The goal of AArk is to prevent the anticipated extinction of hundreds of species (*sensu* Stuart et al. 2004), and to buy time for these species while ongoing basic research and conservation programs develop effective long-term measures to restore and protect populations in the wild. The program will accomplish this goal primarily by developing capacity globally to provide amphibian species with care and protection when needed, and by forming effective partnerships between *ex situ* and *in situ* components of conservation. This framework will provide a model for responding to future species conservation crises.

A Call to Action for Conservation Breeding

Ex situ management for conservation is not just a job for zoos, aquariums, and botanic gardens. Currently there is not enough capacity among these institutions to meet the needs of most of the amphibian species that require help. We estimate that the global zoo community can currently manage viable populations of ~50 amphibian species, which amounts to perhaps 10% of those requiring *ex-situ* intervention. If we are to understand the needs and requirements of the diverse amphibian species that will need help, and which by the very nature of their vulnerability are likely to be specialized, it will be important to acknowledge the valuable contribution academic institutions and individuals can provide. In an effort to increase global capacity, and also harness the skills of such specialists, the AArk must include the expertise and facilities of universities, natural history museums, and the private sector. Several existing programs serve as models in this arena.

Model Programs

Pontificia Universidad Católica in Quito, Ecuador

LUIS A. COLOMA
e-mail: lcoloma@puce.edu.ec

The Ecuadorian amphibian fauna comprise 464 described species, plus an estimated 270 awaiting discovery and/or description (Coloma et al. 2007). This fauna is not only one of the most diverse on the planet, but one the most threatened. Threats such as global warming, pathogens, and habitat destruction are magnified in this country. For example, mean temperatures in the Andes are increasing at least twice as fast as the mean global temperature (Merino-Viteri et al. 2005; Ron et al. 2003), pathogens are widespread in the Andean region (Merino-Viteri et al. 2005; Ron and Merino 2000), and the national deforestation rate (about half a million acres per year) is among the tenth highest in the world (CLIRSEN 2006). Most recent estimates indicate that at least 141 Ecuadorian amphibian species are threatened with extinction, with 5–25 presumably already extinct (updated from the Global Amphibian Assessment, IUCN et al. 2004); 84% of the threatened species live in the Andean Region. These numbers could be underestimated because at least 131 additional species are classified Data Deficient. It is expected that the number of threatened species will increase with further data.

The potential to have great losses or gains in the conservation of amphibians depends on immediate actions. Under these circumstances, Pontificia Universidad Católica del Ecuador (PUCE) is implementing the Project "Balsa de los Sapos" (Life raft for frogs), which includes a Strategic Plan for the Conservation of the Ecuadorian Amphibians in Risk of Extinction. Goals and activities within this initiative follow major guidelines of the global strategy for conservation of amphibians and the ACAP (Gascon et al. 2007). Thus, the Strategic Plan includes five major programs that are complementary to each other: 1) monitoring and research of native amphibians; 2) improvement of local capacities; 3) *ex-situ* management of all endangered species; 4) environmental education and public awareness; and 5) bioinformatics and diffusion of information. Diverse research laboratories of PUCE and its Museum of Zoology (QCAZ) have converged towards this ambitious project and several activities are already underway. Among them, PUCE raised awareness about the importance of amphibians and their crisis through a public amphibian exhibit "Sapari," carried out in Quito in 2005, which was attended by 105,000 visitors in 90 days. As well, PUCE is building capacity and initiating breeding programs for amphibians. A pilot center (with seed support of the Saint Louis Zoo) has been established for research, management, and *ex-situ* conservation of some species, such as the surviving species of *Atelopus* (i.e., *A. exiguus, A. elegans, A. palmatus, A. spumarius, A.* sp.) and around a dozen species of Andean and Chocoan frogs under high risk of extinction. More information on this Ecuadorian "Life-Raft" initiative is available at AmphibiaWebEcuador (2006).

Manchester University, England

ANDREW GRAY
e-mail: andrew.r.gray@manchester.ac.uk

At Manchester University's Museum, an important and specialized collection of Costa Rican anuran species is being professionally and demographically managed. Although unusual to find such a conservation-focused live collection in a museum, the level of animal care and commitment Manchester is providing also fulfils many aspects of the institution's key objectives. Increasingly, museums are considered to have to play an important role in delivering some of societies global responsibilities. Endeavoring to put something real back to the planet through their amphibian conservation work allows Manchester Museum to do this in a very powerful and meaningful way. The development of global academic links and the support and involvement with 'in country' initiatives further validates this. Large captive colonies for several selected species form a backbone to the successful captive breeding programs that have been initiated here and, apart from having the benefit of a multi-disciplined team of scientists on the doorstep, the collection forms part of a valuable educational and research resource. Many students conduct non-invasive behavioral and biological projects in the department and are also directly involved in supporting conservation efforts through fieldwork.

Amphibian Research Centre in Australia

GERRY MARANTELLI

Established by Gerry Marantelli in 1994, the ARC is a self-funded private facility dedicated to conservation management, research, and education for Australian anurans. The ARC currently holds one of the largest amphibian collections in the world and is involved with ten Nationally Endangered frog species (e.g., Hunter et al. 1999). Management activities also include pioneering work with hygiene and automation: in December 2004 the ARC hosted a workshop "Captivity, Reintroduction and Disease Control Technologies for Amphibians," from which the presentations are available online at the ARC website (www.frogs.org.au). Research efforts include work on assisted reproductive technologies, cryopreservation, and control of introduced Cane Toads. ARC was involved in the discovery of the amphibian chytrid (Berger et al. 1998) and subsequent research on its biology (Berger et al. 2004, 2005; Marantelli et al. 2004), diagnostics (Berger et al. 2002), and treatment (Woodhams et al. 2003). The ARC also focuses on public education through activities like school group tours, classroom tadpole projects, and housing the community outreach Victorian Frog Group. The ARC website is one of the largest and most popular frog websites in the world with over 1.5 million total hits and 80–100,000 visitors each month. ARC work is funded through corporate and private sponsorships, government threatened species recovery programs, and through sales of captive-bred pet frogs of common native species, live food items and other hobbyist supplies, and popular frog-related items.

CONCLUDING COMMENTS

The ACAP is an outline of proposed programs and priorities to work towards a synthetic research and conservation program to amphibian conservation. It is not a detailed plan of action, complete with tasks and funding assigned to specific labs and persons. Sadly, such critical details and organization must await remarkably ambitious and successful fundraising programs (e.g., 2008 Year of the Frog, and proposed activities by the Amphibian Specialist Group). In the meantime, however, inaction is not acceptable and we put forward that the questions and initiatives outlined in ACAP and in various Amphibian Ark documents offer a broad menu of academic honors thesis, dissertation, and post-graduate topics, as well as questions and challenges tractable to zoos, aquariums, botanical gardens, and natural history museums of all sizes.

LITERATURE CITED

AMPHIBIAWEBECUADOR. 2006. Información sobre los anfibios de Ecuador. Museo de Zoología, Centro de Biodiversidad y Ambiente, Pontificia Universidad Católica del Ecuador. Quito. http://www.puce.edu.ec/zoologia/vertebrados/amphibiawebec

BERGER, L., A. HYATT, V. OLSEN, D. BOYLE, S. HENGSTBERGER, G. MARANTELLI, K. HUMPHREYS, AND J. E. LONGCORE. 2002. Production of polyclonal antibodies to *Batrachochytrium dendrobatidis* and their use in an immunoperoxidase test for chytridiomycosis in amphibians. Dis. Aquat. Org. 48:213–220.

———, G. MARANTELLI, L. F. SKERRATT, AND R. SPEARE. 2005. Virulence of the amphibian chytrid fungus *Batrachochytrium dendrobatidis* varies with the strain. Dis. Aquat. Org. 68:47–50.

———, R. SPEARE, P. DASZAK., E. D. GREEN, A. A. CUNNINGHAM, C. L. GOGGIN, R. SLOCOMBE, M. A. RAGAN, A. D. HYATT, K. R. MCDONALD, H. B. HINES, K. R. LIPS, G. MARANTELLI, AND H. PARKES. 1998. Chytridiomycosis causes amphibian mortality associated with population declines in the rain forests of Australia and Central America. Proc. Nat. Acad. Sci. 95:9031–9036.

———, ———, H. HINES, G. MARANTELLI, A. D. HYATT, K. R. MCDONALD, L. F. SKERRATT, V. OLSEN, J. M. CLARKE, G. GILLESPIE, M. MAHONY, N. SHEPPARD, C. WILLIAMS, AND M. TYLER. 2004. Effect of season and temperature on mortality in amphibians due to chytridiomycosis. Austral. Vet. J. 82:434–439.

BLAUSTEIN, A. R., J. M. ROMANSIC, J. M. KIESECKER, AND A. C. HATCH. 2003. Ultraviolet radiation, toxic chemicals and amphibian population declines. Divers. Dist. 9:123–140.

BOONE, M. D., AND C. M. BRIDGES. 2003. A hierarchical approach in studying the effects of an insecticide on amphibians. *In* G. Linder, D. W. Sparling, and S. K. Krest (eds.), Multiple Stressors and Declining Amphibian Populations: Evaluating Cause and Effect, pp. 265–270. SETAC Press, Pensacola, Florida.

BOONE, M. D., AND S. M. JAMES. 2005. Use of aquatic and terrestrial mesocosms in ecotoxicology. Appl. Herpetol. 2:231–257.

———, R. D. SEMLITSCH, J. F. FAIRCHILD, AND B. B. ROTHERMEL. 2004. Effects of an insecticide on amphibians in large-scale experimental ponds. Ecol. Appl. 14:685–691.

———, ———, E. E. LITTLE, AND M. C. DOYLE. 2007. Multiple stressors in amphibian communities: Effects of chemical contamination, bullfrogs, and fish. Ecol. Appl. 17:291–301.

BOSCH, J., L. M. CARRASCAL, L. DURÁN, S. WALKER, AND M. C. FISHER. 2006. Climate change and outbreaks of amphibian chytridiomycosis in a montane area of central Spain; is there a link? Proc. Royal Soc. B-Biol. Sci. online.

Carey, C., J. E. Bruzgul, L. J. Livo, M. L. Walling, K. A. Kuehl, B. F. Dixon, A. P. Pessier, R. A. Alford, and K. B. Rogers. 2006. Experimental exposures of boreal toads (*Bufo boreas*) to a pathogenic chytrid fungus (*Batrachochytrium dendrobatidis*). Ecohealth 3:5–21.

Christin, M. S., A. D. Gendron, P. Brousseau, L. Menard, D. J. Marcogliese, D. Cyr, S. Ruby, and M. Fournier. 2003. Effects of agricultural pesticides on the immune system of *Rana pipiens* and on its resistance to parasitic infection. Environ. Toxicol. Chem. 22:1127–1133.

CLIRSEN. 2006. Deforestación. Centro de Levantamientos Integrados de Recursos Naturales por Sensores Remotos. Quito. http://www.clirsen.com

Coloma, L. A., S. Lötters, W. E. Duellman, and A. Miranda-Leiva. 2007. A taxonomic revision of *Atelopus pachydermus*, and description of two new (extinct?) species of *Atelopus* from Ecuador (Anura: Bufonidae). Zootaxa 1557:1–32.

Cullum, L., and J. T. Justus. 1973. Housing for aquatic animals. Lab Anim. Sci. 23:126–129.

Daszak, P., A. A. Cunningham, and A. D. Hyatt. 2003. Infectious disease and amphibian population declines. Divers. Dist. 9:141–150.

Davidson, C., H. B. Shaffer, and M. R. Jennings. 2002. Spatial tests of the pesticide drift, habitat destruction, UV-B, and climate-change hypotheses for California amphibian declines. Cons. Biol. 16:1588–1601.

Di Rosa, I., F. Simoncelli, A. Fagotti, and R. Pascolini. 2007. The proximate cause of frog declines? Arising from: J. A. Pounds et al. Nature 439, 161–167 (2006). Nature 447:E4–E5.

Gascon, C., J. P. Collins, R. D. Moore, D. R. Church, J. E. McKay, and J. R. Mendelson III (eds.). 2007. Amphibian Conservation Action Plan. IUCN/SSC Amphibian Specialist Group. Gland, Switzerland and Cambridge, UK. 64 pp.

Gilbertson, M., G. D. Haffner, K. G. Drouillard, A. Albert, and B. Dixon. 2003. Immunosuppression in the northern leopard frog (*Rana pipiens*) induced by pesticide exposure. Environ. Toxicol. Chem. 22:101–110.

Harris, R. N., T. Y. James, A. Lauer, M. A. Simon, and A. Patel. 2006. Amphibian Pathogen *Batrachochytrium dendrobatidis* is inhibited by the cutaneous bacteria of amphibian species. Ecohealth 3:53–57.

Harvell, C. D., C. E. Mitchell, J. W. Ward, S. Altizer, A. P. Dobson, R. S. Ostefeld, and M. D. Samuel. 2002. Climate warming and disease risks for terrestrial and marine biota. Science 296:2158–2162.

Hunter, D., W. Osborne, G. Marantelli, and K. Green. 1999. Implementation of a population augmentation project for remnant populations of the southern corroboree frog (*Pseudophryne corroboree*). *In* A. Campbell (ed.), Declines and Disappearances of Australian Frogs, pp. 158–167. Environment Australia, Canberra.

IUCN. 2002. Technical Guidelines on the Management of *Ex-situ* populations for Conservation. 14th Meeting of the Programme Committee of Council, Gland Switzerland, 10 December 2002.

IUCN, Conservation International & NatureServe. 2004. Global Amphibian Assessment. Available from http://www.globalamphibians.org (accessed 13 December 2007).

Johnson, P. T. J., and J. M. Chase. 2004. Parasites in the food web: Linking amphibian malformations and aquatic eutrophication. Ecol. Lett. 7:521–526.

Kluge, A. G. 1996. The early collections and curators, Division of Reptiles and Amphibians, Museum of Zoology, University of Michigan. Herpetol. Rev. 27:176–178.

Kriger, K. M. and J. M. Hero. 2007. Large-scale seasonal variation in the prevalence and severity of chytridiomycosis. J. Zool. 271: 352–359

Lips, K .R., F. Brem, R. Brenes, J. D. Reeve, R. A. Alford, J. Voyles, C. Carey, and J. P. Collins. 2006. Infectious disease and global biodiversity loss: pathogens and enigmatic amphibian extinctions. Proc. Nat. Acad. Sci. 103:3165–3170.

Marantelli, G., L. Berger, R. Speare, and L. Keegan. 2004. Changes in distribution of *Batrachochytrium dendrobatidis* and keratin during tadpole development leading to high mortality after metamorphosis. Pac. Cons. Biol. 10:173–179.

Mendelson III, J.R., and 49 other authors. 2006. Confronting amphibian declines and extinctions. Science 313:48.

Merino-Viteri, A., L. A. Coloma, and A. Almendáriz. 2005. Los *Telmatobius* (Leptodactylidae) de los Andes del Ecuador y su declive poblacional. *In* E. O. Lavilla and I. De la Riva (eds.), Estudios Sobre las Ranas Andinas de los Géneros *Telmatobius* y *Batrachophrynus* (Anura: Leptodactylidae), pp. 9–37. Asociación Herpetológica Española, Monog. Herpetol. 7, Valencia, Spain.

Mills, N. E., and R. D. Semlitsch. 2004. Competition and predation mediate indirect effects of an insecticide on southern leopard frogs. Ecol. Appl. 14:1041–1054.

Myers, C. W. 2000. A history of herpetology at the American Museum of Natural History. Bull. Am. Mus. Nat. Hist. 252:1–232.

Nace, G. W. 1968. The amphibian facility of the University of Michigan. Bioscience 18:767–775.

———, D. D. Culley, M. B. Emmons, E. L. Gibbs, V. H. Hutchison, R. G. McKinnell. 1974. Amphibians: Guidelines for the Breeding, Care, and Management of Laboratory Animals. A Report of the Subcommittee on Amphibian Standards, Committee of Standards, Institute of Laboratory Animal Resources, National Research Council. Washington, D.C., National Academy of Sciences.

Parmesan, C. 2006. Ecological and evolutionary responses to recent climate change. Ann. Rev. Ecol. Syst. 37:637–669.

Pavajeau, L., K.C. Zippel, R. Gibson, and K. Johnson. Amphibian Ark and the 2008 Year of the Frog campaign. in press, Int. Zoo Ybk., vol. 38.

Pounds, J. A., J. A. Consuegra, M. P. L. Fogden, P. N. Foster, K. L. Masters, R. Puschendorf, S. R. Ron, G. A. Sánchez-Azofeifa, and C. J. Still. 2006. Widespread amphibian extinctions from epidemic disease driven by global warming. Nature 439:161–167

Rachowicz, L. J., and V. T. Vredenburg. 2004. Transmission of *Batrachochytrium dendrobatidis* within and between amphibian life stages. Dis. Aquat. Org. 61:75–83.

Relyea, R. A., and N. M. Mills. 2001. Predator-induced stress makes the pesticide carbaryl more deadly to gray treefrog tadpoles (*Hyla versicolor*). Proc. Nat. Acad. Sci. USA 98:2491–2496.

Rollins-Smith, L. A, J. K. Doersam, J. E. Longcore, S. K. Taylor, J. C. Shamblin, C. Carey, and M. A. Zasloff. 2002. Antimicrobial peptide defenses against pathogens associated with global amphibian declines. Dev. Comp. Immunol. 26:63–72.

Ron, S. 2005. Predicting the distribution of the amphibian pathogen *Batrachochytrium dendrobatidis* in the New World. Biotropica 37:209–221.

———, W. E. Duellman, L. A. Coloma, and M. R. Bustamante. 2003. Population decline of the jambato toad *Atelopus ignescens* (Anura: Bufonidae) in the Andes of Ecuador. J. Herpetol. 37:116–126.

———, and A. Merino. 2000. Amphibian declines in Ecuador: overview and first report of chytridiomycosis from South America/Declinación de anfibios en el Ecuador: información general y primer reporte de chytridiomycosis para Sudamérica. Froglog 42:2–3.

Seimon, T. A., A. Seimon, P. Daszak, S. R. P. Halloy, L. M. Schloegel, C. A. Aguilar, P. Sowell, A. D. Hyatt, B. Konecky, and J. E. Simmons. 2007. Upward range extension of Andean anurans and chytridiomycosis to extreme elevations in response to tropical deglaciation. Global Change Biol. 13:288–299.

Semlitsch, R. D., D. E. Scott, J. H. K. Pechmann, and J. W. Gibbons. 1996. Structure and dynamics of an amphibian community: evidence from a 16-year study of a natural pond. *In* M. L. Cody and J. A. Smallwood (eds.), Long-term Studies of Vertebrate Communities, pp. 217–248. Academic Press, San Diego, California.

Stuart, S., J. S. Chanson, N. A. Cox, B. E. Young, A. S. L. Rodrigues, D. L. Fishman, and R. W. Waller. 2004. Status and trends of amphibian declines and extinctions worldwide. Science 306:1783–1786.

Thomas, C. D., A. Cameron, R. E. Green, M. Bakkenes, L. J. Beaumont, Y. C. Collingham, B. F. N. Erasmus, M. F. de Siqueira, A. Grainger, L. Hannah, L. Hughes, B. Huntley, A. S. van Jaarsveld, G. F. Midgley, L. Miles, M. A. Ortega-Huerta, A. T. Peterson, O. L. Phillips, and S. E. Williams. 2004. Extinction risk from climate change. Nature, 427:145–148.

Trueb, L., and W. E. Duellman. 1996. The University of Kansas: A long-time center of herpetological research and training. Herpetol. Rev. 27:56–57.

Wilcox, B. A. 2006. Amphibian decline: more support for biocomplexity as a research paradigm. Ecohealth 3:1–2.

Woodhams D. C., R. A. Alford, and G. Marantelli. 2003. Emerging disease of amphibians cured by elevated body temperature. Dis. Aquat. Org. 55:65–67.

———, K. Ardipradja, R. A. Alford, G. Marantelli, L. K. Reinert, and L. A. Rollins-Smith. 2007. Resistance to chytridiomycosis varies among amphibian species and is correlated with skin peptide defenses. Anim. Cons. 10:409–417.

ARTICLES

Herpetological Review, 2008, 39(1), 29–33.

Biphasic Oviposition by Northwestern Salamanders (*Ambystoma gracile*) in the Cascade Mountains of Southern Washington, USA

JAMES G. MacCRACKEN
Longview Timberlands, LLC, 10 International Way, Box 667
Longview, Washington 98632, USA
e-mail: jgmaccracken@longviewtimber.com

Little has been published about temporal patterns of oviposition by Northwestern Salamanders (*Ambystoma gracile*), either within or among years. Timing of oviposition can be critical to reproductive success and thus subject to strong selection pressures (*sensu* Resetarits 1996). In unpredictable, variable environments more than one tactic may be successful and evolutionarily stable. In this paper, I report the results of a four-year study that documented biphasic oviposition patterns of a population of Northwestern Salamanders, with a preliminary assessment of potential explanatory factors. Five hypotheses follow that may explain this pattern based on the effects of weather on breeding activity, or variation in breeding among different population cohorts. These hypotheses are neither exhaustive nor mutually exclusive. Testable predictions are provided for the first three, but I did not collect data relevant to the latter hypotheses.

H_1: interrupted rainfall.—Breeding in many pond-breeding amphibians appears linked to rainfall and temperature (Anderson 1967; Chadwick et al. 2006; Greenberg and Tanner 2004; Palis 1997; Semlitsch 1985; Sexton et al. 1990; Todd and Winne 2006). A period of either little or no rainfall (which is also often cooler) bracketed by wet and warm weather could interrupt breeding immigrations of Northwestern Salamanders resulting in two distinct oviposition bouts by metamorphic adults within a season. A prediction of this hypothesis is that the appearance of new egg masses is strongly positively correlated (r 0.7) with rainfall. In addition, cloudless dry periods also produce low night temperatures and the number of new egg masses should also be positively correlated with minimum air temperatures.

H_2: periodic drought.—Early breeders may have increased reproductive success during drought years because declining water levels over the summer favor earlier hatching, thus avoiding desiccation of embryos. In addition, larvae of early breeders go through metamorphosis earlier and avoid death due to complete pond drying. Furthermore, larvae of early breeders have more time to feed and develop prior to their first winter. During wetter years, late oviposition may serve to insure that embryos are not subject to late spring freezes, avoid periods of high water flow that could displace egg masses during spring run-off, and reduce exposure of embryos to predators (MacCracken 2007) through accelerated development associated with higher water temperatures later in the season (Brown 1976). Thus, periodic drought at a frequency less than the average individual life span would maintain cohorts of early and late breeders resulting in two peaks in oviposition within a breeding season.

For the advantages of early breeding to be realized: 1) oviposition should begin earlier as drought results in a shallow snowpack, early snowmelt, and relatively warm temperatures; 2) the active oviposition period should be shorter; and 3) the majority of egg masses should be deposited earlier compared to non-drought years because late breeders experience unfavorable weather during immigration.

H_3: paedomorph/metamorph.—Life history variation may result in two distinct oviposition bouts in some ambystomatid salamanders. Earlier breeding of 6–8 weeks by paedomorph versus metamorph Mole Salamanders (*A. talpoideum*) has been documented (Krenz and Sever 1995; Scott 1993; Whiteman and Semlitsch 2005). In addition, paedomorphs breed, and presumably, deposit eggs at greater water depths (Whiteman and Semlitsch 2005). Many Northwestern Salamander populations also contain both morphologies (Carl and Cowan 1945; Eagleson 1976; Efford and Mathias 1969; Snyder 1956; Sprules 1974), as did the population I studied. Contrary to the pattern reported for *A. talpoideum*, Richter and Kerr (2001) found no difference in oviposition chronology for paedomorphic and metamorphic Northwestern Salamanders, but did find that egg masses were deposited at greater depths by paedomorphs. A prediction of this hypothesis is that the peak in the number of new egg masses observed between early and late periods should be several weeks apart. In addition, the number of new egg masses should be nearly equal between early and late oviposition periods, because the proportion of paedomorphs in Northwestern Salamander populations increases with elevation (Carl and Cowan 1945; Eagelson 1976; Snyder 1956) and given the moderate elevation of my study area, I would expect the size of the two cohorts to be close to unity.

H_4: alternate year breeding.—Some ambystomatids have alternate (or more) year breeding (Church et al. 2007; Harris and Ludwig 2004; Kinkead and Otis 2007; Nussbaum et al. 1983), suggesting that resources may not be sufficient to support annual breeding by all individuals in a population. Alternate year breeders may reproduce later in the season because limited resources result in a longer period for ova to complete the yolking stage. For this scenario to produce two oviposition bouts, populations would have to contain both bi-annual and annual breeders.

H_5: double clutching.—Double-clutching also could produce two distinct periods of oviposition within a breeding season. Northwestern Salamanders are presumed to only lay one egg mass/season (Thoms et al. 1997), and this assertion likely rests with the physiological improbability of double-clutching. However, the lack of documentation prevents dismissing this hypothesis.

Methods.—This study was conducted in a 25.5-ha wetland in the Cascade Range of southwestern Washington, USA. The wetland had a southeast aspect, 3% slope, an elevation range of 915–940 m, and had over 40 ponds of various sizes (see MacCracken 2007) and in various stages of vegetation succession. MacCracken (2005, 2007) provided a more detailed description of the study area. Snow normally begins accumulating in late-November/early-December and persists until late March/early June. Rainfall is generally greatest in April–June, followed by a summer drought beginning in mid-July and persisting until September–October.

Thirty-three ponds consistently used for breeding by Northwestern Salamanders were surveyed annually from 1999–2002 (MacCracken 2007). I surveyed each pond every 5–7 days starting with the onset of Northwestern Salamander oviposition activity (early April–early May) until all egg masses had completely hatched, typically in mid-August. I searched each pond for egg masses by traversing the perimeter, and wading sections 1.3 m deep that were not visible from the shore. Water clarity allowed good visibility to a depth of about 2 m. When an egg mass was first discovered, I marked each with a numbered pin flag inserted into the substrate next to the egg mass, and measured the depth of the top of the egg mass below the water surface with a meter stick (MacCracken 2007).

Data on air temperature and precipitation were collected from 2–4 sites at each visit, except in 2000. Weather stations were established in the north and south portions of the wetland in 1999 with two more stations added in 2001 at intermediate locations. Each station had two rain gauges about 50 m apart and a maximum-minimum thermometer placed in a shaded location between the rain gauges. Instruments were set at ground level and read and reset at each visit. In addition, water temperature of each pond was measured with a liquid thermometer about 12 cm below the surface at each visit. These data allow for an assessment of hypotheses one, two, and three as outlined below.

For H_1, Spearman rank-order correlations were estimated comparing weekly rainfall and minimum air temperature with the number of new egg masses for 1999, 2001, and 2002.

For H_2, mean total rainfall and snow water equivalent for 1998–2006 water years (October–September) was calculated from data for the South Fork Bull Run Snotel station (15 km SE of my study area, 902 m elev., http://www.or.nrcs.usda.gov/snow/data/historic.html). The mean of these metrics were compared to the individual water year data for 1999–2002 to identify drought years during my study. The date of the onset of oviposition and the duration of the active oviposition period was compared between drought and non-drought years. The mean number of new egg masses laid between the early and late oviposition periods was compared with a Mann-Whitney *U*-test as the data were slightly skewed in some years (Fig. 1). Early and late periods were delineated based on the date of the fewest number of new egg masses observed following the initial peak.

For H_3, the mean number of new egg masses was compared between early and late periods with a Mann-Whitney *U*-test. Furthermore, paedomorphs of Northwestern Salamanders oviposit at greater water depths (Richter and Kerr 2001), thus early deposited egg masses should be at greater depths than late deposited egg masses. Mean egg mass depth was compared between early and late periods with a Mann-Whitney *U*-test as these data were also slightly skewed toward smaller values.

Results.—I observed a total of 762 egg masses during this study, with the greatest number in 2001. The temporal distribution of oviposition had two peaks each year (Fig. 1).

To examine H_1, rainfall data from the study area were available for each year except 2000. In 1999, 2001, and 2002, 1–5 weeks of either little or no rainfall, bracketed by wetter weather, occurred during the active oviposition period (Fig. 1). The strongest positive correlations between number of new egg masses and weekly rainfall occurred in 2001 ($r_s = 0.75$, $P = 0.05$) and 1999 ($r_s = 0.49$, $P = 0.13$; Fig. 1). However, the relationship was negative and weak in 2002 ($r_s = -0.19$, $P = 0.37$). There was a lag of about a week between the occurrence of new egg masses and rain in 2002. When that lag effect was removed, the correlation was moderate and positive ($r_s = 0.65$, $P = 0.09$). In addition, the strongest correlation between new egg masses and minimum air temperatures was for 2001 ($r_s = -0.67$, $P = 0.06$), but was in the opposite direction of that predicted. These comparisons provide mixed support for the interrupted rainfall hypothesis.

Relative to H_2, the winter of 2000–2001 experienced a strong El Niño event. Precipitation for the 2001 water year was 65% of normal; the snowpack was shallow, ephemeral, and had melted completely by mid-March on the study area (MacCracken, pers. obs.). The average January–April 2001 snowpack at the South Bull Run station had less water content (1.5[SE = 0.8] cm) than the average for 1998–2005 (5.8[1.0] cm).

Both average water and maximum air temperatures on the study area were 3° and 7°C warmer, respectively, in May 2001 than other years, despite May and June rainfall in 2001 being 145% and 35% greater than the same months in 1999 and 2002, respectively (Table 1). Oviposition by Northwestern Salamanders began 1–3 weeks earlier in 2001 compared to 1999, 2000, and 2002 (Fig. 1). Duration of the active oviposition period in 2001 was six weeks compared to 7–9 weeks for the other years. In addition, more egg masses were deposited during the early than late period in 2001, a pattern that was reversed in the other years (Fig. 1). These comparisons agree with the predictions of the periodic drought hypothesis.

For H_3, an average of 3.2(0.1) weeks separated the peaks between the early and late oviposition periods (Fig. 1). However, on average 22% fewer new egg masses were deposited during the early period (76[32]) when compared to the late period (93[18], $U = 8.0$, $P = 1.0$) over the four years of study. In addition, the average depth of egg masses below the water surface was 25(1) and 23(1) cm ($U = 63849$, $P = 0.01$) for early and late periods, respectively. This result was not a function of pond drying as water levels were stable during this time period (J. G. MacCracken, unpubl. data). These comparisons are consistent with the predictions of the paedomorph/metamorph hypothesis (H_3).

Discussion.—None of the hypotheses examined can be dismissed based on my analyses and are best viewed in the context of Chamberlin's (1965) method of multiple working hypotheses in that several mechanisms may act in concert (Elliott and Brook

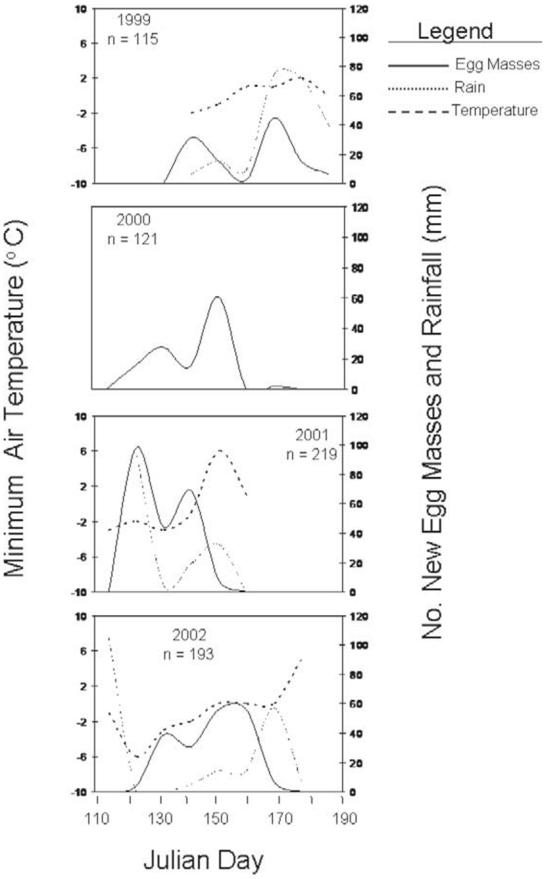


FIG. 1. Counts of new *Ambystoma gracile* egg masses and average rainfall and minimum daily air temperature by 10-day increments for 1999–2002 in the Cascade Mountains of southwestern Washington, USA. Rainfall and air temperature data were not collected in 2000.

2007) to produce the pattern I observed. Furthermore, additional relevant hypotheses also may be developed in the future.

Given the caveat above, my analyses and those in the published literature lend the greatest support to the two weather-related hypotheses as explanations for biphasic oviposition by Northwestern Salamanders. At the seasonal level, numerous studies of a variety of amphibians have found two or more waves of immigrations to breeding ponds that corresponded to precipitation, air temperatures, or both (Anderson 1967; Beneski et al. 1986; Brodman 1995; Chadwick et al. 2006; Cook et al. 2006; Greenberg and Tanner 2004, 2005; Harris 1980; Healy 1975; Hurlbert 1969; Loredo and Van Vuren 1996; Palis 1997; Semlitsch 1981, 1985; Sexton et al. 1990; Timm et al. 2007; Todd and Winne 2006; Vasconcelos and Calhoun 2004). The weight of evidence from these studies lends strong support to the interrupted rainfall hypothesis, despite the relatively inconclusive results from this study, suggesting that: 1) Northwestern Salamanders might use other clues to initiate immigration, (e.g., declining barometric pressure); 2) that my sampling regime was temporally too coarse to capture the details of the relationship; 3) the occurrence of rain may be more important than the actual amount; or 4) paedomorphs make up a relatively large portion of the population.

My data lend some support to the periodic drought hypothesis as a mechanism sustaining two oviposition bouts. The response

TABLE 1. Mean (standard error) maximum and minimum air temperature (°C), pond water temperature (°C), and precipitation (mm) for May–September (5–9) from 1999–2002 in the Cascade Mountains in southwestern Washington, USA. nd = no data.

	1999					2000					2001					2002				
	5	6	7	8	9	5	6	7	8	9	5	6	7	8	9	5	6	7	8	9
Air$_{maximum}$	21(3)	15(1)	25(1)	27(1)	22(1)	nd	nd	28(2)	nd	23(2)	24(1)	20(1)	24(1)	23(1)	23(2)	15(1)	24(1)	26(1)	25(1)	27(1)
Air$_{minimum}$	-1(1)	1(1)	4(1)	5(1)	0(1)	nd	nd	0(1)	nd	3(2)	-1(1)	2(1)	5(1)	5(1)	5(2)	-2(1)	1(1)	3(1)	2(1)	1(1)
Water	9(5)	13(1)	15(1)	15(1)	11(1)	11(1)	14(1)	14(1)	17(2)	nd	13(1)	13(1)	15(1)	15(2)	nd	11(1)	14(1)	16(1)	13(1)	nd
Precipitation	6(0)	14(6)	15(7)	5(1)	9(2)	nd	nd	0	nd	1(1)	49(16)	20(5)	6(2)	21(9)	0(0)	38(33)	23(7)	22(12)	1(1)	11(6)

by Northwestern Salamanders to drought conditions during the winter of 2000–2001 may provide insight into the effects of climate change on this species. If increased frequency of drought is part of climate change (see Lueng et al. 2004 and Payne et al. 2004) then a pattern similar to that observed in 2001 also may occur more frequently. Precipitation amount and timing are likely the most important variables (Russell et al. 2005; Todd and Winne 2006). Warm winter/early-spring temperatures with adequate rainfall will favor early breeding. Chadwick et al. (2006) speculated that earlier breeding by *Triturus* spp. could increase reproductive success by: 1) increasing the length of time larvae have to feed before the next winter; 2) reducing the age at first breeding; 3) reducing competition with other larval amphibians; and 4) creating greater temporal separation between growing larvae and size-limited predators. However, these advantages will be realized only if ponds maintain adequate water levels, which is less likely during drought years.

The paedomorph/metamorph hypothesis also was weakly supported by the data. However, the prediction of equal numbers of egg masses being laid in early and late periods was based on my assumption that the paedomorph and metamorph cohorts were equal in size, and the prediction could also be consistent with the interrupted rainfall and periodic drought hypotheses. In addition, although the 2-cm difference in egg mass average depth between early and late periods was statistically significant, its biological significance is questionable. Furthermore, my findings both conflict and agree with Richter and Kerr (2001) who reported no temporal separation in oviposition, but a 5-cm difference in egg mass depth between the two morphologies in an experimentally manipulated Northwestern Salamander population in Washington. The temporal discrepancy could be due to differences in study areas, the experimental nature of Richter and Kerr (2001), or other unknown factors. Again, the biological significance of a 2–5-cm difference in egg mass depth is unknown. Surveys designed to quantify breeding activity of the two morphologies are needed to provide better information about the veracity of the paedomorph/metamorph hypothesis.

In summary, biphasic oviposition occurred in each of four years, with the temporal properties apparently influenced by seasonal and annual rainfall patterns and to a lesser degree by average minimum temperatures. A droughty winter followed by a wet spring resulted in an earlier and shorter oviposition period with more egg masses in the early peak; which was the opposite of wetter years. Although my data are consistent with predictions of the hypothesis that the biphasic pattern was a result of differential breeding by paedomorphs and metamorphs, the assumptions behind those predictions need validation.

Acknowledgments.—A. DeYoung, E. Smith, and S. Lampman assisted in data collection. Comments by M. P. Hayes, A. J. Kroll, K. O. Richter, and anonymous reviewers improved this paper. This research was conducted under annual renewal of Washington Department of Fish and Wildlife Scientific Collection Permits.

Literature Cited

Anderson, J. D. 1967. A comparison of the life histories of coastal and montane populations of *Ambystoma macrodactylum* in California. Am. Midl. Nat. 77:232–355.

Beneski, Jr., J.T ., E. J. Zalisko, and J. H. Larsen, Jr. 1986. Demography and migratory patterns of the eastern long-toed salamander, *Ambystoma macrodactylum columbianum*. Copeia 1986:398–409.

Brodman, R. 1995. Annual variation in breeding success of two syntopic species of *Ambystoma* salamanders. J. Herpetol. 29:111–113.

Brown, H. A. 1976. The time-temperature relation of embryonic development in the Northwestern Salamander, *Ambystoma gracile*. Can. J. Zool. 54:552–558.

Carl, G. C., and I. M. Cowan. 1945. Notes on the salamanders of British Columbia. Copeia 1945:43–44.

Chadwick, E. A., F. M. Slater, and S. J. Ormerod. 2006. Inter- and intraspecific differences in climatically mediated phonological change in coexisting *Tritrtus* species. Global Change Biol. 12:1069–1078.

Chamberlin, T. C. 1965. The method of multiple working hypotheses. Sci. 148:754–759. (reprint of 1890 paper, Sci. 15:93)

Church, D. R., L. L. Bailey, H. M. Wilbur, W. M. Kendall, and J. E. Hines. 2007. Iteroparity in the variable environment of the salamander, *Ambystoma tigrinum*. Ecology 88:891–903.

Cook, D. G., P. C. Trenham, and P. T. Northern. 2006. Demography and breeding phenology of the California tiger salamander (*Ambystoma californiense*) in an urban landscape. Northwestern Nat. 87:215–224.

Eagleson, G. W. 1976. A comparison of the life histories and growth patterns of populations of the salamander *Ambystoma gracile* (Baird) from permanent low-altitude and montane lakes. Can. J. Zool. 54:2098–2111.

Efford, I. E., and J. A. Mathias. 1969. A comparison of two salamander populations in Marion Lake, British Columbia. Copeia 1969:723–736.

Elliott, L. P., and B. W. Brook. 2007. Revisiting Chamberlin: multiple working hypotheses for the 21st century. BioSci. 57:608–614.

Greenberg, C. H., and G.W . Tanner. 2004. Breeding pond selection and movement patterns by eastern spadefoot toads (*Scaphiopus holbrookii*) in relation to weather and edaphic conditions. J. Herpetol. 38:569–577.

———, and ———. 2005. Spatial and temporal ecology of oak toads (*Bufo quercicus*) on a Florida landscape. Herpetologica 61:422–434.

Harris, R. N. 1980. The consequences of within-year timing of breeding in *Abystoma maculatum*. Copeia 1980:719–722.

———, and P. M. Ludwig. 2004. Resource level and reproductive frequency in female four-toed salamanders, *Hemidactylium scutatum*. Ecology 85:1585–590.

Healy, W. R. 1975. Breeding and postlarval migrations of the red-spotted newt, *Notophthalmus viridescens*, in Massachusetts. Ecology 56:673–680.

Hurlbert, S. H. 1969. The breeding migrations and interhabitat wandering of the Vermillion-Spotted Newt *Notophthalmus viridescens* (Rafinesque). Ecol. Monogr. 39:465–488.

Kinkead, K. E., and D. L. Otis. 2007. Estimating superpopulation size and annual probability of breeding for pond-breeding salamanders. Herpetologica 63:151–162.

Krenz, J. D., and D. M. Sever. 1995. Mating and oviposition in paedomorphic *Ambystoma talpoideum* precedes the arrival of terrestrial males. Herpetologica 51:387–393.

Loredo, I., and D. Van Vuren. 1996. Reproductive ecology of a population of the California tiger salamander. Copeia 1996:895–091.

Lueng, L. R., Y, Quian, X. Bian, W. M. Washington, J. Han, and J. O. Roads. 2004. Mid-Century Ensemble Regional Climate Change Scenarios for the Western United States. Climate Change 62:75–113.

MacCracken, J. G. 2005. Effects of uneven-aged timber harvest on forest floor vertebrates in the Cascade Mountains of southern Washington. For. Ecol. Manage. 208:123–135.

———. 2007. Northwestern salamander (*Ambystoma gracile*) oviposition sites and hatching success in the Cascade Mountains of southern Washington. Herpetol. Conserv. Biol. 2:127–134.

Nussbaum, R. A., E. D. Brodie, Jr., and R. M. Storm. 1983. Amphibians and Reptiles of the Pacific Northwest. University of Idaho Press, Moscow.

PALIS, J. G. 1997. Breeding migration of *Ambystoma cingulatum* in Florida. J. Herpetol. 31:71–78.

PAYNE, J. T., A. W. WOOD, A. F. HAMLET, R. N. PALMER, AND D. P. LETTENMAIER. 2004. Mitigating the effects of climate change on the water resources of the Columbia River Basin. Climate Change 62:233–256.

RESETARITS, JR., W. J. 1996. Oviposition site choice and life history evolution. Am. Zool. 36:205–215.

RICHTER, K. O., AND D. W. KERR. 2001. Oviposition biology of a sympatric population of transformed and gilled *Ambystoma gracile* (northwestern salamander). Northwest. Nat. 62:79.

RUSSELL, A. P., A. M. BAUER, AND M. K. JOHNSON. 2005. Migration in amphibians and reptiles: an overview of patterns and orientation mechanisms in relation to life history strategies. *In* A.S.M.T. Elewa (ed.), Migration of Organisms: Climate, Geography and Ecology, pp. 151–203. Springer Verlag, Berlin.

SCOTT, D. E. 1993. Timing of reproduction of paedomorphic and metamorphic *Ambystoma talpoideum*. Am. Midl. Nat. 129:397–402.

SEMLITSCH, R. D. 1981. Terrestrial activity and summer home range of the mole salamander (*Ambystoma talpoideum*). Can. J. Zool. 59:315–322.

———. 1985. Analysis of climactic factors influencing migrations of the salamander *Ambystoma talpoideum*. Copeia 1985:447–489.

SEXTON, O. J., C. PHILLIPS, AND J. E. BRAMBLE. 1990. The effects of temperature and precipitation on the breeding migration of the spotted salamander (*Ambystoma maculatum*). Copeia 1990:781–787.

SPRULES, W. G. 1974. Environmental factors and the incidence of neotony in *Ambystoma gracile* (Baird) (Amphibia: Caudata). Can. J. Zool. 52:1545–1552.

SNYDER, R. C. 1956. Comparative features of the life histories of *Ambystoma gracile* (Baird) from populations at low and high altitudes. Copeia 1956:41–50.

THOMS, C., C. C. CORKRAN, AND D. H. OLSON. 1997. Basic amphibian survey for inventory and monitoring in lentic habitats. Northwest Fauna 4:35–46.

TIMM, B. C., K. MCGARIGAL, AND B. W. COMPTON. 2007. Timing of large movement events of pond-breeding amphibians in western Massachusetts, USA. Biol. Conserv. 136:442–454.

TODD, B. D., AND C. T. WINNE. 2006. Ontogenetic and interspecific variation in timing of movement and responses to climatic factors during migrations by pond–breeding amphibians. Can. J. Zool. 84:715–722.

VASCONCELOS, D., AND A. J. K. CALHOUN. 2004. Movement pattern of adult and juvenile *Rana sylvatica* (LeConte) and *Ambystoma maculatum* (Shaw) in three restored seasonal pools in Maine. J. Herpetol. 38:551–561.

WHITEMAN, H. H., AND R. D. SEMLITSCH. 2005. Asymmetric reproductive isolation among polymorphic salamanders. Biol. J. Linn. Soc. 86:265–281.

Herpetological Review, 2008, 39(1), 33–35.

Eastern Kingsnake (*Lampropeltis g. getula*) Population Decline in Northern Florida and Southern Georgia

SETH P. STAPLETON[1,2,*]
KIM J. SASH[1,2]
D. BRUCE MEANS[3]
WILLIAM E. PALMER[1]
and
JOHN P. CARROLL[2]
[1] *Tall Timbers Research Station, 13093 Henry Beadel Drive*
Tallahassee Florida, 32312, USA

[2] *University of Georgia, Warnell School of Forestry and Natural Resources,*
Athens, Georgia, 30605, USA

[3] *Coastal Plains Institute and Land Conservancy,*
1313 Milton Street, Tallahassee, Florida, 32303, USA

**Corresponding author: seth.stapleton@gmail.com*

The Common Kingsnake (*Lampropeltis getula*), distributed across much of the United States (Ernst and Ernst 2003) and inhabiting numerous habitats in the Southeast (Ernst and Ernst 2003; Gibbons and Semlitsch 1991; Means 2006), was historically a common member of Florida's snake communities (Krysko and Smith 2005). However, kingsnake populations have declined statewide, and many appear to have been locally extirpated (Krysko and Smith 2005). This decline has generated concern among conservationists, and speculation as to potential causes ranges from habitat loss, road mortality, and pollution, to collection for the pet trade (Ernst and Ernst 2003; Krysko 2002; Krysko and Smith 2005). Further ecological research including population monitoring is needed to enhance conservation efforts (Krysko 2002; Krysko and Smith 2005). Here, we examine long-term population trends of the eastern kingsnake (*L. g. getula*) in northern Florida and southern Georgia by comparing data from two studies conducted more than two decades apart.

Methods.—Tall Timbers Research Station (TTRS), Pebble Hill (PH), and the Wade Tract (WT), all located in the Red Hills of northern Georgia and southern Florida, served as the study sites. The Red Hills region lies between Tallahassee, Florida, and Thomasville, Georgia, and is bounded by the Ochlockonee River to the west and the Aucilla River to the east. TTRS is the southernmost site and is located ca. 30 km (20 miles) N of Tallahassee, Florida. The landscape, characterized by a rolling topography and rich clay soils, is dominated by open pine forests interspersed with hardwood bottomland drains and annually harrowed fields. The Red Hills region is intensively managed for Northern Bobwhite (*Colinus virginianus*) and other Longleaf Pine (*Pinus palustris*) associated species (Stoddard 1931). Management focuses on the maintenance of open upland habitats via prescribed burning and mechanical methods to inhibit hardwood encroachment and maintain herbaceous groundcover.

The initial study took place at TTRS from May 1976 to July 1983. Snakes were opportunistically collected during the daytime as we obtained radiotelemetry fixes for the Eastern Diamondback Rattlesnake (Means 1985), searching for Cottonmouths (Cook

1983), or conducting other field activities. Individuals were marked via scute clipping (Brown and Parker 1976) and basic biological data were obtained (Cook 1983; Means 2005).

During 2002–2005, an intensive study using a systematic trapping protocol on TTRS, PH, and WT reexamined the Red Hill's snake community. Snakes were captured with drift fences used in conjunction with box traps (D. Richardson, pers. comm.; Burgdorf et al. 2005), and single-ended funnel traps (Fitch 1987) were set at the end of each of the four fence arms. Fences were placed randomly across the landscape in bottomland hardwood drains, a forested "buffer" zone bordering hardwood drains, upland pine forests, and fields. Twenty-four fences were set at TTRS and PH for at least 11 days per month from March to October, 2002. In 2003, trapping effort was increased and standardized, with 42 fences set for 18 trap days per month from March to October at TTRS, PH, and WT. Thirty-six fences were set at TTRS and PH during 2004–2005 using a trapping protocol identical to 2003. Additional snakes were collected via opportunistic encounters by individuals conducting other research activities. Snakes were initially marked either via a passive integrated transponder (PIT) tag or via a unique scute clip.

Results and Discussion.—Methodological differences between the two studies prohibit rigorous statistical comparison of the data sets. However, the large change in detection of the Eastern Kingsnake is striking. During the initial 7-year study, 119 nonvenomous snakes representing 10 species were recorded. Twenty-four Eastern Kingsnakes were documented, constituting nearly 20% of captures during the period. The intensive drift fence trapping protocol of the second study period spanned 19,380 trapping array days and, with supplemental opportunistic captures, yielded 2310 individuals representing 16 nonvenomous species. However, only one Eastern Kingsnake was documented during this period. This individual was captured at PH and represents < 0.05% of total nonvenomous individuals.

Despite a more intensive and systematic sampling effort, the 2002–2005 study documented a tremendous reduction in absolute and relative detection of the Eastern Kingsnake. We acknowledge that biases exist with all survey methods of snakes (e.g., Dodd and Franz 1995; Enge 2001; Gibbons and Semlitsch 1987; Greenberg et al. 1994; Kjoss and Litvaitis 2001; Prior et al. 2001). Different sampling protocols thus may have contributed to the observed decline in detection. However, the intensity of the 2002–2005 study, the ability of the sampling protocol to capture other large, semi-fossorial species such as the Corn Snake (*Elaphe guttata*) and Florida Pine Snake (*Pituophis melanoleucus mugitus*) (Stapleton 2005; Sash, unpubl. data), and the presence of many field personnel to potentially encounter Eastern Kingsnakes strongly suggest a real and remarkable population decline.

Research conducted at the Joseph W. Jones Ecological Research Center at Ichauway (JWJERC), located in Newton, Georgia, ca. 105 km (70 miles) north, stands in contrast to the Red Hills' Eastern Kingsnake decline. Like our study sites, JWJERC primarily consists of open canopied pine forests managed extensively with prescribed fire. Sampling at JWJERC employed a similar drift fence design using box traps which, despite a less intensive trapping regime, resulted in the capture of 60 Eastern Kingsnakes from 2003 to 2006 (L. Smith, pers. comm.). These data thus illustrate the ability of our drift fence design to capture Eastern Kingsnakes and, interestingly, point to the localized nature of the kingsnake decline. This localization is further demonstrated by surveillance cameras identifying Northern Bobwhite nest predators in the region. During an 8-year period beginning in 1999, nest monitoring cameras did not document Eastern Kingsnakes on TTRS or PH. Conversely, Eastern Kingsnakes were the most commonly documented snake species in the Albany, Georgia region (Staller et al. 2005) and predated 40% of all video-monitored nests during 2005–2006 (J. Carroll, W. Palmer, and D. Sisson, unpubl. data).

Although the Eastern Kingsnake was not documented on TTRS with either the drift fence trapping regime or nest-monitoring cameras, non-detection of a species does not indicate its true absence from the site (Kery 2002; Mackenzie 2005; Mackenzie et al. 2002). Therefore, we do not suggest that the Eastern Kingsnake has been completely extirpated from TTRS. In fact, a DOR specimen was noted in 2004 on a secondary road < 1.5 km from TTRS. However, our data strongly suggest a severe decrease in the Red Hills' Eastern Kingsnake population.

Causes for the kingsnake decline in the Red Hills are currently unknown. In the southeastern U.S., *L. getula* is found in a variety of habitats, often associated with low-lying wetland areas and overlapping much of the historical range of longleaf pine forests (Ernst and Ernst 2003; Gibbons and Semlitsch 1991; Krysko 2002; Means 2006; Means and Krysko 2001; Palmer and Braswell 1995). Our study sites appear to provide a landscape suitable for Eastern Kingsnakes; fingerlike drains and permanent and ephemeral wetlands are scattered across the open pine forest landscape and a large lake forms the southern boundary of TTRS. Additionally, the current landscape at our sites is generally similar to habitat in the 1970s. A thinning to reduce hardwood encroachment in the upland pine forests did occur at TTRS and PH during the late 1990s, but similar hardwood thinnings occurred at the Albany sites which documented Eastern Kingsnakes with surveillance cameras (W. Palmer, pers. obs.). Moreover, any land management activities that restore or maintain the open-canopy, pineland habitat with a vigorous herbaceous groundcover—the native habitat of the Eastern Kingsnake—should be beneficial to kingsnakes.

Our data provide further documentation of the statewide decline documented by Krysko and Smith (2005). The location of some of our study sites in southern Georgia additionally suggests that kingsnake declines may extend beyond Florida and into other portions of the southeastern United States. Current population statuses and the uncertainties surrounding the causes of kingsnake declines suggest a directive for future research (Krysko 2002; Krysko and Smith 2005). Studies should assess the status of the Southeast's populations and identify the underlying causes from which conservation solutions may be developed (Krysko 2002; Krysko and Smith 2005).

Acknowledgments.—We thank the staff of Tall Timbers Research Station, Inc. for their assistance with this project. Lora Smith and the Joseph W. Jones Ecological Research Center graciously shared Eastern Kingsnake capture records. Three anonymous reviewers provided useful comments to improve the manuscript. Funding was provided in part by grants from the United States Department of Agriculture – Wildlife Services and the National Geographic Society's Committee for Research and Exploration, the Warnell School of Forestry and Natural Resources, and Tall Timbers Research Station, Inc. Research during 2002–2005 was conducted under University of Georgia Institutional Animal Care and Use Committee

(IACUC) permits A2001-10100-c1 and c2.

Literature Cited

Brown, W. S., and W. S. Parker. 1976. A ventral scale clipping system for permanently marking snakes. J. Herpetol. 10:247–249.

Burgdorf, S. J., D. C. Rudolph, R. N. Conner, D. Saenz, and R. R. Schaefer. 2005. A successful trap design for capturing large terrestrial snakes. Herpetol. Rev. 36:421–424.

Cook, D. G. 1983. Activity patterns of the cottonmouth water moccasin, *Agkistrodon piscivorus* Lacepede, on a northwest Florida headwater stream. Unpubl. M.S. thesis. Univ. of Florida, Gainesville, Florida. 89 pp.

Dodd, C. K., Jr., and R. Franz. 1995. Seasonal abundance and habitat use of selected snakes trapped in xeric and mesic communities of north-central Florida. Bull. Florida Mus. Nat. Hist. 38, Part I(2):43–67.

Enge, K. M. 2001. The pitfalls of pitfall traps. J. Herpetol. 35:467–478.

Ernst, C. H., and E. M. Ernst. 2003. Snakes of the United States and Canada. Smithsonian Institution Press, Washington, D.C. 668 pp.

Fitch, H. S. 1987. Collecting and life-history techniques. *In* R. A. Seigel, J. T. Collins, and S. S. Novak (eds.), Snakes: Ecology and Evolutionary Biology, pp. 143–164. McGraw Hill, Inc., New York.

Gibbons, J. W., and R. D. Semlitsch. 1987. Activity patterns. *In* R. A. Seigel, J. T. Collins, and S. S. Novak (eds.), Snakes: Ecology and Evolutionary Biology, pp. 396–421. McGraw Hill, Inc., New York.

———, and ——— 1991. Guide to the Reptiles and Amphibians of the Savannah River Site. University of Georgia Press, Athens, Georgia. 131 pp.

Greenberg, C. H., D. G. Neary, and L. D. Harris. 1994. A comparison of herpetofaunal sampling effectiveness of pitfall, single-ended, and double-ended funnel traps used with drift fences. J. Herpetol. 28:319–324.

Kery, M. 2002. Inferring the absence of a species—a case study of snakes. J. Wildl. Manage. 66:330–338.

Kjoss, V. A., and J. A. Litvaitis. 2001. Comparison of 2 methods to sample snake communities in early successional habitats. Wildl. Soc. Bull. 29:153–157.

Krysko, K. L. 2002. Seasonal activity of the Florida kingsnake *Lampropeltis getula floridana* (Serpentes: Colubridae) in southern Florida. Amer. Midl. Nat. 148:102–114.

———, and D. J. Smith. 2005. The decline and extirpation of kingsnakes *Lampropeltis getula*, in Florida. *In* W. E. Meshaka, Jr. and K. J. Babbitt (eds.), Status and Conservation of Florida Amphibians and Reptiles, pp. 132–141. Krieger Publishing Co., Malabar, Florida.

MacKenzie, D. I. 2005. Was it there? Dealing with imperfect detection for species presence / absence data. Aust. N.Z. J. Stat. 47:65–74.

MacKenzie, D. I., J. D. Nichols, G. B. Lachman, S. Droege, J. A. Royle, and C. A. Langtimm. 2002. Estimating site occupancy rates when detection probabilities are less than one. Ecology 83:2248–2255.

Means, D. B. 1985. Radio-tracking the eastern diamondback rattlesnake. Nat. Geog. Soc. Res. Rep. 18:529–536.

———. 2005. The value of dead tree bases and stumpholes as habitat for wildlife. *In* W. E. Meshaka, Jr. and K. J. Babbitt (eds.), Status and Conservation of Florida Amphibians and Reptiles, pp. 74–78. Krieger Publishing Co., Malabar, Florida.

———. 2006. Vertebrate faunal diversity in longleaf pine savannahs. *In* S. Jose, E. Jokela, and D. Miller (eds.), The Longleaf Pine Ecosystem: Ecology, Restoration, and Silviculture, pp. 157–214. Springer Verlag, New York.

———, and K. L. Krysko. 2001. Biogeography and pattern variation of kingsnakes, *Lampropeltis getula*, in the Apalachicola region of Florida. Contemp. Herpetol. 2001(5):1–33.

Palmer, W. M., and A. L. Braswell. 1995. Reptiles of North Carolina. University of North Carolina Press, Chapel Hill, North Carolina. 412 pp.

Prior, K. A., G. Blouin-Demers, and P. J. Weatherhead. 2001. Sampling biases in demographic analyses of black rat snakes (*Elaphe obsoleta*). Herpetologica 57:460–469.

Staller, E. L., W. E. Palmer, J. P. Carroll, R. P. Thornton, and D. C. Sisson. 2005. Identifying predators at northern bobwhite nests. J. Wildl. Manage. 69:124–132.

Stapleton, S. P. 2005. Snake ecology in the Red Hills of Georgia and Florida. Unpubl. M.S. Thesis. University of Georgia, Athens, Georgia. 159 pp.

Stoddard, H. L. 1931. The Bobwhite Quail: Its Habits, Preservation, and Increase. Charles Scribner's Sons, New York. 559 pp.

Herpetological Review, 2008, 39(1), 35–40.

Midsummer Ground Surface Activity Patterns of Western Toads (*Bufo boreas*) in Southeast Idaho

STEPHEN R. SULLIVAN*
Department of Biology and Chemistry, Carroll College
Helena, Montana 59625 USA

PAUL E. BARTELT**
and
CHARLES R. PETERSON
Herpetology Laboratory, Department of Biological Sciences
Idaho Museum of Natural History, Idaho State University
Pocatello, Idaho 83209-8007 USA

**Corresponding author and current address:*
Department of Plastic Surgery, Harvard Medical School
Children's Hospital Boston, 300 Longwood Avenue, Hunnewell 1
Boston, Massachusetts 02115, USA
e-mail: stephen.sullivan3@childrens.harvard.edu

***Current address: Department of Biology, Waldorf College*
Forest City, Iowa 50436 USA

While some behavioral aspects of Western Toads (*Bufo boreas*) have been documented (Campbell 1970; Carey 1978; Hailman 1984; Lilliwhite et al. 1973; Mullally 1958; Muths 2003; Muths and Corn 1997; Pimentel 1955), a great deal remains unknown. Toads are ectotherms that rely on environmental conditions to maintain body temperature and conserve water. Activity patterns appear to be critical in determining body temperature (Stevenson 1985), but the specific daily behaviors used by Western Toads for thermoregulation are not fully known. The greatest gap in our understanding of amphibians derives from how little is known about daily activity (Shoemaker 1992).

Conservation measures must incorporate aspects of amphibian ecology and behavior, such as habitat relationships and factors that affect their daily movements and other activities. The response of amphibian populations to landscape changes may range from negative (e.g., habitat fragmentation; Funk et al. 2004) to positive (e.g., fire; Hossack and Corn 2007). Thus, more detailed information on their daily behaviors may help us better anticipate the effects of landscape changes on amphibians.

We conducted a focal animal study with radiotelemetry to investigate and quantitatively characterize activity patterns, behavior, and microsite selection by Western Toads in southeastern Idaho. Whereas habitat selection has been studied in this area (Bartelt et al. 2004), we advance our understanding of this species' behavior by focusing on how time of day/night, environmental conditions, and habitat relate to activity. Our results may be particularly relevant to the species' conservation in this region because the Western Toad has experienced population declines within the Greater Yellowstone Ecosystem (Koch and Peterson 1995).

Methods.—Our study was conducted within the Greater Yellowstone Ecosystem at Stamp Meadows in the Targhee National Forest (UTM 471117 E, 4928908 N; elevation approx. 2000 m) in southeastern Idaho, USA. Stamp Meadows was a seasonally flooded meadow that contained a 13 ha pond, bordered by willows (*Salix*) and aspen (*Populus*), and surrounded by coniferous forest habitats (*Pinus*, *Picea*, and *Abies*) that had been modified by logging.

We studied the daily activity of 4 male (nuptial pads present) Western Toads (range 60–85 mm snout–urostyle length; 40–70 g). We used a plastic belt to secure a 1.85 g BD-2GT transmitter (Holohil Systems, Ltd., Carp, Ontario, Canada) to each toad, a method that appears not to affect toad behavior (Bartelt and Peterson 2000; Murray and Fuller 2000).

We used focal animal sampling and observed one toad at a time with continuous recording of all instances of behavior and time (Martin and Bateson 1993). We made observations from 17 July to 3 August 1995 and divided each 24 h day into 8 observation periods of 3 h (periods: 1 = 0000–0259 h, 2 = 0300–0559 h, 3 = 0600–0859 h, 4 = 0900–1159 h, 5 = 1200–1459 h, 6 = 1500–1759 h, 7 = 1800–2059 h, and 8 = 2100–2359 h). Periods 1, 2, and 8 included times of darkness and are described as night while other periods are described as day. Periods 4, 5, and 6 are specifically described as midday. We randomly selected 2 periods each day and randomly selected a toad to observe during each of these periods. We observed each toad for each 3 h observation period.

We located toads with a Telonics TR-2 receiver and H-type directional antenna (Telonics, Inc., Mesa, Arizona). The observer sat approximately 3 m away from the toad under a camouflage net to minimize toad disturbance. We made night observations with a red-filtered 3.8 V incandescent light and used diffuse light peripheral to the central beam to reduce light intensity (Buchanan 1993). To test the effects of our light on the toads, we changed position relative to toads on several occasions during nighttime observations and the light did not appear to change direction of travel or orientation of toads.

We measured ambient (the area encompassing the study site as a whole) and microsite (the area within 3 m of the toad) weather conditions. Terrestrial ambient air temperature 1 m above ground and relative humidity were recorded with a CR10 data logger (Campbell Scientific, Inc., Logan, Utah) and PCRC-11 humidity transducer (Phys-Chem Scientific Corp., New York, New York) (<3% error between 12 to 100% RH, <1% error between 25 to 94% RH) placed near the center of the study site. Ambient measurements were taken every second, then averaged for each observation period. Terrestrial microsite air temperature was recorded 1 m above ground in shade with a Schultheis rapid registering mercury thermometer (Miller and Weber, Inc., New York, New York) at the onset, termination, and periodically during each observation period, then averaged for each observation period. Aquatic microsite water temperature, 1 cm below the water surface, also was recorded when toads were located in an aquatic microsite.

We recorded the sequence and time of all activity (e.g., attempt to capture prey using a tongue strike (feeding strike), distance traveled, position change, direction change, vocalization, habitat change, interaction with other toads) during observation periods. A minute was considered active if any detectable activity, other than breathing, was observed. Distance traveled, a subcategory and more conservative measure of activity, was used for data analysis. Distance traveled was measured in body lengths and distance in cm was estimated based on the known snout–urostyle length of each study animal. Microsite habitats (e.g., terrestrial, aquatic, underground) used by toads also were recorded. When toads were underground, ground level active minutes and distance traveled were recorded as zero because toads could not be observed directly and were not exposed to ground level environmental conditions.

Independent variables were time (observation period), average temperature (terrestrial ambient, terrestrial microsite, and aquatic microsite) during the observation period, and average relative humidity during the observation period. Temperature was categorized for analysis (< 5°C, 5–9.9°C, 10–14.9°C, 15–19.9°C, and >19.9°C). Dependent variables were active minutes and distance traveled. We used one–way analysis of variance (ANOVA) to test for potential differences between independent and dependent variables. To examine for potential differences between microsite temperature and ambient temperature, we conducted a regression analysis with indicator variables for observation periods. To examine for a potential association between relative humidity and distance traveled, we conducted a linear regression analysis. Statistical analyses were performed using Stata 8.0 software (StataCorp 2004, College Station, Texas).

Results.—We made a total of 32 observations of the four toads over the 8 observation periods (Table 1). Individual toads were seldom observed for the same number of minutes for each observation period. This was usually due to the toad disappearing from view. Toads were active during all observation periods with peaks in activity during the midday and night (Fig. 1). Activity differed among observation periods ($F_{7, 24} = 2.4$, $P = 0.05$).

During the day, toads were located at terrestrial retreat microsites, where they moved short distances and frequently changed orientation or position while basking in the sunlight. At these microsites, toads were seen making feeding strikes (Table 1) at crawling insects such as ants, spiders, and beetles. Only once did we see a toad strike at a flying ant. Toads were observed to feed during all observation periods except at night during period 1.

At night, the four toads traveled up to 40 m during period 8 from terrestrial microsites to aquatic microsites where other toads were also observed (Fig. 2). We also observed toads returning to terrestrial microsites in the early morning during periods 2 and 3. Relatively shorter distances of movement (0–0.95 m) occurred during midday observation periods 4, 5, and 6. Distance traveled differed among observation periods ($F_{7, 24} = 3$, $P = 0.02$).

In addition to a relationship between distance traveled and observation period, distance traveled was significantly different among microsite temperature categories ($F_{4, 27} = 2.9$, $P = 0.04$). Toads traveled furthest when microsite air temperature was 5–9.9°C or 10–14.9°C, and traveled shorter distances when temperature exceeded 15.0°C (Fig. 3). Observation period and temperature are related and the relationship represents a sinusoidal pattern

TABLE 1. Observation periods and activities of four Western Toads radiotracked in southeastern Idaho, USA, in July and August 1995.

	Observation Period							
	1 (0000–0259)	2 (0300–0559)	3 (0600–0859)	4 (0900–1159)	5 (1200–1459)	6 (1500–1759)	7 (1800–2059)	8 (2100–2359)
Toad 1								
Observation minutes	173	133	156	170	28	143	16	112
Active minutes	41	16	13	47	11	3	4	70
Distance traveled (cm)	1050	340	2	0	0	0	1	3971
Feeding strikes	0	0	2	6	0	2	0	2
Toad 2								
Observation minutes	180	180	180	117	115	153	72	103
Active minutes	0	0	0	17	13	0	2	44
Distance traveled (cm)	0	0	0	82	0	0	0	995
Feeding strikes	0	0	0	0	0	0	0	7
Toad 3								
Observation minutes	166	157	146	123	167	164	157	156
Active minutes	3	72	3	18	9	42	4	52
Distance traveled (cm)	0	3720	0	3	0	7	5	765
Feeding strikes	0	2	1	6	1	0	1	2
Toad 4								
Observation minutes	180	180	187	136	180	166	96	148
Active minutes	0	0	71	23	52	37	12	97
Distance traveled (cm)	0	0	1846	10	65	0	45	4045
Feeding strikes	0	0	3	1	6	2	3	1
Total all toads								
Observation minutes	699	650	669	546	490	626	341	519
Active minutes	44	88	87	105	85	82	22	263
Distance traveled (cm)	1050	4060	1848	95	65	7	51	9776
Feeding strikes	0	2	6	13	7	4	4	12

(Fig. 4). Ideally, a multivariate analysis would be performed to evaluate the relative contributions of observation period and temperature to distance traveled, but the small sample size of this study prevents such analysis. However, distance traveled appears to be related more to observation period than temperature. For example, the average microsite temperatures during observation period 8 were category 2 (5–9.9°C) and 3 (10–14.9°C); and the average distance traveled during this period was 24.4 m. However, when the same average microsite temperature categories 2 and 3 occurred during other observation periods, the average distance traveled was only 7.9 m. This suggests that the longer distances traveled during observation period 8 were related in part to time of day rather than simply microsite temperature. Relative humidity also appears to contribute to distance traveled. For example, ambient relative humidity was significantly greater when toads traveled 9 m (88.3 ± 2.5% vs. 78.3 ± 2.4%; $t = 1.73$; $P = 0.047$).

Microsite temperature appears to be related to microsite habitat selection. Microsite air temperatures tended to be warmer than the surrounding terrestrial ambient air temperatures during periods 3, 4, 5, and 6 (Fig. 4) and were significantly warmer during midday periods 4 and 5 ($F_{1,14} = 7.6$, $P = 0.02$ and $F_{1,16} = 10.6$, $P = 0.005$, respectively). This suggests toads selected midday microsite habitat based at least in part on temperature. At night, ambient and microsite air temperatures were often near freezing. In these cold conditions, toads were found in warmer habitats, such as aquatic microsites. When toads were in aquatic microsites, water temperatures were up to 3°C warmer than terrestrial ambient air temperatures and 13°C warmer than terrestrial microsite air temperatures. In the later dates of the study, two toads traveled greater than 80 m from aquatic microsites and did not return to aquatic microsites at night. These two toads were located underground in a rodent hole and under a tree stump during cold nights. While driving to and from the study site at night on these cold nights, we also observed several toads in crouched positions on exposed dirt roads.

Vocalization was recorded on separate occasions for each toad, including occasions not provoked by contact with another animal. A toad was heard vocalizing while: 1) an ant was crawling on its head; 2) turning body orientation; 3) in an underground hole; and 4) in the process of digging in plant material. On two occasions at night, a toad vocalized when another toad attempted to amplex with it; upon vocalization, the other toad released its grasp and moved toward and clasped another nearby toad. On two other occasions at night, we heard three male toads, positioned at separate and distant (e.g., 50–70 m) points in the pond, repeatedly and sequentially vocalize.

Discussion.—We observed four male Western Toads during midsummer at high elevation in southeastern Idaho. We found them to be active during all observation periods with a peak in activity in the midday and early night. Despite being active in the midday, distance traveled was limited during midday. Distance traveled was furthest in the early night when toads traveled to warmer aquatic microsites. Our results are similar to Mullally (1958) who described the activity patterns of Western Toads, especially in the higher elevations, as primarily nocturnal with some diurnal activity. However, like other amphibians, activity patterns of Western Toads are not adequately described simply as diurnal, nocturnal, or crepuscular (Hailman 1984). Data from our study show behavior patterns and how variation in environmental conditions may affect toad biology.

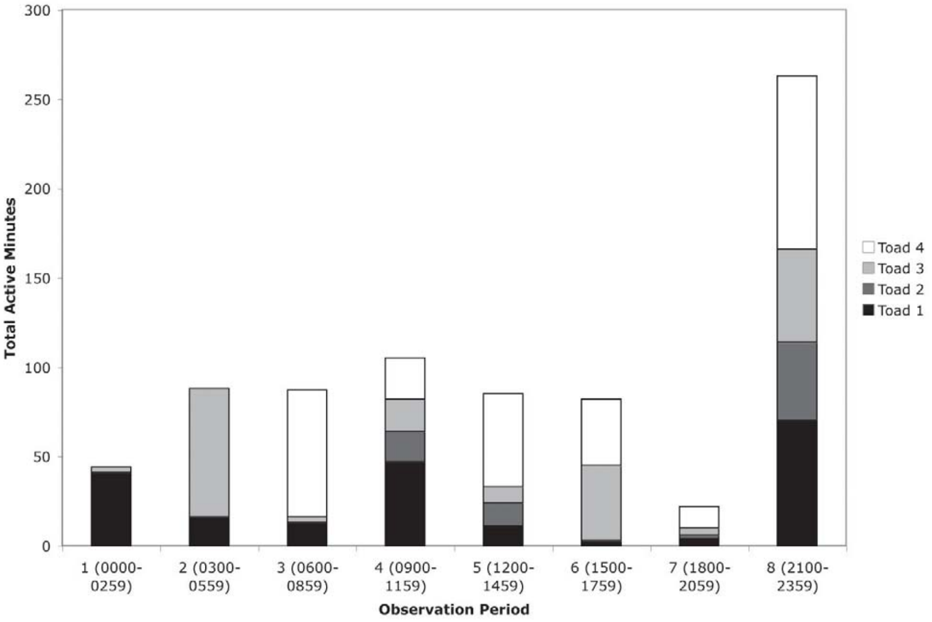


FIG. 1. Western Toad activity patterns were associated with observation period in southeastern Idaho, USA (ANOVA, P = 0.05).

We observed behavior patterns we believe were used to regulate body temperatures and water balance. The regulation of body temperature is primarily behavioral (Spotila et al. 1992) and the choices of times for daily activity appear to be the most critical behavior in determining body temperature (Stevenson 1985). During the day, each toad was located while basking in sunlight at a terrestrial retreat site. These microsites may have been selected, in part, due to their warmer temperatures relative to the surrounding habitat. Our observations that air temperatures of terrestrial retreat microsites were warmer than surrounding ambient air temperatures supports those of Campbell (1970), that toads basked in sunlight as a method to increase body temperature.

Distance traveled was furthest at night, consistent with the observations of other studies. Western Toads emerge and become active (including travel) at night (Mullally 1958; Pimentel 1955), during the hours of our observation period 8, 2100–2359 h (Hailman 1984). The toads may reduce water loss by traveling at these times when temperatures did not exceed 14.9°C and relative humidity generally did not drop below 78%. In addition, when < 80 m from an aquatic microsite, all four toads in our study were found to travel to an aquatic microsite at night. We suggest two reasons for the presence of toads in an aquatic microsite at night. First, during cold nights, the microsite water temperature was warmer than both ambient and terrestrial microsite air temperatures. Water is more thermally stable than many of the terrestrial microhabitats available to toads (Lillywhite 1970). The toads were, therefore, likely able to maintain a warmer body temperature by entering the aquatic microsite because an amphibian in water rarely experiences a body temperature more than 2°C different than the water (Spotila et al. 1992). Second, by entering the aquatic microsite, toads likely were able to regain, by absorption, any water lost during midday sun basking. Many amphibians move between aquatic and terrestrial microsites (Shoemaker 1992). *Bufo punctatus*, for example, has been described to have a pattern of behavior similar to the Western Toads we observed with up to 85 m of travel from its burrow to a small stream at night and returning to the same burrow early in the morning (McClanahan et al. 1994). Mullally (1956) also described Western Toads traveling to aquatic microsites

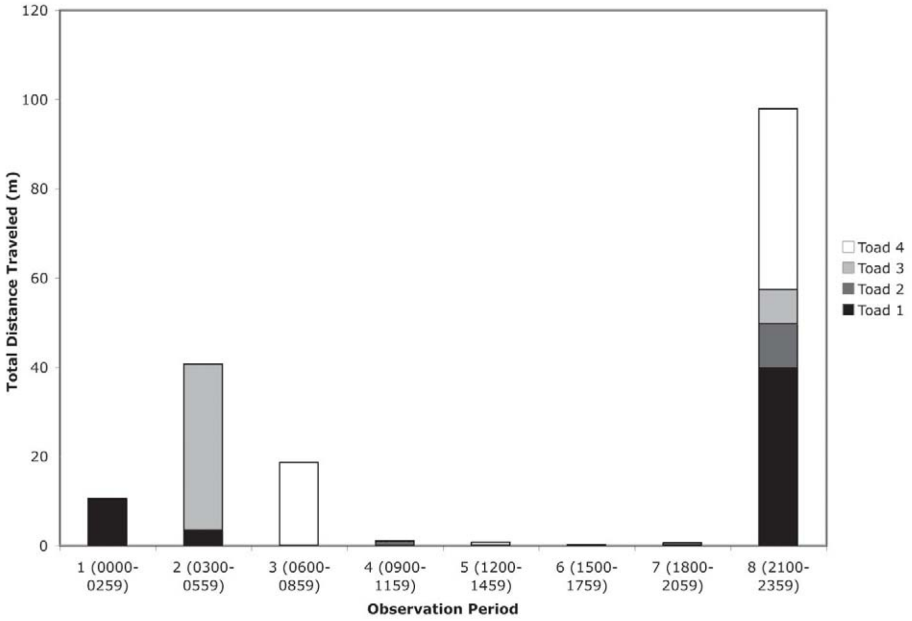


FIG. 2. Distance traveled by Western Toads was associated with observation period in southeastern Idaho, USA (ANOVA, P = 0.02).

in times of dry weather, probably to rehydrate.

Bartelt et al. (2004) observed Western Toads traveling 1.3 km from breeding sites in the late summer and these toads did not return to water. In the later dates of our study, two toads traveled from and were not found to return to aquatic microsites. These toads were located underground in a rodent hole and tree stump where they had no surface level activity. Limits on activity have been shown to be an important factor in water balance (Tracy 1976). Under moderate conditions of temperature and relative humidity, most amphibians cannot survive for more than a day in circulating air, because they quickly dehydrate (McClanahan et al. 1994). Retreating to cover likely reduces both evaporative water loss from midday basking and rehydration needs. Therefore, Western Toads without access to an aquatic microsite likely retreat from exposed sites during times of dryness, as we observed.

In addition, we often found several non–telemetered toads on dry dirt roads at night. Mullally (1958) also found Western Toads on the road and Hailman (1984) discussed toads specifically seeking the road at night. The toads we found on roads were often in a crouched position, which maximized surface contact. Roads are not covered by vegetation and are exposed to radiant energy from the sun, which is stored and lost at the dirt surface. Typical of any system with storage and resistance, there is a time lag in heat flux out of dry soil (Campbell and Norman1998). The toads may crouch on the dry soil road to increase body temperature from this release of heat. Campbell (1970) and Carey (1978) proposed that toads increase temperature by conduction of heat from substrate. For toads on the road, mortality by vehicles may be a significant cause of death. We have previously reported the death by vehicle of a telemetered toad (Bartelt and Peterson 2000).

Mullally (1956) reports that male Western Toads vocalize only when provoked through contact with other animals such as when other males amplex them. In this situation, the clasped male vocalizes, whereupon the clasping male releases its hold. This interaction was observed and the presence of a radio-transmitter did not appear to inhibit or discourage other toads from clasping telemetered toads. We also found that toads vocalize at times when there is no contact by other animals, particularly at night when distant toads repeatedly called in sequence. The fact that vocalization was heard at times other than contact with other animals is interesting and may indicate that vocalization plays a larger role than that of communicating a release during amplexus or provocation.

The observations in our study represent a behavior sample of adult male free-living Western Toads. The activity and habitat use at different times and temperatures likely reflect thermal and water regulatory behavioral strategies. Though limited in sample size, our results demonstrate nocturnal peaks in travel with some level of activity present throughout the 24 h day, including a moderate diurnal component. Toads traveled over a narrow range of tem-

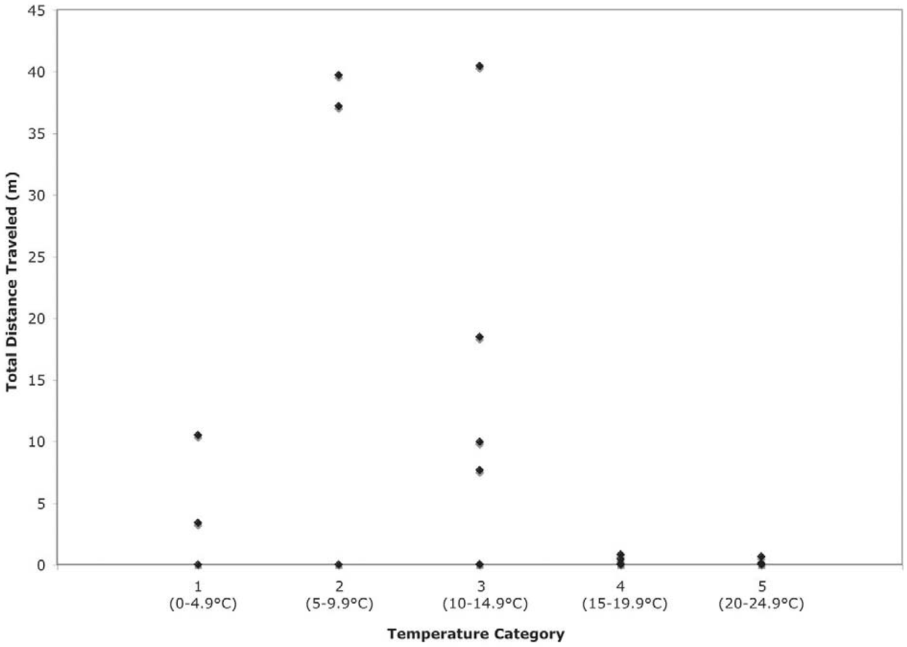


FIG. 3. Distance traveled by Western Toads was associated with microsite temperature category in southeastern Idaho, USA (ANOVA, P = 0.04).

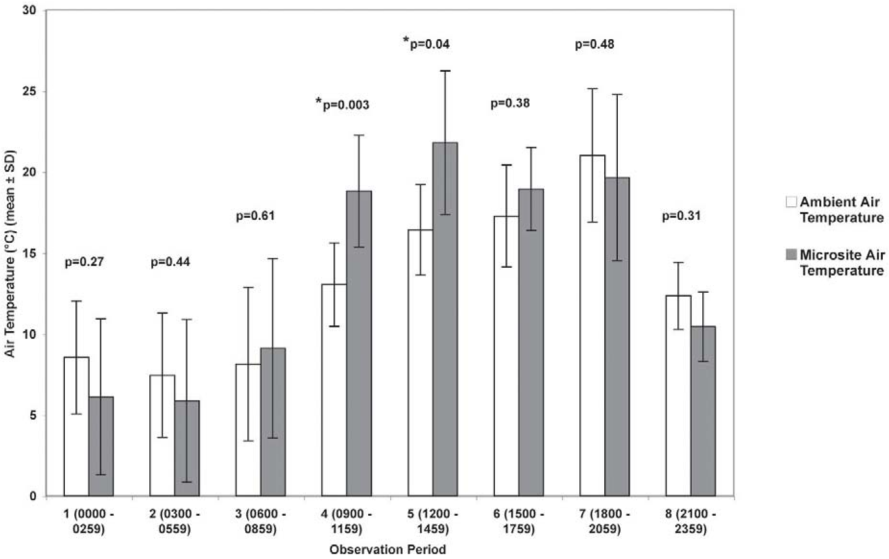


FIG. 4. Average microsite air temperature (°C at 1 m) where a Western Toad was located compared to ambient air temperature (°C at 1 m) during each observation period in southeastern Idaho, USA. Microsite air temperatures of the retreat sites selected by toads during periods 4 and 5 were warmer than ambient air temperatures.

peratures, and traveled longer distances during periods of high humidity. Toads were often found in aquatic microsites and on roads at night and at terrestrial retreat sites during the day.

Acknowledgments.—This research was supported by grants from the USDA Forest Service Intermountain Research Station, the Targhee National Forest, the National Fish and Wildlife Foundation, Theodore Roosevelt Memorial Fund of the American Museum of Natural History, and the Graduate School and Department of Biological Sciences of Idaho State University. The Idaho State University Animal Welfare Committee approved this study (protocols: 12-92-89 and 3-93-106) and state and federal permits were obtained to access the study site and conduct research. We thank John S. Addis, D. Grant Hokit, Kay A. Satre, and Jean Smith for reviewing previous versions of this manuscript.

LITERATURE CITED

BARTELT, P. E., AND C. R. PETERSON. 2000. A description and evaluation of a plastic belt for attaching radio transmitters to western toads (*Bufo boreas*). Northwest. Nat. 81:122–128.

———, ———, AND R. W. KLAVER. 2004. Sexual differences in the post-breeding movements and habitats selected by western toads (*Bufo boreas*) in southeastern Idaho. Herpetologica 60:455–467.

BUCHANAN, B. W. 1993. Effects of enhanced lighting on the behaviour of nocturnal frogs. Anim. Behav. 45:893–899.

CAMPBELL, J. B. 1970. Life history of *Bufo boreas boreas* in the Colorado front range [dissertation]. Univ. of Colorado, Boulder, Colorado. 110 pp.

CAMPBELL, G. S., AND J. M. NORMAN. 1998. An Introduction to Environmental Biophysics, 2nd ed. Spring-Verlag Pub., New York, New York. 304 pp.

CAREY, C. 1978. Factors affecting body temperatures of toads. Oecologia 35:197–219.

FUNK, W. C., A. E. GREENE, P. S. CORN, AND F. W. ALLENDORF. 2005. High dispersal in a frog species suggests that it is vulnerable to habitat fragmentation. Biol. Lett. 1:13–16.

HAILMAN, J. P. 1984. Bimodal nocturnal activity of the western toad (*Bufo boreas*) in relation to ambient illumination. Copeia 1984(2):283–290.

HOSSACK, B. R., AND P. S. CORN. 2007. Responses of pond-breeding amphibians to wildfire: short-term patterns in occupancy and colonization. Ecol. Appl. 17:1403–1410

KOCH, E. D., AND C. R. PETERSON. 1995. The Amphibians and Reptiles of Yellowstone and Grand Teton National Parks. Univ. of Utah Press, Salt Lake City, Utah. 188 pp.

LILLIWHITE, H. B., P. LICHT, AND P. CHELGREN. 1973. The role of behavioral thermoregulation in the growth energetics of the toad, *Bufo boreas*. Ecology 54:375–383.

MARTIN, P., AND P. BATESON. 1993. Measuring Behaviour. Cambridge Univ. Press, New York, New York. 222 pp.

MCCLANAHAN, L. L., R. RUIBAL, AND V. H. SHOEMAKER. 1994. Frogs and toads in deserts. Sci. Am. 270:3:64–70.

MULLALLY, D. P. 1956. The relationship of the Yosemite and western toads. Herpetologica 12:133–135.

———. 1958. Daily period of activity of the western toad. Herpetologica 14:29–31.

MURRAY, D. L., AND M. R. FULLER. 2000. A critical review of the effects of marking on the biology of vertebrates. *In* L. Boitani and T. K. Fuller (eds.), Research Techniques in Animal Ecology: Controversies and Consequences, pp. 15–64. Columbia University Press, New York, New York.

MUTHS, E. 2003. Homerange and movements of boreal toads in undisturbed habitats. Copeia 2003(1):160–165.

———, AND P. S. CORN. 1997. Basking by adult boreal toads (*Bufo boreas boreas*) during the breeding season. J. Herpetol. 31:34–55.

PIMENTEL, R. A. 1955. Habitat distribution and movements of *Bufo boreas boreas*. Herpetologica 11:72.

SHOEMAKER, V. H. 1992. Exchange of water, ions, and gases in terrestrial amphibians. *In* M. E. Feder and W. W. Burggren (eds.), Environmental Physiology of the Amphibians, pp. 125–150. University of Chicago Press, Chicago, Illinois.

SPOTILA, J. R., M. P. O'CONNOR, AND G. S. BAKKEN. 1992. Biophysics of heat and mass transfer. *In* M. E. Feder and W. W. Burggren (eds.), Environmental Physiology of the Amphibians, pp. 55–80. University of Chicago Press, Chicago, Illinois.

STEVENSON, R. D. 1985. The relative importance of behavioral and physiological adjustments controlling body temperature in terrestrial ectotherms. Am. Nat. 126:362–386.

TRACY, C. R. 1976. A model of the dynamic exchanges of water and energy between a terrestrial amphibian and its environment. Ecol. Monogr. 46:293–326.

Herpetological Review, 2008, 39(1), 40–46.

Checklist and Comments on the Terrestrial Amphibian and Reptile Fauna from Utai, Northwestern Papua New Guinea

CHRISTOPHER C. AUSTIN
Museum of Natural Science, 119 Foster Hall, Louisiana State University
Baton Rouge, Louisiana 70803, USA
e-mail: ccaustin@lsu.edu

CHRISTOPHER J. HAYDEN
Museum of Natural Science, 119 Foster Hall, Louisiana State University
Baton Rouge, Louisiana 70803, USA

ILAIAH BIGILALE
Papua New Guinea National Museum and Art Gallery
Natural History Department
P.O. Box 5560 Boroko, NCD, Papua New Guinea

CHRIS DAHL
Conservation International, c/o Wildlife Conservation Society
P.O. Box 277, Goroka
Eastern Highlands Province, Papua New Guinea
and
Department of Biology, University of Papua New Guinea
P.O. Box 320, University Post Office, NCD, Papua New Guinea

and
JIM ANAMINIATO
Papua New Guinea National Museum and Art Gallery
Natural History Department
P.O. Box 5560 Boroko, NCD, Papua New Guinea

New Guinea, the world's largest and highest tropical island, has been identified as one of the world's five High Biodiversity Wilderness Areas (Mittermeier et al. 2003). The island occupies less than 0.6% of global land area but harbors 5–7% of the world's biodiversity (Beehler 1993; Dinerstein and Wikramanayake 1993; Myers et al. 2000). The herpetofauna of New Guinea currently accounts for approximately 5% of the world's reptile and amphibian diversity (Allison et al. 1998). Remarkably, this is an underestimate of true diversity as species accumulation curves for both lizards and frogs continue to increase dramatically without hint of plateau (Fig. 1). The tallest peak on the island exceeds 5000 m and the rugged topography encompasses a wide range of habitat

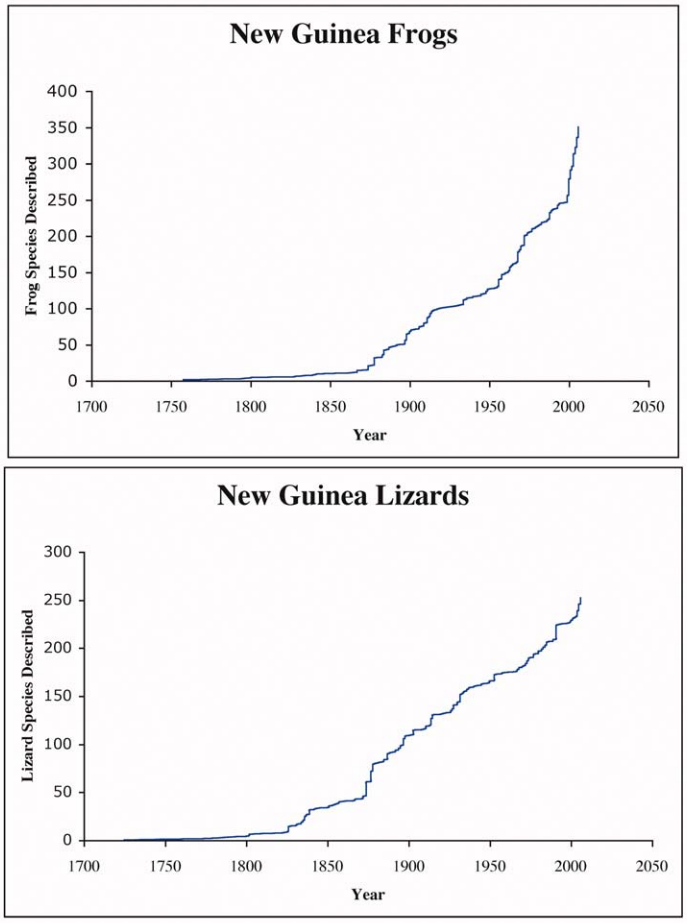


FIG. 1. Species accumulation curves for New Guinea frogs and lizards.

types packed into an area one-tenth the size of the United States. These habitats include mangrove forests, lowland eucalypt savannas, dense lowland rainforests, sago palm swamps, montane moss forests, cloud forests, alpine grasslands, and tropical glaciers on the tallest peaks. The New Guinea herpetofauna can be found in all but the last of these habitats.

Over the last several decades extensive logging has severely impacted the expansive lowland rainforest on the north coast of Papua New Guinea (Beehler 1993). Here we report on a 15-day survey of terrestrial amphibians and reptiles from the area surrounding Utai Village (3°23.765'S, 141°34.974'E, 208 m elev.) in north-central New Guinea in Sandaun Province, Papua New Guinea. Utai Village is situated on the Gedik River, a northern tributary of the Sepik River. The Sepik is the longest river on the island and possibly the largest uncontaminated drainage system in Australasia with a catchment of approximately 80,000 km^2 (Allen and Coates 1990). Utai Village is located at the intersection of upland and forest flood plains. North of Utai the rolling hills give rise to foothills of the Bewani Mountains and to the south is vast expanse of nipa flood plain forest that extends to the Sepik River. The forest includes primary, successional, and riparian lowland broadleaf evergreen forest as well as extensive Sago Palm (*Metroxylon sagu*) swamps. The immediate areas surrounding Utai village have been used for traditional shifting fruit and vegetable gardens and extensive harvesting of Sago Palms for food. The larger surrounding areas are largely unspoiled lowland rainforest in the extensive and broadly contiguous Northern New Guinea lowland ecoregion that contains diverse habitats including lowland and hill forest, grassy swamps, swamp forests, savannas, peat swamp forests, and woodlands (Conn 1995; Gressitt 1982; Henty 1981; Womersley 1978). Although the forests around Utai have

FIG. 2. Map of New Guinea showing Utai in northwestern Papua New Guinea and Kau Wildlife Area in northcentral Papua New Guinea.

Table 1. Checklist of the terrestrial amphibian and reptile fauna of Utai northwestern Papua New Guinea. We refer to a species as 'Common' if it is typically encountered in an approximate 8-person-hour search period. Species listed as 'Uncommon' are encountered infrequently in the area around Utai and typically require more than a single day/night search of 8-person-hours to locate. References refer to general literature associated with each species.

Species		Family	Comments and References
Snakes			
Candoia	*aspera*	Boidae	Uncommon; (Austin, 2000; O'Shea, 1996)
Candoia	*carinata*	Boidae	Uncommon; (Austin, 2000; O'Shea, 1996)
Boiga	*irregularis*	Colubridae	Uncommon; (O'Shea, 1996)
Dendrelaphis	*punctulatus*	Colubridae	Uncommon; (McDowell, 1974; O'Shea, 1996; McDowell, 1975; McDowell, 1979)
Dendrelaphis	sp.	Colubridae	Uncommon; (McDowell, 1974; O'Shea, 1996; McDowell, 1975; McDowell, 1979)
Stegonotus	*modestus*	Colubridae	Uncommon; (O'Shea 1996; McDowell, 1972)
Stegonotus	*parvus*	Colubridae	Uncommon; (O'Shea 1996; McDowell, 1972)
Stegonotus	sp.	Colubridae	Uncommon; (O'Shea 1996; McDowell, 1972)
Tropidonophis	*doriae*	Colubridae	Uncommon; (O'Shea, 1996; Malnate and Underwood, 1988)
Tropidonophis	*mcdowelli*	Colubridae	Uncommon; (O'Shea, 1996; Malnate and Underwood, 1988)
Tropidonophis	*multiscutellatus*	Colubridae	Uncommon; (O'Shea, 1996; Malnate and Underwood, 1988)
Tropidonophis	*picturatus*	Colubridae	Uncommon; (O'Shea, 1996; Malnate and Underwood, 1988)
Aspidomorphus	*muelleri*	Elapidae	Uncommon; (O'Shea, 1996; McDowell, 1967)
Aspidomorphus	sp.	Elapidae	Uncommon; (O'Shea, 1996; McDowell, 1967)
Micropechis	*ikaheka*	Elapidae	Uncommon; (O'Shea 1996)
Morelia	*viridis*	Pythonidae	Uncommon; (low density) (Rawlings and Donnellan, 2003)
Lizards			
Hypsilurus	*modestus*	Agamidae	Common; (Moody, 1980; Manthey and Denzer, 2006)
Hypsilurus	sp.	Agamidae	Uncommon; (Moody, 1980; Manthey and Denzer, 2006)
Cyrtodactylus	*sermowaiensis*	Gekkonidae	Common; (de Rooij, 1915; Brown and Parker, 1973)
Cyrtodactylus	sp.	Gekkonidae	Uncommon; (de Rooij, 1915; Brown and Parker, 1973)
Gehyra	cf. *lampei*	Gekkonidae	Uncommon; (King and Horner, 1989; King, 1984; Chrapliwy et al., 1961; Beckon, 1992)
Gekko	*vittatus*	Gekkonidae	Uncommon; (de Rooij, 1915; Brown and Parker, 1973)
Hemidactylus	*frenatus*	Gekkonidae	Common; (Mortiz et al., 1993)
Lepidodactylus	*lugubris*	Gekkonidae	Uncommon; (Mortiz et al., 1993)
Nactus	cf *multicarinatus*	Gekkonidae	Common; (Donnellan and Moritz 1995; Moritz 1987; Zug and Moon 1995; Kraus, 2005)
Nactus	*vankampeni*	Gekkonidae	Common; (Donnellan and Moritz 1995; Moritz 1987; Zug and Moon 1995; Kraus, 2005)
Cryptoblepharus	sp.	Scincidae	Uncommon; (Mertens, 1928)
Emoia	*caeruleocauda*	Scincidae	Common; (Brown, 1991)
Emoia	*jakati*	Scincidae	Common; (Brown, 1991)
Emoia	*kordoana*	Scincidae	Uncommon; (Brown, 1991)
Emoia	*longicauda*	Scincidae	Uncommon; (Brown, 1991)
Emoia	*obscura*	Scincidae	Common; (Brown, 1991)
Emoia	*pallidiceps*	Scincidae	Common; (Brown, 1991)
Lamprolepis	*smaragdina*	Scincidae	Common; (Greer, 1970)
Lipinia	*albodorsalis*	Scincidae	Uncommon; (Austin, 1998; Zweifel, 1979; Shea and Greer, 2002)
Lipinia	*pulchra*	Scincidae	Uncommon; (Austin, 1998; Zweifel, 1979)
Lobulia	*brongersmai*	Scincidae	Uncommon; (Greer et al., 2005; Zweifel, 1972; Allison and Greer, 1986)
Prasinohaema	*virens*	Scincidae	Uncommon; (Mys, 1988)
Sphenomorphus	*jobiensis*	Scincidae	Common; (Donnellan and Aplin, 1989)
Sphenomorphus	*minutus*	Scincidae	Uncommon; (Greer, 1974)
Sphenomorphus	*pratti*	Scincidae	Uncommon; (Greer, 1974)
Sphenomorphus	*simus*	Scincidae	Common; (Shea and Greer, 1999)
Sphenomorphus	*solomonis*	Scincidae	Common; (de Rooij, 1915)
Tiliqua	*gigas*	Scincidae	Uncommon; (Shea, 1990)
Tribolonotus	*gracilis*	Scincidae	Uncommon; (Zweifel, 1966; Cogger, 1972)
Varanus	*jobiensis*[2]	Varanidae	(Böhme, 2003)

TABLE 1.—Continued.

SPECIES		FAMILY	COMMENTS AND REFERENCES
FROGS			
Litoria	cf. *genimaculata*[1]	Hylidae	(Menzies, 2006)
Litoria	*huntorum*	Hylidae	Uncommon; (Richards et al., 2006)
Litoria	*infrafrenata*	Hylidae	Common; (Zweifel, 1980; Menzies, 2006)
Litoria	*nigropunctata*	Hylidae	Uncommon; (Menzies, 2006)
Litoria	*thesaurensis*	Hylidae	Uncommon; (Menzies, 2006; Kraus and Allison, 2004)
Litoria	sp.	Hylidae	Uncommon; (Menzies, 2006)
Austrochaperina	sp.	Microhylidae	Uncommon; (Menzies, 2006)
Choerophryne	*rostellifer*	Microhylidae	Uncommon; (Menzies, 2006)
Cophixalus	cf. *bewaniensis*	Microhylidae	Uncommon; (Menzies, 2006; Kraus and Allison, 2000)
Cophixalus	*balbus*	Microhylidae	Common; (Menzies, 2006)
Cophixalus	sp.	Microhylidae	Uncommon; (Menzies, 2006)
Copiula	sp.	Microhylidae	Common; (Burton, 1990; Menzies, 2006)
Hylophorbus	sp. 1	Microhylidae	Common; (Günther, 2001; Menzies, 2006)
Hylophorbus	sp. 2	Microhylidae	Uncommon; (Günther, 2001; Menzies, 2006)
Hylophorbus	sp. 3	Microhylidae	Uncommon; (Günther, 2001; Menzies, 2006)
Hylophorbus	sp. 4	Microhylidae	Uncommon; (Günther, 2001; Menzies, 2006)
Mantophryne	*lateralis*	Microhylidae	Uncommon; (Menzies, 2006)
Oreophryne	*biroi*	Microhylidae	Uncommon; (Menzies, 2006; Zweifel et al., 2003)
Oreophryne	*hypsiops*	Microhylidae	Uncommon; (Menzies, 2006; Zweifel et al., 2003)
Oreophryne	sp. 1	Microhylidae	Uncommon; (Menzies, 2006; Zweifel et al., 2003)
Oreophryne	sp. 2	Microhylidae	Uncommon; (Menzies, 2006; Zweifel et al., 2003)
Sphenophryne	*cornuta*	Microhylidae	Common; (Menzies, 2006; Bickford, 2002)
Xenobatrachus	*tumulus*[2]	Microhylidae	Uncommon; (Menzies, 2006; Kraus and Allison, 2002; Blum and Menzies, 1988)
Xenobatrachus	sp.	Microhylidae	Uncommon; (Menzies, 2006; Kraus and Allison, 2002; Blum and Menzies, 1988)
Xenorhina	*oxycephala*[1]	Microhylidae	(Menzies, 2006; Blum and Menzies, 1988)
Lechriodus	*melanopyga*	Myobatrachidae	Uncommon; (Menzies, 2006)
Limnonectes	*grunniens*	Ranidae	Uncommon; (Menzies, 2006)
Platymantis	*cheesmanae*	Ranidae	Uncommon; (Menzies, 2006)
Platymantis	*papuensis*	Ranidae	Common; (Zweifel, 1969; Menzies, 1982a; Menzies, 1982b; Menzies, 2006)
Rana	*arfaki*	Ranidae	Uncommon; (Menzies, 2006)
Rana	*daemeli*	Ranidae	Uncommon; (Menzies, 2006)
Rana	cf *grisea*	Ranidae	Common; (Menzies, 2006)
Rana	*volkerjane*[2]	Ranidae	(Menzies, 2006)

[1] Previously collected by CD in 2005.

[2] Reported by Kraus and Allison 2006.

yet to be commercially logged, logging roads are encroaching rapidly from the north and will reach the village within the next several years.

To date there have been no comprehensive herpetofaunal reports from northwestern Papua New Guinea. Here we compile a list of terrestrial reptile and amphibian species from the vicinity of Utai based on a field survey conducted from 27 June to 12 July 2006 (Table 1) as well as from previous fieldwork (CD) and published reports (Kraus and Allison 2006). Kraus and Allison (2006) reported range extensions for reptiles and amphibians along the northern versant of Papua New Guinea that included three species not collected by our team but reported within 5 km of Utai village. The fauna, exclusive of crocodilians and turtles, includes 79 species comprising 16 snakes, 30 lizards, and 33 frogs representing 40 genera in 11 families (Table 1). This list undoubtedly is an underestimate of the true diversity present in the region. Specific specimen (and associated tissues) and locality information can be accessed via a searchable database of the LSU Museum of Natural Science reptile and amphibian collection (http://www.lsu.edu/museum).

SPECIES RICHNESS, TAXONOMY AND SPECIES-COMPLEX GROUPS

There are a large number of species complexes in the diverse New Guinea herpetofauna, especially among the scincid lizards and microhylid frogs (e.g., Austin 2006). In addition to obvious species complexes, many currently recognized species likely harbor cryptic species (Bickford et al. 2006). Species richness, therefore, is likely much greater than is presently documented from our survey. We are taking a multidisciplinary approach using morphological and molecular data to tease apart many of the species complexes as well as to describe the species obviously new to science. Below we comment on and address some of the taxonomic impediments to understanding the terrestrial amphibian and reptile fauna in the Utai area.

Treesnakes of the genus *Dendrelaphis* are easy to identify to the generic level but extensive geographic variation in color pattern and scalation, inadequate keys, and cryptic diversity make *Dendrelaphis* a problematic group (McDowell 1974; O'Shea 1996). We collected one species that keys out to *Dendrelaphis punctulatus* but has a drastically different color pattern and thus may represent an undescribed species. In addition, we collected several specimens of what is clearly an undescribed species of *Dendrelaphis*. Keelback snakes of the genus *Tropidonophis* have had a taxonomically complicated history, having been assigned to such diverse genera as *Amphiesma*, *Macropophis*, *Natrix*, and *Styporhynchus* (Malnate and Underwood 1988; O'Shea 1996). *Tropidonophis* are easily distinguished from other New Guinean snakes by having prominently keeled dorsal scales, but species identification remains difficult. We collected three specimens of the elapid genus *Aspidomorphus*. One adult and juvenile were easily assigned to *A. muelleri* and a third specimen was not readily assignable to any of the three currently recognized species of *Aspidomorphus* and likely represents a species new to science.

The angle head lizards of the agamid genus *Hypsilurus* have been recently revised (Manthey and Denzer, 2006). Manthey and Denzer (2006) recognize 14 species of *Hypsilurus*, while acknowledging that this diversity is undoubtedly an underestimate. There are two species of *Hypsilurus* in the Utai region. *Hypsilurus modestus* is a small-bodied (maximum SVL = 107 mm) *Hypsilurus* with a broad distribution throughout the Papuan region (New Guinea and nearby associated islands) and is common in the Utai area. A second *Hypsilurus* species does not key to any of the currently described species and likely represents a species new to science.

The gecko fauna of New Guinea consists of seven genera: *Cyrtodactylus*, *Gehyra*, *Gekko*, *Hemidactylus*, *Hemiphyllodactylus*, *Lepidodactylus*, and *Nactus*. Six of these seven gecko genera were found in the Utai region with the seventh, *Hemiphyllodactylus*, representing an uncommon, hard to find and/or patchily distributed gecko poorly represented in collections; one of us (CCA) has collected just three individuals of *Hemiphyllodactylus* in 18 years of fieldwork on the island. Although *Hemiphyllodactylus* was not recorded from the Utai area it may occur in the region. *Cyrtodactylus*, *Gehyra*, and *Nactus* represent taxonomically challenging groups in need of revision and study. The geographic distribution and specific-level delineation for virtually all species of New Guinea geckos is not well understood and many collections have misidentified taxa. Currently ten species of *Cyrtodactylus* are described from New Guinea, but several undescribed species exist in collections (Kraus and Allison 2006). *Cyrtodactylus sermowaiensis* was collected regularly at night and was the most abundant species of *Cyrtodactylus* present. Two specimens of a much larger species of *Cyrtodactylus* where collected. These are similar to *C. novaeguineae*, but differ in several aspects of morphology and likely represent an undescribed species.

The diverse skink fauna of New Guinea includes many species complexes (Austin 2006). Preliminary molecular investigations have revealed significant population structure within most widespread species, indicating a greater level of species-level diversity than currently recognized.

The frog fauna of New Guinea is incredibly diverse with more than 350 described species (Amphibian Species of the World Database http://research.amnh.org/herpetology/amphibia/index.php) with 90% endemism (Mittermeier et al. 2003). The rate at which new species are being described is extremely rapid and there is no sign of an asymptote (Fig. 1). Frog diversity in the region is expected to exceed 600 species (Günther 2006). Microhylid frogs are the most diverse group in New Guinea. Our survey included sound recordings of male frogs in order to identify and describe new taxa. Preliminary analyses suggest multiple species new to science in the microhylid genera *Hylophorbus*, *Oreophryne*, and *Xenobatrachus* (Table 1). In addition to the diverse microhylids, the hylid genus *Litoria* is also species rich with a new *Litoria* recently described from Utai (Richards et al. 2006) and another likely undescribed species recorded on our survey.

Notably absent from the Utai region were the common and widespread introduced toad *Bufo marinus* and the skink genus *Carlia*. The latter has a broad range across New Guinea and its members are typically some of the most abundant diurnal lizards in village, garden, and forest edge habitats. *Bufo* and *Carlia* were, however, common in the Provincial capital of Vanimo 75 km NW of Utai on the coast.

Although our survey targeted terrestrial herpetofauna we did collect one specimen of a juvenile *Elseya novaeguineae* (McDowell 1983). In addition, the New Guinea freshwater crocodile (*Crocodylus novaeguineae*) were reported by locals to be in the general vicinity but had been primarily extirpated from the nearby area. Freshwater crocodiles are distributed in lowland to upland areas throughout the island of New Guinea (Hall 1989).

Regional checklists for New Guinean herpetofauna are extremely limited. The only other checklists for the north coast are both from the Kau Wildlife Area (Austin 2006; Read 1998). These limited data allow partial comparative measurements of species richness across a spatial scale of approximately 500 km (Fig. 2). Alpha diversity refers to the total diversity for a given site or region; alpha diversity is 79 at Utai and 67 at Kau. Gamma diversity is the total summed diversity across all sites and the gamma diversity for Utai and Kau is 104. Beta diversity refers to the change in species richness between different sites and is a summed measure of those species unique to two or more compared areas, thus beta diversity reflects the non-overlap of species diversity. The beta diversity between Utai and Kau is 62, meaning that there are 62 species that are found either solely in Utai or Kau (conversely, there are 42 species in common between the two sites). These data suggest that alpha diversity is similar across these two sites and that beta diversity is quite high.

Acknowledgments.—We thank the people of Utai Village for the privilege to conduct fieldwork on their land. In particular we thank P. Tano and Jebed for their hospitality and support. We also thank B. Roy, V. Kula, and B. Wilmot from the PNG Department of Environment and Conservation, and J. Robins from the PNG National Research Institute who provided assistance with research visas and export permits. The Sandaun Provincial Government kindly granted approval for our research in the Province. Partial support for fieldwork by CD at Utai was provided by Conservation International. Steve Richards (South Australian Museum) assisted with frog identifications and provided valuable comments. This manuscript was improved from comments by David Bickford and two anonymous reviewers. All research was carried out under LSU IACUC protocol 06-071. This research was funded in part by National Science Foundation grants DEB 0445213 to CCA.

Literature Cited

Allen, G. R., and D. Coates. 1990. An ichthyological survey of the Sepik River, Papua New Guinea. . Rec. W. Aust. Mus. Suppl. 34:31–116.

Allison, A., D. Bickford, S. Richards, and G. A. Torr. 1998. Herpetofauna. *In* A. L. Mack (ed.), A Biological Assessment of the

Lakekamu Basin, Papua New Guinea, pp. 58–62. RAP Working Papers 9, Conservation International, Washington D.C.

———, and A. E. Greer. 1986. Egg shells with pustulate surface structures: basis for a new genus of New Guinea skinks (Lacertilia: Scincidae). J. Herpetol. 20:116–123.

Austin, C. C. 1998. Phylogenetic relationships of *Lipinia* (Scincidae) from New Guinea based on DNA sequence variation from the mitochondrial 12 rRNA and nuclear c-mos genes. Hamadryad. 23:93–102.

———. 2000. Molecular phylogeny and historical biogeography of Pacific Island boas (*Candoia*). Copeia. 2000(2):341–352.

———. 2006. Checklist and comments on the terrestrial reptile fauna of Kau Wildlife area, Papua New Guinea. Herpetol. Rev. 37:167–170.

Beckon, W. N. 1992. The giant Pacific geckos of the genus *Gehyra*: morphological variation, distribution and biogeography. Copeia. 1992:443–460.

Beehler, B. M. 1993. Mapping PNG's biodiversity. *In* B. M. Beehler (ed.), Papua New Guinea Conservation Needs Assessment, Vol. 2, pp. 193–209. The Biodiversity Support Program,. Washington, D.C.

Bickford, D. 2002. Male parenting of New Guinea froglets. Nature. 4018:601–602.

———, D. J. Lohman, N. S. Sodhi, P. K. L. Ng, R. Meier, K. Winker, K. K. Ingram, and I. Das. 2006. Cryptic species as a window on diversity and conservation. Trends Ecol. Evol. 22:148–155.

Blum, J. P., and J. I. Menzies. 1988. Notes on *Xenobatrachus* and *Xenorhina* (Amphibia: Microhylidae) from New Guinea with description of nine new species. Alytes 7:125–163.

Böhme, W. 2003. Checklist of the living monitor lizards of the world (family Varanidae). Zool. Verhand. 341:3–43.

Brown, W. C. 1991. Lizards of the genus *Emoia* (Scincidae) with observations on their ecology and biogeography. Mem. California Acad. Sci. 15:1–94.

———, and F. Parker. 1973. A new species of *Cyrtodactylus* (Gekkonidae) from New Guinea with a key to species from the island. Breviora 417:1–7.

Burton, T. C. 1990. The New Guinea genus *Copiula* Méhelÿ (Anura: Microhylidae): A new diagnostic character and a new species. Trans. R. Soc. S. Aust. 114:87–93.

Chrapliwy, P. S., H. M. Smith, andC. Grant. 1961. Systematic status of the geckonid lizard genera *Gehyra, Peropus, Hoplodactylus* and *Naultinus*. Herpetologica 17:5–12.

Cogger, H. G. 1972. A new scincid lizard of the genus *Tribolonotus* from Manus Island, New Guinea. Zoologische Mededelingen (Leiden). 47:201–210.

Conn, B. J. 1995. Handbooks of the Flora of Papua New Guinea, Volume III, Melbourne University Press, Melbourne. 292 pp.

de Rooij, N. 1915. The reptiles of the Indo-Australian Archipelago. I. Lacertilia, Chelonia, Emydosauria. E.J. Brill, Leiden. 334 pp.

Dinerstein, E., and E. D. Wikramanayake. 1993. Beyond "hotspots:" how to prioritize investments to conserve biodiversity in the Indo-Pacific region. Cons. Biol. 7:53–65.

Donnellan, S. C., and K. P. Aplin. 1989. Resolution of cryptic species in the New Guinean lizard *Sphenomorphus jobiensis* (Scincidae) by electrophoresis. Copeia 1989:81–88.

Greer, A. E. 1970. The relationship of the skinks referred to the genus *Dasia*. Breviora 348:1–30.

———. 1974. The generic relationships of the scincid lizard genus *Leiolopisma* and its relatives. Aust. J. Zool., Suppl. Ser. No. 31:1–67.

———, A. Allison, and H. G. Cogger. 2005. Four new species of *Loublia* (Lactertilia: Scincidae from high altitude in New Guinea. Herpetol. Monogr. 19:153–179.

Gressitt, J. L. 1982. Biogeography and Ecology of New Guinea, Vol. 2. W. Junk, The Hague. 533 pp.

Günther, R. 2001. The Papuan frog genus *Hylophorbus* (Anura: Microhylidae) is not monospecific: description of six new species. Russ. J. Herpetol. 8:81–104.

———. 2006. Derived reproductive modes in New Guinean anuran amphibians and description of a new species with paternal care in the genus *Callulops* (Microhylidae). J. Zool., London 268:153–170.

Hall, P. M. 1989. Variation in geographic isolates of the New Guinea crocodile (*Crocodylus novaeguineae* Schmidt) compared with the similar, allopatric, Philippine crocodile (*C. mindorensis* Schmidt). Copeia 1989:71–80.

Henty, E. E. 1981. Handbooks of the Flora of Papua New Guinea, Volume II. Melbourne University Press, Melbourne. 276 pp.

King, M. 1984. Karyotypic evolution in *Gehyra* (Gekkonidae: Reptilia). IV. Chromosome change and speciation. Genetica 64:101–114.

———, and P. Horner. 1989. Karyotypic evolution in *Gehyra* (Gekkonidae: Reptilia). V. A new species from Papua New Guinea and morphometrics of *Gehyra oceanica* (Lesson). Beagle 61:169–178.

Kraus, F. 2005. The genus *Nactus* (Lacertilia: Gekkonidae): a phylogenetic analysis and description of two new species from the Papuan Region. Zootaxa 1061:1–28.

———, and A. Allison. 2000. Two new species of *Cophixalus* from New Guinea. J. Herpetol. 34:535–541.

———, and ———. 2002. A new species of *Xenobatrachus* (Anura: Microhylidae) from Northern Papua New Guinea. Herpetologica 58:56–66.

———, and ———. 2004. Two new tree frogs from Normbanby Island, Papua New Guinea. J. Herpetol. 38:197–207.

———, and ———. 2006. Range extensions for reptiles and amphibians along the northern versant of Papua New Guinea. Herpetol. Rev. 37:364–368.

Malnate, E. V., and G. A. Underwood. 1988. Australasian natricine snakes of the genus *Tropidonophis*. Proc. Acad. Nat. Sci. Philadelphia 140:59–201.

Manthey, U., and W. Denzer. 2006. A revision of the Melanesian-Australian angle head lizards of the genus *Hypsilurus* (Sauria: Agamidae: Amphibolurinae) with description of four new species and one new subgenus. Hamadryad 30:1–40.

McDowell, S. B. 1967. *Aspidomorphus*, a genus of New Guinea snakes of the family Elapidae, with notes on related genera. J. Zool. London 151:497–543.

———. 1972. The species of *Stegonotus* (Serpentes, Colubridae) in Papua New Guinea. Zoologische Mededelingen 47:6–26.

———. 1974. A catalogue of the snakes of New Guinea and the Solomons, with special reference to those in the Bernice P. Bishop Museum. Part I. Scolecophidia. J. Herpetol. 8:1–57.

———. 1975. A catalogue of the snakes of New Guinea and the Solomons, with special reference to those in the Bernice P. Bishop Museum. Part II. Anilioidea and Pythoninae. J. Herpetol. 9:1–79.

———. 1979. A catalogue of the snakes of New Guinea and the Solomons, with special reference to those in the Bernice P. Bishop Museum. Part III. Boinae and Acrochordoidea (Reptilia, Serpentes). J. Herpetol. 13:1–92.

———. 1983. The genus *Emydura* (Testudines: Chelidae) in New Guinea with notes on the penial morphology of Pleurodira. *In* K. Miyata and A. G. J. Rhodin (eds.), Advances in Herpetology and Evolutionary Biology, pp. 169–189. Museum of Comparative Zoology, Harvard University, Cambridge.

Menzies, J. I. 1982a. Systematics of *Platymantis papuensis* (Ampibia: Ranidae) and related species in the New Guinea region. British J. Herpetol. 6:236–240.

———. 1982b. The voices of some male *Platymantis* species of the New Guinea region. British J. Herpetol. 6:241–245.

———. 2006. The frogs of New Guinea and the Solomon Islands. Pensoft, Sofia, Bulgaria. 345 pp.

Mertens, R. 1928. Neue Inselrassen von *Cryptoblepharus boutonii* (Desjardin). Zoologischer Anzeiger 78:82–89.

Mittermeier, R. A., C. G. Mittermeier, T. M. Brooks, J. D. Pilgrim, W. R. Konstant, G. A. B. da Fonseca, andC. Kormos. 2003. Wilderness and biodiversity conservation. Proc. Nat. Acad. Sci., USA. 100:10309–10313.

Moody, S. M. 1980. Phylogenetic and historical biogeographical relationships of the genera in the family Agamidae (Reptilia: Lacertilia). Unpubl. Ph.D. thesis, University of Michigan. 373 pp.

Mortiz, C., T. J. Case, D. T. Bolger, and S. C. Donnellan. 1993. Genetic diversity and the history of Pacific Island house geckos (*Hemidactylus* and *Lepidodactylus*). Biol. J. Linn. Soc. 48:113–133.

Myers, N., R. A. Mittermeier, C. G. Mittermeier, G. A. B. da Fonseca, and J. Kent. 2000. Biodiversity hotspots for conservation priorities. Nature 403:853–858.

Mys, B. 1988. The zoogeography of the scincid lizards from North Papua New Guinea (Reptilia: Scincidae). I. The distribution of the species. Bulletin de L'Institut Royal des Sciences Naturelles de Belgique 58:127–183.

O'Shea, M. 1996. A Guide to the Snakes of Papua New Guinea. Independent Publishing, Independent Group Pty Ltd, Port Moresby, PNG. 239 pp.

Rawlings, L. H., and S. Donnellan. 2003. Phylogeographic analysis of the green python, *Morelia viridis*, reveals a cryptic species. Mol. Phylo. Evol. 27:36–44.

Read, J. L. 1998. Reptiles and amphibians of the Kau Wildlife area near Madang: a valuable conservation resource. Science in New Guinea 23(3):145–151

Richards, S. J., P. Oliver, C. Dahl, and T. Burhan. 2006. A new species of large green treefrog (Anura: Hylidae: *Litoria*) from northern New Guinea. Zootaxa 1208:57–68.

Shea, G. M. 1990. The genera *Tiliqua* and *Cyclodomorphus* (Lacertilia: Acincidae): generic diagnoses and systematic relationships. Mem. Queensland Mus. 29:495–519.

———, and A. E. Greer. 1999. Two senior synonyms and a name change for the New Guinea skink *Sphenomorphus stickeli*. J. Herpetol. 33:136–141.

———, and ———. 2002. From *Sphenomorphus* to *Lipinia*: generic reassignment of two poorly known New Guinea skinks. J. Herpetol. 36:148–156.

Womersley, J. S. 1978. Handbooks of the Flora of Papua New Guinea, Volume I. Melbourne University Press, Melbourne. 278 pp.

Zweifel, R. G. 1966. A new lizard of the genus *Tribolonotus* (Scincidae) from New Britain. Am. Mus. Nov. 2264:1–12.

———. 1969. Frogs of the genus *Platymantis* (Ranidae) in New Guinea, with the description of a new species. Am. Mus. Nov. 2374:1–19.

———. 1972. A new scincid lizard of the genus *Leiolopisma* from New Guinea. Zoologische Mededelingen 47:530–539.

———. 1979. Variation in the scincid lizard *Lipinia noctua* and notes on other *Lipinia* from the New Guinea region. Am. Mus. Nov. 2676:1–21.

———. 1980. Results of the Archbold Expeditions. No. 104. Frogs and lizards from the Huon Peninsula, Papua New Guinea. Bull. Am. Mus. Nat. Hist. 165:387–434.

———, J. I. Menzies, and D. Price. 2003. Systematics of microhylid frogs, genus *Oreophryne*, from the North Coast region of New Guinea. Am. Mus. Nov. 3415:1–31.

Herpetological Review, 2008, 39(1), 46–50.

Genetic Evidence for Single Season Polygyny in the Northern Leopard Frog (*Rana pipiens*)

GREGORY A. WILSON[1]
e-mail: Greg.Wilson@ualberta.ca

TARA L. FULTON[1]
e-mail: Tara.Fulton@ualberta.ca

KRIS KENDELL
Alberta Conservation Association, Edmonton, Alberta, T6H 4P2, Canada
e-mail: Kris.Kendell@ab-conservation.com

DANNA M. SCHOCK
Detroit Zoological Society, Royal Oak, Michigan 48067, USA
e-mail: dschock@dzs.org

CYNTHIA A. PASZKOWSKI[1]
e-mail: Cindy.Paszkowski@ualberta.ca

and
DAVID W. COLTMAN[1]
e-mail: David.Coltman@ualberta.ca

[1]*Department of Biological Sciences, University of Alberta*
Edmonton, Alberta, T6G 2E9, Canada

A species' mating system is an important life history characteristic that impacts many aspects of its ecology and evolution. Animal mating systems are differentiated by the number of mates each gender has in a breeding season. Many temperate pond-breeding anurans display what is likely a polygynandrous mating system, which may vary with the operational sex ratio of a population (Halliday and Tejedo 1995; Lode et al. 2004). There is genetic evidence for multiple paternity in some anuran amphibians, where eggs from a single mass are fertilized by more than one male (D'Orgeix and Turner 1995; Laurila and Seppa 1998; Lode and Lesbarreres 2004; Roberts et al. 1999; Sztatecsny et al. 2006). However, the ability of males to fertilize eggs from more than one female during a breeding season has not been demonstrated using genetic techniques. In the course of a larger study, we had the opportunity to look for this phenomenon in a population of Northern Leopard Frogs (*Rana pipiens*).

What is known about the mating system of *R. pipiens* suggests that males may be capable of breeding with multiple females in a single season. *Rana pipiens* is an explosive breeder, and the breeding season can last from a few days to a few weeks (Gilbert et al. 1994 and references therein). If female synchrony is high, there is a limited time when males can fertilize eggs, which could result in a functionally monogamous mating system where only full-sibs and unrelated individuals will be present among the progeny produced in a given year. However, as is true for other anuran species, males sometimes remain at breeding sites after being observed in amplexus (C. A. Paszkowski, unpubl. data), which suggests they may be attempting to find additional mating partners. Males can increase their fitness by producing offspring with more than one female in a breeding season (Arnold and Duvall 1994; Clutton-Brock and Vincent 1991). If males are successful in fertilizing eggs from more than one female, then half-sibs will also be found within a cohort. In laboratory studies, male *R. pipiens* have been observed breeding with more than one female during a reproductive bout, although a decline in fertilization rates may have resulted (Paula Jackman, Environment Canada, pers. comm.).

Rana pipiens is considered a Species of Special Concern by the Committee on the Status of Endangered Wildlife in Canada (COSEWIC 2000) as its numbers have been declining, especially in the western part of its range (Rorabaugh 2005). During a study on the conservation genetics of 14 populations of *R. pipiens* from across central and western Canada, we found that the distribution of genotypes from 31 tadpoles sampled from Belza Pond, Grasslands National Park, Saskatchewan, deviated significantly from linkage equilibrium at 10 of 28 pairwise locus comparisons (see *Results*). This was unusual, as no locus pairs deviated from linkage equilibrium when all populations were considered as a whole (data not shown). In this larger scale study, "populations" were defined as either individuals from a single pond or genetically similar nearby ponds. Deviation from linkage equilibrium can occur if related individuals are present in a sample. *Rana pipiens* lays up to 7000 eggs per breeding season (Corn and Livo 1989), so it is possible that multiple larvae sampled from a pond may be from the same egg mass. Because the Belza population was the only one from which tadpoles were sampled (as opposed to terrestrial juveniles and adults) we predicted that the distinct pattern observed at Belza was a result of sampling some combination of full-sibs (tadpoles likely originating from the same egg mass sharing both parents), maternal half-sibs (tadpoles likely originating from the same egg mass sharing a mother but not a father), and paternal half-sibs (tadpoles likely originating from different egg masses sharing a father but not a mother). To test this we estimated the genetic relationships among the tadpoles from Belza to determine if the sampled individuals shared a familial relationship. If paternal half-sibs are present in the population, this would be the first genetic confirmation that multiple fertilizations with different females in a single breeding season are possible by male *R. pipiens*, and increases our knowledge of the mating system in this species.

Materials and Methods.—Grasslands National Park, Saskatchewan, is located within the mixed-grass prairie ecosystem of North America. Belza Pond is ca. 20 × 20 m, located at 49°09'128"N, 107°31'546"W within Grasslands National Park. It is 500 m from the Frenchman River, but areas between ponds are quite dry and movement of frogs among ponds is likely infrequent and highly dependent upon rare weather patterns that cause high rainfalls. As Belza Pond is spring-fed and in a valley, it generally holds water throughout the year, despite having a maximum depth of 150 cm. The average precipitation at the Val Marie, Saskatchewan, weather station (approximately 10 km from Belza Pond) over a 12-month period from July to the following June from 1990 to 2004 was calculated as 410.19 mm (s.d. 104.47 mm, National Climate Data and Information Archive, Environment Canada 2007; years with missing data were not included in the analysis). Annual precipitation for 2003–2004 was 397.1 mm. Tailclips were collected from 31 tadpoles of *R. pipiens* at Belza Pond on a single date (19 July 2004). Tadpoles were collected opportunistically, but explicit effort was made to sample animals from throughout the pond. Little is known about the Belza population of *R. pipiens*, but at the time of sampling no adults and approximately 100 young of year were observed. Tadpoles were at a minimum of Gosner Stage 42 (where forelimbs erupt, Gosner 1960), and therefore capable of having moved throughout the pond since hatching.

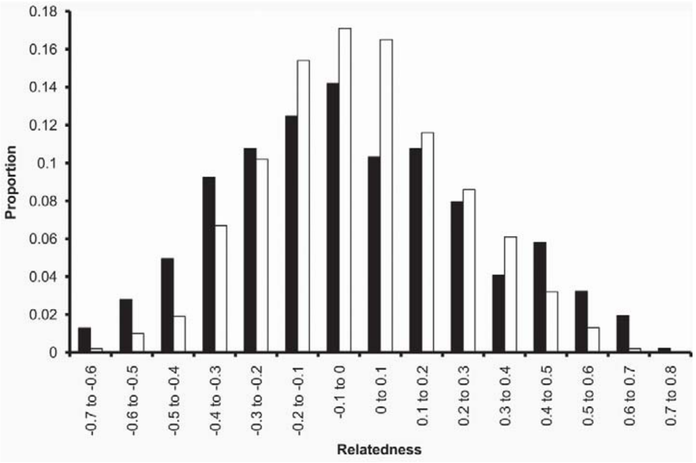


FIG. 1. Frequency histograms of Queller and Goodnight's (1989) relatedness values for the 31 Belza tadpoles (black) and 1000 simulated unrelated individuals (white). Some of the relatedness values are higher in the Belza population than in individuals that are simulated to be unrelated, suggesting the presence of related individuals in our sample.

DNA was extracted using a DNeasy Tissue Kit (Qiagen). For microsatellite analysis, we used loci Rpi100, Rpi101, Rpi103, Rpi104, Rpi107, Rpi108 (Hoffman et al. 2003), Rp193, and Rp415 (Hoffman and Blouin 2004a). Each PCR amplification contained 120 μM dNTPs, 0.27 μM each primer (one of which was dye-labeled), 1.5 U Taq polymerase, 1X PCR buffer (10 mM Tris buffer, pH 8.8, 0.1% Triton X-100, 0.16 mg/mL bovine serum albumin, 50 mM KCl), and ~40 ng genomic DNA in a final volume of 15μL. Rpi101 and Rpi193 were amplified with 2.0 mM $MgCl_2$, while all other PCR reactions contained 2.5 mM $MgCl_2$. PCR cycling conditions were as follows: 1 min at 94°C; 3 cycles of 30 s at 94°C, 20 s at 44°C, 5 s at 72°C; 30 cycles of 15 s at 94°C, 20 s at 45°C, 1 s at 72°C; 30 min extension at 72°C. PCRs were performed on an Eppendorf Mastercycler and electrophoresed on a 3100-Avant Genetic Analyzer (ABI).

All loci were examined for deviations from Hardy-Weinberg equilibrium and linkage disequilibrium using GENEPOP 3.4 (Raymond and Rousset 1995). Queller and Goodnight's (1989) relatedness was calculated with KINSHIP 1.3.1 (Goodnight and Queller 1999). Genotypes for 1000 pairs of unrelated individuals were also simulated with this program by choosing alleles at each locus based on the allele frequencies in the Belza population sample. The relatedness values from simulated unrelated individuals can be compared to those observed in the Belza population to determine if the Belza individuals had higher relatedness values (and were thus more closely related) than would be expected by chance. Individuals were assigned to full-sib families nested within half-sib groups using COLONY 12 (Wang 2004). This program is known to be effective at identifying full- and half-sib groups in a number of species (e.g., Carlsson et al. 2007; A. J. Wilson et al. 2005). The COLONY program utilizes maximum likelihood and allows for errors through allelic dropout and mistyping. Both types of error rates were set to 0, 0.01 and 0.02, and runs were performed at each of these settings with 10 initial random seeds.

All individuals but one were sequenced for an 812 base pair (bp) fragment of mitochondrial DNA, corresponding to bases 27-838 of the NADH dehydrogenase subunit 1 (ND1) gene. The region was amplified using newly designed primers, RpND1F (5' GGT TCA AAT CCC CTT ACT A 3') and RpND1R (5' AGT TGG TCA TAG CGG AAT CGT G 3'), in a 25 μl reaction containing 1X PCR buffer, 2.5 mM $MgCl_2$, 160 μM dNTPs, 0.4 μM each primer, 1 U Taq polymerase, and ~50 ng genomic DNA. Cycling conditions were: 5 min at 95°C; 35 cycles of 1 min at 94°C, 1 min at 54°C, 90 s at 72°C; 5 min extension at 72°C. PCR products were purified using the QIAquick PCR purification kit (Qiagen) and directly sequenced using the forward primer and the BigDye v.3.1.1 (Applied Biosystems) chemistry following the manufacturer's protocol. Individuals with variable sites near the priming region were also sequenced using a reverse primer RpND1R.int (5' TTG AGG ATA CCG AGG CAG AGC 3') for sequence confirmation. Unincorporated dye terminators were removed using the DyeEx 96 kit (Qiagen) and the fragments were resolved using an Applied Biosystems 3730 capillary sequencer. Sequences were analyzed, basecalled, and aligned with ABI Prism SeqScape software v.2.1 (Applied Biosystems).

Results.—The number of alleles in the Belza Pond population per microsatellite locus ranged from 2 to 10, with a mean of 5.25. The Belza population was significantly out of Hardy-Weinberg equilibrium when the exact Hardy-Weinberg test was used (Haldane 1954) and all loci were considered ($\chi^2_{16} = 31.16$, $P < 0.05$). We also observed significant ($P < 0.05$) linkage disequilibria for 10 of 28 locus comparisons. Two of these tests remained significant following Bonferroni correction for multiple tests ($P < 0.0018$). Neither linkage nor Hardy-Weinberg disequilibrium was observed in the 13 other populations of *R. pipiens* we examined from across western and central Canada (data not shown). The distribution of pairwise relatedness values for the Belza population and unrelated individuals simulated from this population's allele frequencies are shown in Fig. 1. On average, unrelated individuals have a relatedness value of 0. This increases to 0.25 for individuals related at the half-sib level, and 0.5 for full-sibs. Twenty-five of the 465 observed pairwise comparisons within the Belza population (5.4%) had relatedness values at the full-sib level or higher. Four of these comparisons (0.9%) were higher than any of the 1000 simulated r-values for unrelated individuals, and 28 (6.0%) were outside of the upper bound of the 95% confidence interval of the distribution of simulated unrelated individuals (-0.425, 0.473), suggesting a high degree of relatedness in these samples. The shapes of these distributions were significantly different from one another (G-test for heterogeneity based on counts grouped by 0.1; d.f. = 14; $P < 0.0005$).

Relatives were common in our sample, as only two individuals from the Belza population did not assign to a nested half-sib group generated by COLONY (full-sib groups 8 and 9 in Fig. 2). The most common arrangement of full-sib groups within half-sib groups (after examining all error rates and initial seeds) and the mitochondrial haplotypes possessed by these individuals is shown in Fig. 2. Other arrangements were similar, and saw either full-sib group 4 merged with full-sib group 3 or full-sib group 2 merged with full-sib group 1. Half-sib group B was the largest, containing 4 full-sib groups and 22 individuals (71% of the sampled population). Full-sib group membership ranged from 1 (groups 1, 2, 8, and 9) to 10 (32% of the sampled population, group 5). Two different mtDNA haplotypes were observed in the Belza population, differing by three synonymous mutations. These mutations all occurred in the first 580 bp of our sequenced region which overlapped the region sequenced by Hoffman and Blouin (2004b), and thus their haplotype numbering system was used. One of the nested half-sib groups (group B) possessed both of the haplotypes observed at Belza. Individuals in full-sib groups 3 and 4 shared haplotype 1 (GenBank accession AF548568) and all other individuals shared haplotype 7 (GenBank AF548575). The individual for which mtDNA sequence was unavailable was assigned to full-sib group 5.

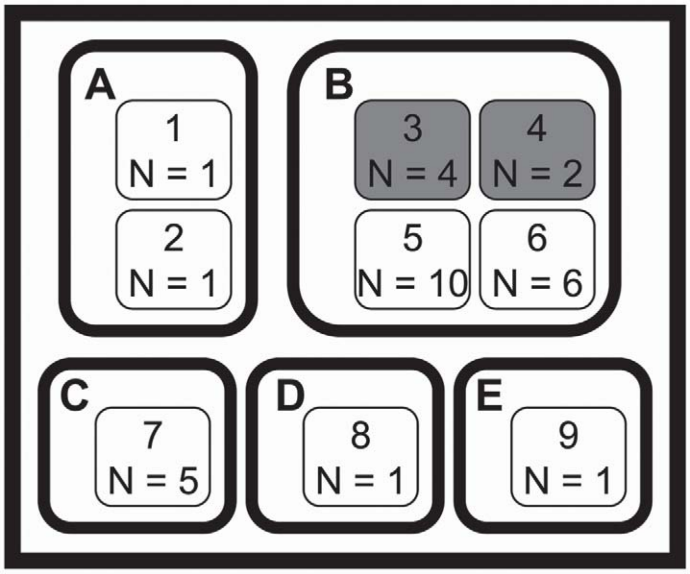


FIG. 2. Partitioned full-sib groups (1–9) nested within half-sib groups (A–E) and the sample size for each full-sib group based on microsatellite data. The largest square represents the sample from Belza Pond, Saskatchewan. Gray full-sib squares possess mitochondrial haplotype 1 and white squares possess haplotype 7.

Discussion.—Sampling tadpoles from the same age cohort allowed us to examine parentage in a population of *R. pipiens* using genetic techniques. Our results show that sib-groups may be commonly encountered among tadpoles. When a sample of individuals from a population consists of related individuals, the genetic material they share is overrepresented. For example, if a pair of full-sibs sharing 50% of their genetic material is sampled, then the shared material will occur in the sample not once, but twice. As populations in both Hardy-Weinberg and linkage equilibrium are assumed to consist of unrelated individuals, the presence of genetic material shared between relatives violates this assumption resulting in the rejection of these hypotheses. Under the assumption that the entire population is randomly mating, larvae analyzed from Belza did not represent a random sampling of the genetic diversity in this population, as suggested by the rejection of the null hypotheses of Hardy-Weinberg and linkage equilibria. This is likely due to the presence of related individuals within our sample, which is also supported by the fact that some pairwise relatedness values in the Belza population are elevated over what would be expected in an unrelated sample of individuals (Fig. 1).

The presence of related tadpoles in our Belza sample allowed us to test if male *R. pipiens* are capable of reproducing successfully with more than one female per breeding season. Sibship analysis identified high membership in full- and half-sib groups. These patterns could be explained by multiple paternity (a female breeding with multiple males) which is known to occur in anurans (D'Orgeix and Turner 1995; Laurila and Seppa 1998; Lode and Lesbarreres 2004; Roberts et al. 1999; Sztatecsny et al. 2006), or multiple fertilizations by males, which has not been documented. Within half-sib group B (Fig. 2), two full-sib groups possess haplotype 1 while the other two full-sib groups possess haplotype 7. The only way this pattern can occur is if a single male bred with at least two females that possessed different mitochondrial haplotypes. This does not mean, however, that all other half-sib groups we observed were paternal half-sibs. If multiple paternity occurs, all members of the half-sib group must share the same mitochondrial haplotype, as it is maternally inherited. However, paternal half-sibs will also possess identical mitochondrial haplotypes if their mothers share the same haplotype. Therefore, if nested full-sib groups within a half-sib group share the same mitochondrial haplotype, it is impossible to determine which parent they have in common. This lack of resolution typified half-sib group A (Fig. 2), where both nested full-sib groups possessed haplotype 7.

Males entering amplexus and successfully breeding with more than one female in succession is the best explanation for our observation of different haplotypes occurring within a half-sib group. However, this is not the only possible explanation for the phenomenon observed. Spermatozoa can survive for a short period of time in aquatic environments, and may be able to diffuse into nearby egg masses if amplexus is synchronous and pairs occur in close proximity (Laurila and Seppa 1998). However, spermatozoa diffusion has been deemed unlikely in other anurans (Lode et al. 2005; Sztatecsny et al. 2006). Consequently, we do not consider diffused spermatozoa a likely explanation.

This first genetic evidence that wild male anurans are able to reproduce successfully with more than one female offers new insight into anuran breeding systems. Some male *R. pipiens* are able to sire offspring from multiple egg masses in a single breeding season. This suggests that remaining at a breeding site after fertilizing one female's eggs is a good reproductive strategy for males and will increase their fitness, as they are sometimes able to mate with an additional female. This finding also has repercussions for the sampling of any anuran population for genetic study if sampling unrelated individuals is the goal. As the majority of tadpoles captured were related at least at the half-sib level, adults should be targeted to ensure a representative sample of the genetic diversity within a population. It should be noted, however, that the large number of half-sibs in our sample was likely exacerbated by the small size of this population and its isolation from other populations, which would both result in a higher proportion of related individuals than would be found in a large population with gene flow between regions.

Our results also apply to the selection of source individuals to be used in translocation efforts to restore or augment anuran populations if maximizing genetic diversity is a management goal (e.g., Goossens et al. 2002; Moritz 1999; G. A. Wilson et al. 2005). The use of subadults or adults as founding stock is more likely to result in a representative sampling of the genetic diversity present in the source population. However, since survivorship of translocated adults can be low (Dodd and Seigel 1991), obtaining subsamples from many egg masses may maximize genetic diversity in translocated animals while minimizing the effect on the source population.

Acknowledgments.—Thanks to R. Sissons of Parks Canada for sample collection. We thank J. Austin and M. Mahoney for their helpful reviews. Funding for this project was supplied by the Alberta Conservation Association. DWC is supported by an Alberta Ingenuity Scholar award.

Literature Cited

Arnold, S. J., and D. Duvall. 1994. Animal mating systems: a synthesis based on selection theory. Am. Nat. 143:317–348.

Carlsson, J., J. R. McDowell, J. E. L. Carlsson, and J. E. Graves. 2007. Genetic identity of YOY bluefin tuna from the eastern and western Atlantic spawning areas. J. Hered. 98:23–28.

Clutton-Brock, T. H., and A. C. J. Vincent. 1991. Sexual selection and the potential reproductive rate of males and females. Nature 351:58–60.

Corn, P. S., and L. J. Livo. 1989. Leopard frog and wood frog reproduction in Colorado and Wyoming. Northwest Nat. 70:1–9.

COSEWIC. 2000. COSEWIC Assessment and Update Status Report on the Northern Leopard Frog *Rana pipiens* (Southern Mountain and Prairie Populations) in Canada. Committee on the Status of Endangered Wildlife in Canada. Ottawa, Canada. 40 pp.

D'Orgeix, C. A., and B. J. Turner. 1995. Multiple paternity in the red-eyed treefrog *Agalychnis callidryas* (Cope). Mol. Ecol. 4:505–508.

Dodd, C. K., and R. A. Seigel. 1991. Relocation, repatriation, and translocation of amphibians and reptiles: are they conservation strategies that work? Herpetologica 47:336–350.

Gilbert, M., R. Leclair, and R. Fortin. 1994. Reproduction of the northern leopard frog (*Rana pipiens*) in floodplain habitat in the Richelieu River, P Quebec, Canada. J. Herpetol. 28:465–470.

Goodnight, K. F., and D. C. Queller. 1999. Computer software for performing likelihood tests of pedigree relationship using genetic markers. Mol. Ecol. 8:1231–1234.

Goossens, B., S. M. Funk, C. Vidal, S. Latour, A. Jamart, M. Ancrenaz, E. J. Wickings, C. E. G. Tutin, and M. W. Bruford. 2002. Measuring genetic diversity in translocation programmes: principles and application to a chimpanzee release project. Anim. Conserv. 5:225–236.

Gosner, K. L. 1960. A simplified table for staging anuran embryos and larvae with notes on identification. Herpetologica 16:183–190.

Haldane, J. B. S. 1954. An exact test for randomness of mating. J. Genet. 52:631–635.

Halliday, T., and M. Tejedo. 1995. Intrasexual selection and alternative mating behaviour. *In* H. Heatwole and B. K. Sullivan (eds.), Amphibian Biology, Volume II, pp. 419–468. Surrey Beatty and Sons, Chipping Norton, Australia.

Hoffman, E. A., W. R. Ardren, and M. S. Blouin. 2003. Nine polymorphic microsatellite loci for the northern leopard frog (*Rana pipiens*). Mol. Ecol. Notes 3:115–116.

———, and M. S. Blouin. 2004a. Historical data refute recent range contraction as cause of low genetic diversity in isolated frog populations. Mol. Ecol. 13:271–276.

———, and ———. 2004b. Evolutionary history of the northern leopard frog: reconstruction of phylogeny, phylogeography, and historical changes in population demography from mitochondrial DNA. Evolution 58:145–159.

Laurila, A., and P. Seppa. 1998. Multiple paternity in the common frog (*Rana temporaria*): genetic evidence from tadpole kin groups. Biol. J. Linn. Soc. 63:221–232.

Lode, T., M. J. Holveck, and D. Lesbarreres. 2005. Asynchronous arrival pattern, operational sex ratio and occurrence of multiple paternities in a territorial breeding anuran, *Rana dalmatina*. Biol. J. Linn. Soc. 86**:**191–200.

———, ———, ———, and A. Pagano. 2004. Sex-biased predation by polecats influences the mating system of frogs. Proc. R. Soc. Lond. B. Biol. Sci. (Suppl.) 271**:**S399–S401.

———, and D. Lesbarreres. 2004. Multiple paternity in *Rana dalmatina*, a monogamous territorial breeding anuran. Naturwissenschaften 91**:**44–47.

Moritz, C. 1999. Conservation units and translocations: strategies for conserving evolutionary processes. Hereditas 130**:**217–228.

Queller, D. C., and K. F. Goodnight. 1989. Estimating relatedness using genetic markers. Evolution 43**:**258–275.

Raymond, M., and F. Rousset. 1995. Genepop (Version 1.2): population genetics software for exact tests and ecumenicism. J. Hered. 86**:**248–249.

Roberts, J. D., R. J. Standish, P. G. Byrne, and P. Doughty. 1999. Synchronous polyandry and multiple paternity in the frog *Crinia georgiana* (Anura : Myobatrachidae). Anim. Behav. 57**:**721–726.

Rorabaugh, J. C. 2005. *Rana pipiens* Schreber 1782. *In* M. Lannoo (ed.), Amphibian Declines: The Conservation Status of United States Species, pp. 570–577. University of California Press, Berkeley, California.

Sztatecsny, M., R. Jehle, T. Burke, and W. Hoedl. 2006. Female polyandry under male harassment: the case of the common toad (*Bufo bufo*). J. Zool. 270**:**517–522.

Wang, J. L. 2004. Sibship reconstruction from genetic data with typing errors. Genetics 166**:**1963–1979.

Wilson, A. J., L. E. B. Kruuk, and D. W. Coltman. 2005. Ontogenetic patterns in heritable variation for body size: using random regression models in a wild ungulate population. Am. Nat. 166**:**E177–E192.

Wilson, G. A., J. S. Nishi, B. T. Elkin, and C. Strobeck. 2005. Effects of a recent founding event and intrinsic population dynamics on genetic diversity in an ungulate population. Conserv. Genet. 6**:**905–916.

Herpetological Review, 2008, 39(1), 50–51.

A Fluorescent Vertebrate: the Iberian Worm-lizard *Blanus cinereus* (Amphisbaenidae)

DAVID P. MAITLAND
and
ADAM G. HART*
Department of Natural and Social Sciences
University of Gloucestershire, Cheltenham, GL50 4AZ, UK

**Corresponding author; e-mail: ahart@glos.ac.uk*

Luminescence is the emission of radiation in excess of thermal radiation (Leverenz 1968). In biological systems there are two main types of luminescence: chemiluminescence (including bioluminescence) which results from biochemical reactions, and photoluminescence (including fluorescence) which results from low-energy photo-excitation (visible light, UV) (Leverenz 1968). Luminescence is found sporadically among different groups of organisms (Herring et al. 1990).

Most of the known bioluminescent organisms are marine, and few are bioluminescent over their entire body surface (Farrant 1997). Bioluminescent terrestrial vertebrates are unknown, but photoluminescencent fluorescence (where UV light is absorbed and re-emitted at longer wavelengths without a detectable afterglow i.e., phosphorescence; Leverenz 1968) is well documented for some vertebrate structures such as parts of bird feathers (Arnold et al. 2002). Among other terrestrial organisms whole-body fluorescence is documented in scorpions (Fasel et al. 1997; Frostet al. 2001), and has not been shown previously in vertebrates (although anecdotal reference is made to this phenomenon in young Western Banded Geckos [*Coleonyx variegatus*] Stahnke 1972; and the blind snake *Leptotyphlops humilis*, Hulse 1971).

We show here for the first time that the Iberian Worm-lizard *Blanus cinereus* (Vandelli) 1797 (Reptilia, Lacertilia, Amphisbaenidae) (Gans 2005) fluoresces blue/green light over its entire body when irradiated by UV radiation.

Materials and Methods.—During our undergraduate terrestrial biology field course a live specimen of the amphisbaenian *Blanus cinereus* was collected (and released after examination) in the Algarve, Portugal, along the sea cliffs of Albuifera at the west end of Praia de Oura Beach during April 2006. Digital photographs were taken using a Canon 1Ds mark II camera (at 16.7 megapixels) in Adobe RGB, 12 bit RAW capture mode at ISO 100 and 400. Macro lenses of 65mm and 100mm focal length and bulb exposure settings were used for fluorescent light capture. White light images were taken using electronic flash. Images were converted and analyzed using Adobe CS2 and RGB pixel values were converted to wavelengths using "*efg's Computer Lab*" "*WaveLengthToRGB*" program in the "*SpectraLibrary.PAS*" available at:

<http://www.efg2.com/Lab/ScienceAndEngineering/Spectra.htm>

Blanus cinereus was found to fluoresce using an excitation wavelength of 368 nm supplied by a portable Ushio ultraviolet lamp, Blacklight Blue (BLB), fitted with a low-pressure mercury-arc lamp code F4T5BLB that emits a narrow band of UV radiation with a sharp spectral peak at 368 nm. Specifications and emission spectra for the lamp are available at:

<http://www.ushio.com/Files/BLBLB.pdf>

Five representative fluorescent regions around the head and body of the worm lizard were sampled for RGB color pixel values (8 bit) using the eyedropper tool in "Info Palette" of Adobe Photoshop CS2. Ten readings were sampled at each of the five regions and the mean (± SD) was obtained from these. Similar measurements were obtained from the central non-fluorescing scale regions, and background (soil) for comparison.

Results.—*Blanus cinereus* fluoresces with a blue-green light between 437 and 550 nm (mean 465 ± 28 nm; ± SD; N = 5) when excited by UV at 368 nm (Fig. 1). Fluorescence between, and around, scale edges occurs over the entire body but is brightest around the head and tail tip (Fig. 1). A fluorescent scale (brille) covers the eyes (Fig. 1). The polished surfaces of some scales might reflect UV, but we were unable to quantify this (Fig. 1b). No phosphorescence was observed.

Discussion.—The fluorophor (Leverenz 1968) responsible for fluorescence in this amphisbaenian is unknown. However, intact scorpion cuticle fluoresces in the range 440–560 nm (Fasel et al. 1997) as a result of at least two fluorophors; beta-carboline and 7-

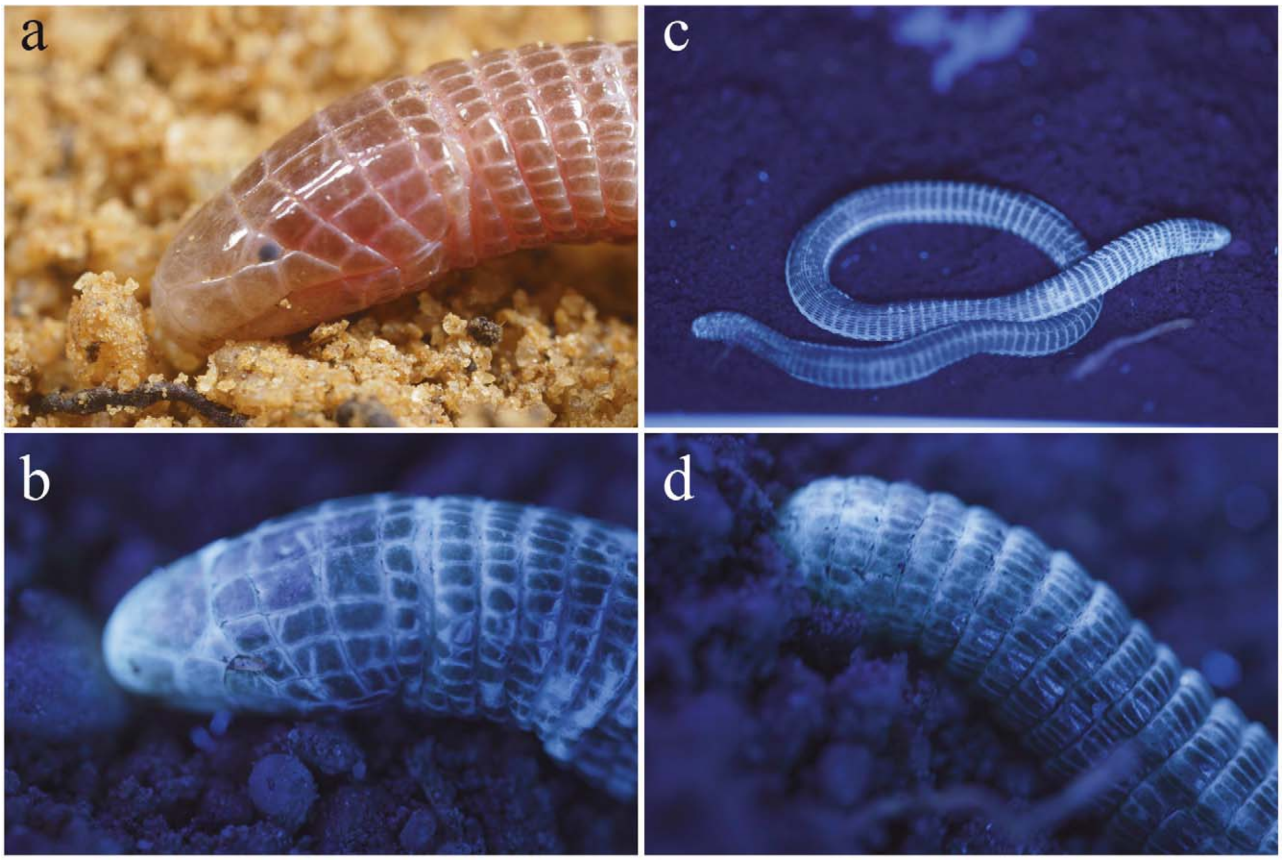


FIG. 1. Fluorescence in the Iberian Worm-lizard *Blanus cinereus* (specimen total body length ~155 mm, 5 mm diameter): a) the head photographed under white light; b) head, c) body, and e) tail, under UV light (368 nm). Photographs by DPM.

hydroxy-4-methylcoumarin, fluorescing at 450 nm and 441 nm respectively (Frost et al. 2001). It is possible, therefore, that scorpions and amphisbaenians possess similar fluorophors.

The function of whole-body fluorescence in *Blanus cinereus* and scorpions is unknown. In scorpions it has been suggested that the fluorophor 7-hydroxy-4-methylcoumarin might act as a block against the UV component of sunlight, because this is a function that coumarins perform in plants (Frost et al. 2001). Another suggestion for scorpions is that the fluorescence might simply be a byproduct of cuticle development in arid adapted organisms (Frost et al. 2001). Indeed, the majority of documented incidences of whole-body fluorescence occurs in invertebrates inhabiting arid environments (Stahnke 1972). Arthropod fluorophors are laid down in the epicuticle as part of the sclerotization process and freshly moulted scorpions and first instar juveniles do not fluoresce (Frost et al. 2001). Thus, whole-body fluorescence in desert-dwelling animals might indicate cuticular adaptations against desiccation and protection against UV radiation; it might not be coincidence therefore, that *Blanus cinereus*, also living in an arid environment and lacking a completely overlapping scale covering, displays whole-body cuticular fluorescence.

Although this report raises many more questions than it answers, further research is warranted. The elucidation of fluorescence in *Blanus cinereus* might lead to the development of new fluorescent tags for DNA and immunological investigations in biochemistry and medicine.

LITERATURE CITED

ARNOLD, K. E., I. P. F. OWENS, AND N. J. MARSHALL. 2002. Fluorescent signalling in parrots. Science 295:92.

FARRANT, P. A. 1997. Colour in Nature: A Visual & Scientific Exploration Blandford, London. 202 pp.

FASEL, A., P. A. MULLER, P. SUPPAN, AND E. VAUTHEY. 1997. Photoluminescence of the African scorpion *Pandinus imperator*. J. Photochem. Photobiol. B: Biol. 39:96–98.

FROST, L. M., D. R. BUTLER, B. O'DELL, AND V. A. FET. 2001. Coumarin as a fluorescent compound in scorpion cuticle. *In* V. Fet and P. A. Selden (eds.), Scorpions 2001. In Memoriam Gary A. Polis, pp. 363–368. British Arachnological Society, Bucks, UK.

GANS, C. 2005. Checklist and bibliography of the *Amphisbaenia* of the world. Bull. Amer. Mus. Nat. Hist. 289:7–130.

HERRING, P. J., A. K. CAMPBELL, M. WHITFIELD, AND L. MADDOCK. 1990. Light and Life in the Sea. Cambridge University Press, Cambridge. 357 pp.

HULSE, A. C. 1971. Fluoresence in *Leptotyphlops humilis* (Serpentes: Leptotyphlopidae). Southwest. Nat. 16:123–124.

LEVERENZ, H.W. 1968. An Introduction to Luminescence of Solids. Dover, New York. 569 pp.

STAHNKE, H. L. 1972. UV light, a useful field tool. Bioscience 22:604–607.

TECHNIQUES

Herpetological Review, 2008, 39(1), 52–54.

A Protocol for Clearing and Double-Staining Squamate Specimens

JESSICA ANDERSON MAISANO
Jackson School of Geosciences
The University of Texas, Austin, Texas 78712, USA
e-mail: Maisano@mail.utexas.edu

Clearing and double-staining of specimens for bone and cartilage is an invaluable tool for the study of skeletal development and morphology, and published protocols exist that are specific to fish (e.g., Song and Parenti 1995; Taylor and Van Dyke 1985) and amphibians (Hanken and Wassersug 1981). Because none is specific to squamates, the purpose of this paper is to describe a modification of the protocol of Potthoff (1984) that has been applied with excellent results to more than 600 squamate specimens with snout–vent lengths (SVL) ranging from 16 mm (neonates) to more than 120 mm (Fig. 1). For time-sensitive steps, data from previous preparations (Maisano 2000) are provided to allow the user to approximate durations for each step according to snout–vent length of the specimen (Figs. 2–7).

Step 1: Specimens must be formalin-fixed or they will disarticulate. Specimens should be measured (SVL), skinned, and gutted. If a dark peritoneum is present, it should be removed as it will not clear. Specimens are then dehydrated in 95–100% ethanol for a minimum of two days. Remaining steps will be accelerated slightly if conducted on a light table.

Step 2: Specimens are stained for cartilage in a solution of 30% glacial acetic acid/70% ethanol, with enough Alcian blue powder added to approximate the color value of window cleaner. Specimens remain in this solution until cartilaginous elements such as the suprascapular and epiphyseal cartilages are visible through the flesh (Fig. 2). Specimens that dessicated prior to fixation will absorb little or no Alcian blue. When preparing immature (small) specimens, it is important to avoid over-staining as this will make it difficult to detect initial centers of ossification and calcification. When preparing mature (large) specimens, cartilage should be stained an intense blue because the longer duration of subsequent steps will partially leach the stain, making it difficult to see elements that persist as un-mineralized cartilage in adult squamates (e.g., portions of the hyoid apparatus, sternum, etc.). Specimens cannot be re-stained for cartilage after they have been stained for bone (Step 5).

Step 3: Specimens are neutralized in a saturated sodium borate solution for at least 12 h. This step can be used to 'pause' the preparation for up to several days if necessary.

Step 4: Specimens undergo initial digestion in a saturated pancreatin (3X or 4X USP specifications) solution of 65% distilled water/35% saturated sodium borate. Specimens remain in this solution until the limb bones are visible (Fig. 3), and the solution should be changed every two days because it becomes increasingly acidic and the enzyme decreases in activity.

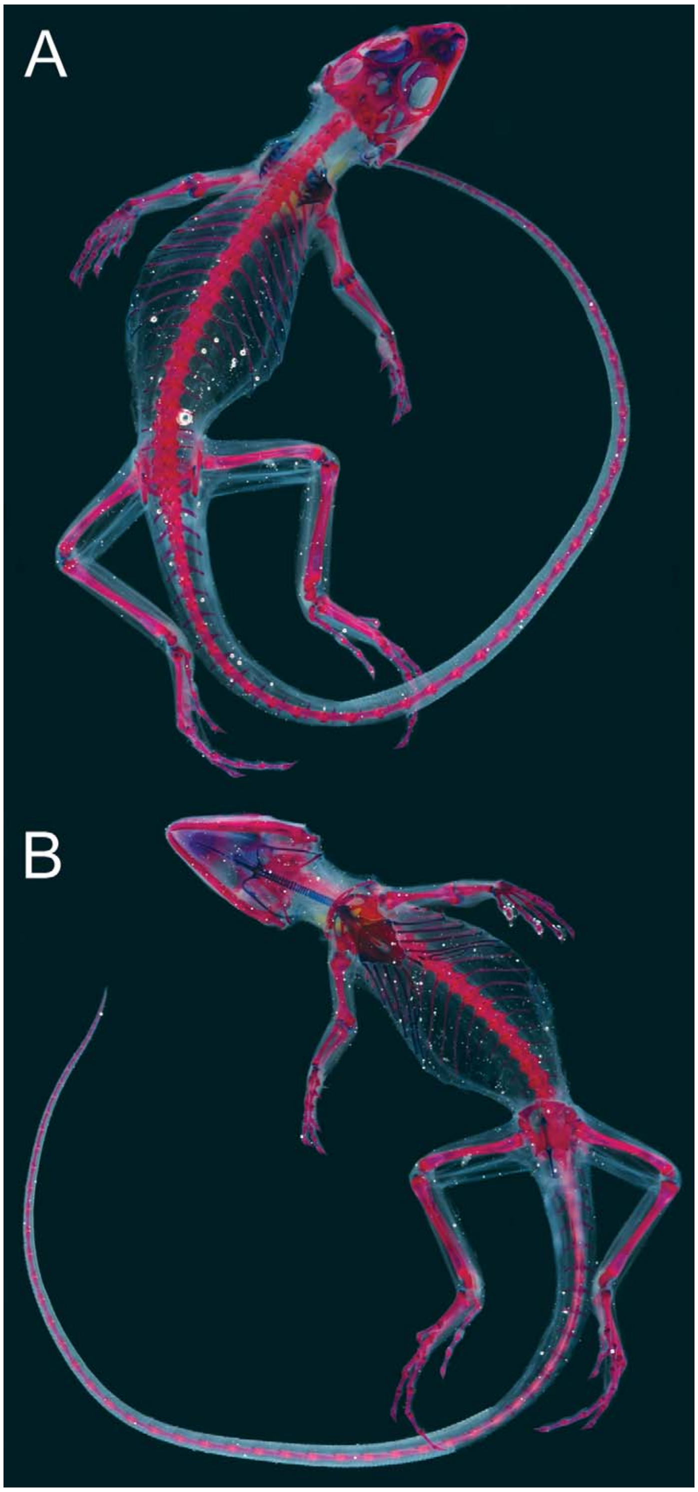


FIG. 1. A) Dorsal and B) ventral views of *Gambelia wislizenii* (Museum of Vertebrate Zoology, MVZ 50268, SVL = 72.8 mm) processed using the protocol described here.

Step 5: Specimens are stained for bone in a 0.75% potassium hydroxide (KOH) solution, with enough Alizarin red powder to make the solution a rich, dark purple. Specimens remain in this solution until their bones are stained a deep, vibrant purple-red (Fig. 4). It is better to over-stain bone rather than under-stain it, as subsequent steps may slightly leach the stain. Specimens may also

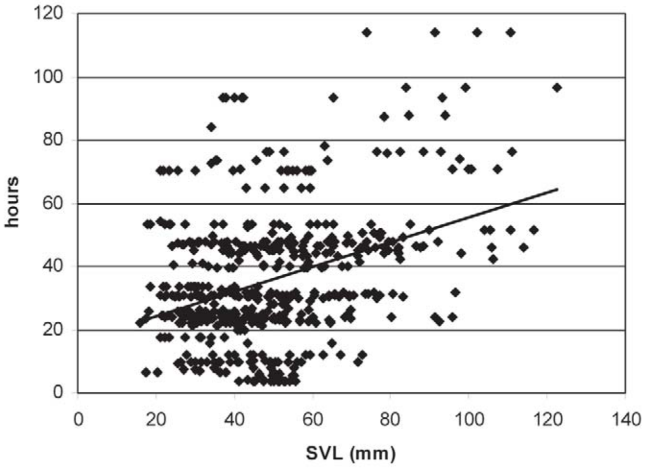


FIG. 2. Time required in Alcian blue stain for cartilaginous elements to become visible through the skin, as it relates to snout–vent length (SVL) (Step 2). Data here and in subsequent figures from Maisano (2000).

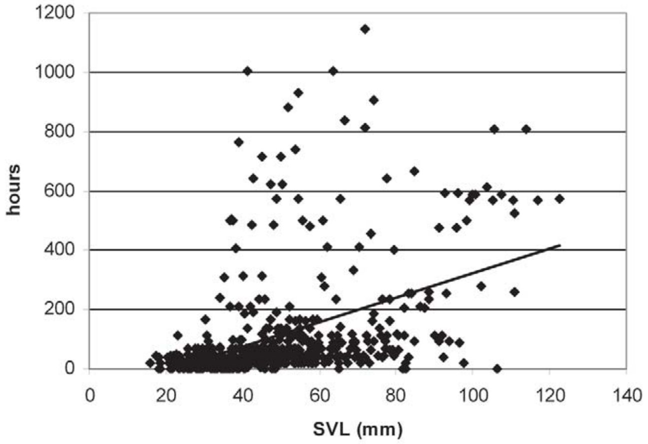


FIG. 5. Time required in second digestion for specimens to become limp, as it relates to SVL (Step 6).

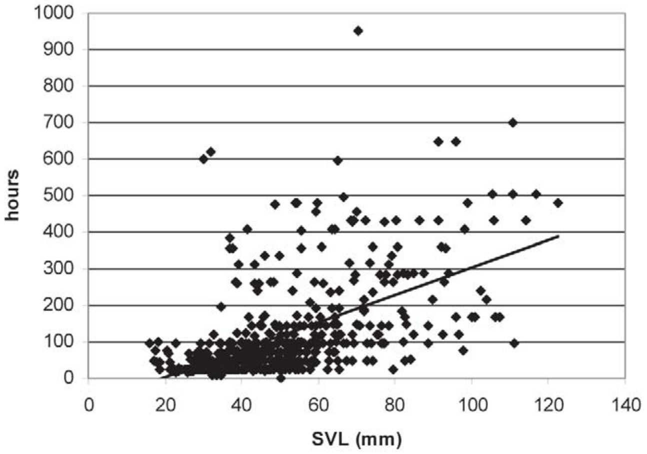


FIG. 3. Time required in first digestion for limb bones to become visible, as it relates to SVL (Step 4).

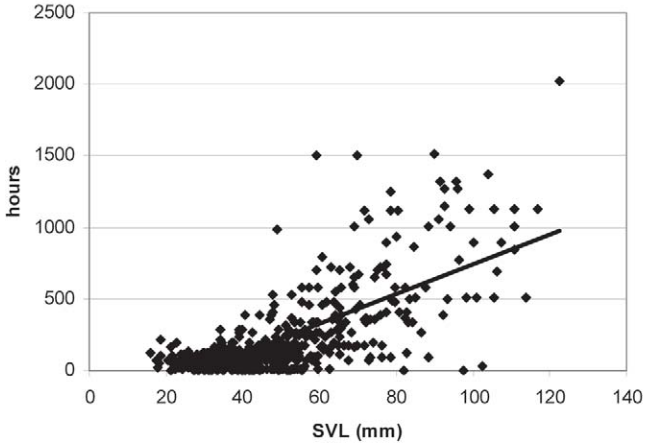


FIG. 6. Time required in first glycerol/KOH clearing solution for all but most resistant areas to clear, as it relates to SVL (Step 7).

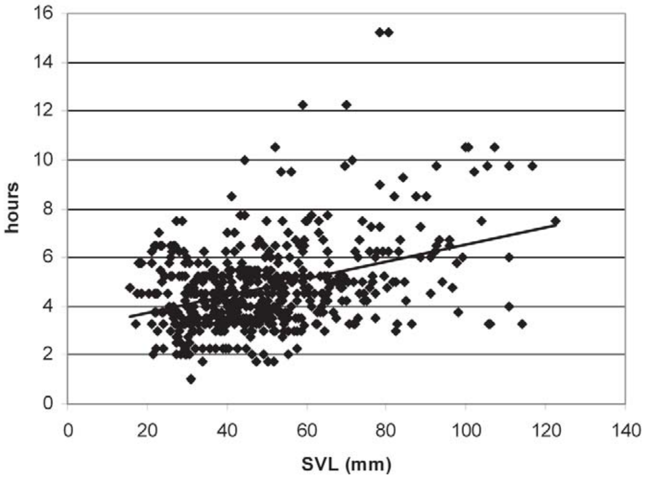


FIG. 4. Time required in Alizarin red stain for bones to stain a vibrant purple-red, as it relates to SVL (Step 5).

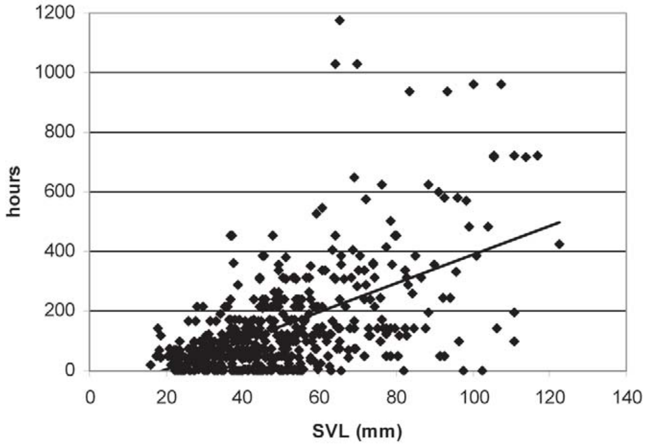


FIG. 7. Time required in second glycerol/KOH clearing solution for clearing to be complete, as it relates to SVL (Step 7).

be re-stained for bone at any time up to the completion of preparation.

Step 6: Specimens are placed in their second and final pancreatin digestion (same solution as in Step 4). Pothoff (1984) recommended that the bulk of the final clearing be accomplished in pancreatin. However, this enzyme substantially weakens the soft tissues, sometimes resulting in disarticulation. Instead, specimens are digested in pancreatin only up to the point where they become limp (i.e., droop when lifted at mid-body) (Fig. 5). Specimens that are already very limp after Alizarin red staining should skip the second digestion as they may begin to disarticulate.

Step 7: Final clearing is accomplished in this step, in two solutions, producing more durable specimens than does prolonged exposure to pancreatin. In fact, specimens will stiffen somewhat during these final steps. Be advised, however, that 0.75% KOH will eventually leach Alcian blue from cartilage, and in very long treatments (large specimens) will lighten Alizarin red staining as well. Specimens are placed in a solution of 30% glycerol/70% KOH until all but the most resistant areas (e.g., neck, base of tail) are clear (Fig. 6). Specimens are then moved to a 60% glycerol/40% KOH solution until clearing is complete (Fig. 7).

Step 8: Specimens are stored in 99% glycerol with a few crystals of thymol added to prevent fungal growth. Specimens should be stored in darkness to prevent fading.

Literature Cited

Hanken, J., and R. Wassersug. 1981. The visible skeleton. A new double-stain technique reveals the nature of the "hard" tissues. Funct. Photog. 16:22–26.

Maisano, J. A. 2000. Postnatal Skeletal Development in Squamates: Its Relationship to Life History and Potential Phylogenetic Informativeness. Ph.D. dissertation, Yale University, New Haven, Connecticut. 1227 pp.

Potthoff, T. 1984. Clearing and staining techniques. *In* H. G. Moser, W. J. Richards, D. M. Cohen, M. P. Fahay, A. W. Kendall Jr., and S. L. Richardson (eds.), Ontogeny and Systematics of Fishes, pp. 35–37. American Society of Ichthyologists and Herpetologists Special Publication 1, Allen Press, Lawrence, Kansas.

Song, J., and L. R. Parenti. 1995. Clearing and staining whole fish specimens for simultaneous demonstration of bone, cartilage, and nerves. Copeia 1995:114–118.

Taylor, W., and G. van Dyke. 1985. Revised procedure for staining and clearing small fishes and other vertebrates for bone and cartilage study. Cybium 9:107–119.

Herpetological Review, 2008, 39(1), 54–55.

Rapid New Protocol for Clearing and Staining of Specimens for Osteological Examination

ERIC H. DIEFENBACHER
and
VALERIE L. WIGGINS
Department of Biological Sciences, Marshall University
Huntington, West Virginia 25755, USA

e-mail (EHD): ericdiefenbacher@yahoo.com
(VLW) : wigginsvl@yahoo.com

Comparative osteology of small vertebrates has been greatly facilitated by the preparation of cleared and stained whole mounts. This procedure is relatively easy and rapid, especially with the most frequently used method: Alcian blue/ trypsin/ Alizarin red-S clearing and staining technique, as modified by Hanken and Wassersug (1981). This protocol requires the specimens to incubate in trypsin for 16 h or overnight. Trypsin, a proteolytic enzyme, breaks down soft tissues around the bone, after which these tissues are further macerated in KOH. This method produces exquisitely cleared and stained whole mounts. This protocol is used by both developmental biologists and herpetologists who study morphological characteristics.

Unfortunately, there are several disadvantages to the trypsin technique. One is the destruction of soft tissues so that the specimen cannot be used for other purposes, such as histology. Another disadvantage is the possibility of over-digestion, especially of delicate parts such as digits, in both large and small specimens. The length of time needed for the complete clearing and staining of a specimen is approximately 10–12 days, another disadvantage.

Materials and Methods.—We set out to develop a technique that would generate cleared and stained whole mounts of preserved amphibian specimens without excessive tissue destruction and in a faster time frame. This technique employs methyl salicylate and eliminates the trypsin and KOH:glycerol stages.

Formalin-fixed specimens were skinned and eviscerated using caution around the delicate digits. Next, specimens were placed into a solution of Alcian Blue Cartilage Stain 8Gx (20 mg/50 ml) in 3:1 ethanol:glacial acetic acid for 24 h, following the Hanken and Wassersug (1981) technique. This step was followed by two rinses in fresh 3:1 ethanol:glacial acetic acid, each lasting 1 h, and a final rinse in 100% ETOH for 2 h. Specimens were then soaked in several changes of distilled water with the last change sitting overnight to dilute out the ethanol. After being re-hydrated, specimens were transferred 0.5% KOH with 1.5 ml of Alizarin red-S stain from a saturated stock solution for 24 h followed by a rinse in distilled water for 2 h. Usually, several rinses were needed to remove excess stain. Next, specimens were placed into 70% ETOH for 12 h then transferred into two changes of 100% ethanol for 1 h each, followed by a third overnight incubation in 100% ethanol. Specimens were then placed into an appropriately sized vial of methyl salicylate which effectively cleared the specimens within minutes.

Results and Discussion.—Several advantages accompany this technique. This protocol takes 5–6 days to complete, with speci-

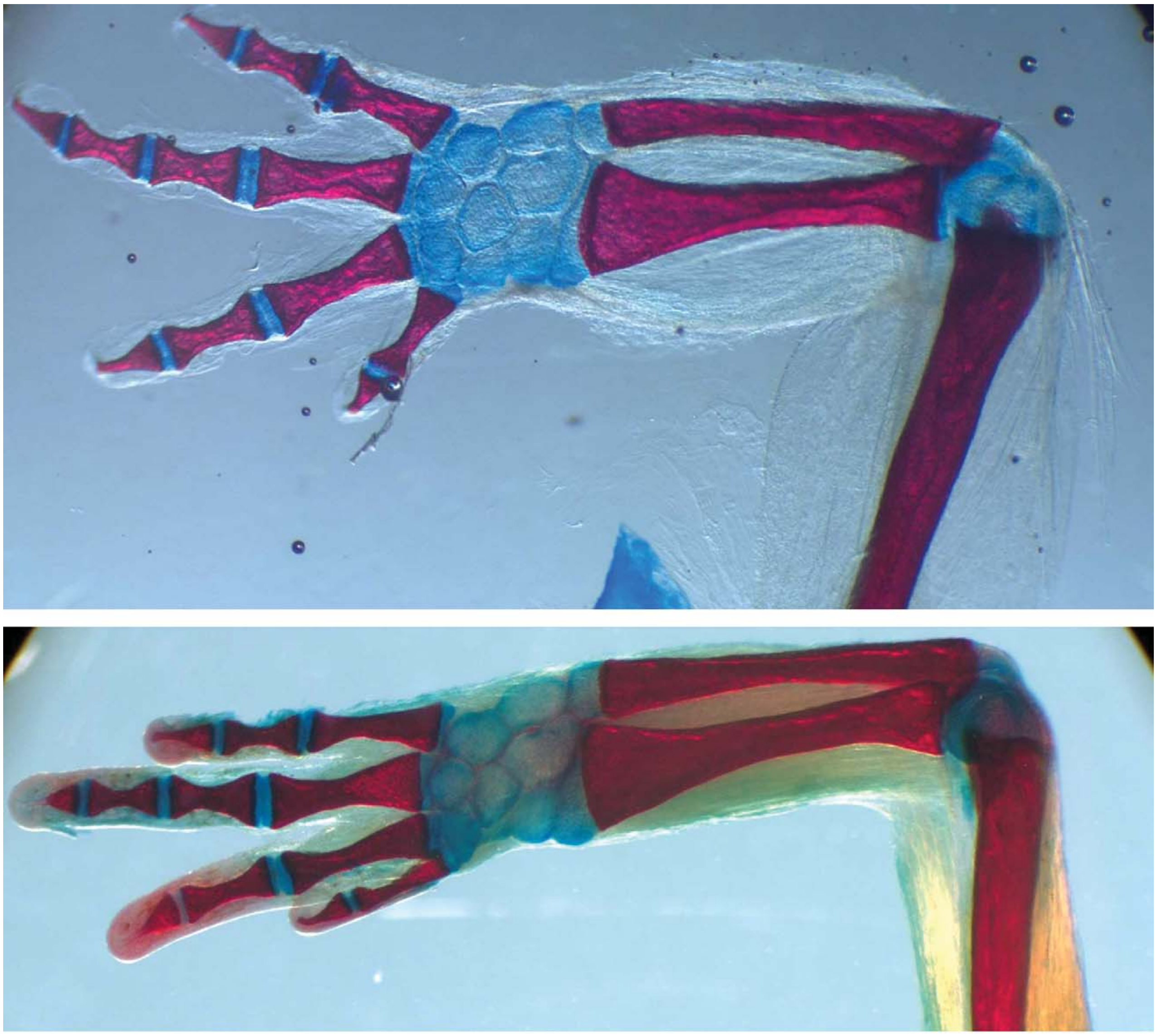


FIG. 1. Photographs showing comparison between a Hanken and Wassersug (1981) cleared and stained specimen (*Eurycea bislineata,* top) and our new approach (*E. bislineata*, bottom).

mens cleared well enough to examine skeletal morphologies (Fig. 1). All of the tissues remain intact and undamaged, and can be removed from methyl salicylate, dehydrated in 100% ethanol, and put back into 70% ethanol for storage. This also allows for histology to be performed on pre-stained specimens with the added benefit of visualizing bone and cartilage. The skinning step can be eliminated for small larvae.

Although this technique works well for specimens under 20 g, it does not clear larger specimens as well as the Hanken and Wassersug (1981) approach. One drop of 30% hydrogen peroxide can be added to approximately 50 ml during the 3:1 rinsings following the Alcian blue staining to aid in clearing any residual pigment left over from the initial skinning and evisceration.

Acknowledgments.—We would like to thank Stanley K. Sessions of Hartwick College for the use of his laboratory and for his advice while modifying this technique.

LITERATURE CITED

HANKEN, J., AND R. J. WASSERSUG. 1981. The visible skeleton. Funct. Photog. 16:22–26.

Herpetological Review, 2008, 39(1), 56–58.

A Fast, Non-invasive Method of Measuring Growth in Tadpoles Using Image Analysis

ANDREW K. DAVIS*
LEAH L. CONNELL
ANDREW GROSSE
and
JOHN C. MAERZ
D.B. Warnell School of Forestry and Natural Resources
The University of Georgia, Athens, GA, USA 30602

**Corresponding author; e-mail: akdavis@uga.edu*

Measuring the performance of anuran tadpoles under a variety of conditions is a common research theme in herpetology. There are a number of variations in how performance is measured including assessing tadpole survival (e.g., Brown et al. 2006; Loman 2004) or time to metamorphosis (e.g., Holbrook and Petranka 2004). The most often-used variable is the growth rate of tadpoles, measured either as the size of the animals at the end of the experiment (e.g., Pahkala et al. 2003) or by subtracting initial mass from final (e.g., Relyea 2004; Relyea and Hoverman 2003; Rosenberg and Pierce 1995). Rarely though, do researchers measure tadpole mass at multiple times throughout the duration of the experiment, likely due to the time required to perform the measurements or the stress induced upon the tadpoles. Indeed, to do this tadpoles must be weighed by removing them from water and blotting them dry, which if not done quickly, can result in mortality.

In this paper, we describe a novel measure for assessing anuran tadpole growth that is simple, inexpensive, requires minimal removal from water, and is nearly directly correlated with tadpole mass. This measure is therefore useful for obtaining repeated measures throughout experiments without harming individuals. Furthermore, this measure allows for bulk assessments of growth on multiple tadpoles at once if time is limited, or if large numbers of individuals are involved.

Our method involves the use of image analysis, whereby digital photographs of specimens are taken and computer software used to measure features of the specimens (Davis et al. 2004; Davis et al. 2007; Davis and Grayson 2007; Davis and Maerz 2007). Variations of this technique are already frequently employed by herpetological researchers interested in tadpole shape (Relyea 2004, 2005; Relyea and Hoverman 2003; Van Buskirk 2002; Wilson et al. 2005), though to our knowledge, this technique has not been used to assess growth rates or directly compared to other measured of size. We therefore set out here to determine if this technique could be used to assess tadpole size and if this measure of size was related to traditional mass measurements. There were two components to this research. First, we examined a set of lab-reared tadpoles that varied in size, using traditional (tadpole mass) and the image analysis size measures. Second, we employed this technique to measure tadpoles that were being raised in an ongoing, unrelated experiment to evaluate its utility in the field.

For our first component, we obtained egg masses of *Rana sylvatica* from Habersham Co., Georgia during February 2006 and divided them into four tubs in the lab where they hatched. Tadpoles were fed ad libitum thereafter with a powder, ground from a 60:40 ratio of rabbit food pellets to Reptomin floating aquatic turtle sticks. Two weeks later there was a natural range of tadpole sizes in each tub. At this point we haphazardly caught single individuals from each tub with dip nets and placed them each in a standard plastic weighing dish filled with water from its tub. The dish was then photographed from above with a Canon Powershot G6 digital camera that was mounted on a mini copy stand at a fixed distance from the specimens (Fig. 1A). Care was taken to ensure that the tadpole was not moving at the time of the photograph (to avoid blurring), and that its dorsal surface was fully in view. After photographing, the tadpole was removed from the dish, blotted dry and weighed on an electronic balance. This was repeated until 58 individuals were processed. Throughout the experiment, we made an effort to capture tadpoles that varied in size. We also photographed a standard metric ruler to provide a calibration image for the image analysis software (see below).

To measure the tadpoles we imported all digital photos into an image analysis program, Fovea Pro (Reindeer Graphics, Inc.), used previously to measure features of herpetofauna (Davis and Grayson 2007; Davis and Maerz 2007). We first calibrated the software using the ruler image so that the pixel-to-millimeter ratio could be retained for the tadpole images. For each tadpole image we digitally selected the entire tadpole (head and tail), which was made easier by the fact that the tadpole was darkly colored on a white background (Fig. 1B). A Fovea Pro measure routine was then initiated, which measured the total surface area of the selection, which in this case was the surface area of the tadpole (43 mm^2 in Fig. 1B). This surface area measure was the unit of 'tadpole size' for our analyses. Once all images were measured we compared the tadpole surface area data (log-transformed) to the mass data (log-transformed) using Pearson Correlation. There was a highly significant positive relationship between individual tadpole mass and our surface area measure ($r = 0.996$, $p < 0.001$; Fig. 2).

Because a tadpole's body is relatively spherical, one might expect a curvilinear (e.g., exponential) relationship between tadpole dorsal area and mass, and therefore be surprised by the strong linear correlation we observed. However, the minimum and maximum tadpole area measurements were 18 and 200 mm^2, and the relationship between dorsal area and volume [mass] is relatively linear between these values. The relationship would only be significantly curvilinear if extended to the origin and out well beyond 200 mm^2, but a tadpole never has zero area or mass, and is not infinitely large.

In the second part of this project, we tested the utility of this technique while conducting an unrelated experiment that involved monitoring the growth of tadpoles within 18 plastic 55-L tubs. Each tub contained 50 tadpoles of *Rana sylvatica,* and our goal was to measure the average size of tadpoles within each tub with minimal mortality. On the day of sampling, the camera and photographing stand were set on a portable table next to the tubs (as in Fig. 1A). Ten tadpoles were dipnetted from the first tub and placed into two large petri dishes (5 in each) along with water from their tub. When the tadpoles settled, a photograph of each dish was obtained as described above. The tadpoles were then released back to their respective tubs. This process was repeated until all tubs had been sampled. During this process we recorded the time to sample (i.e., dipnet and photograph 10 tadpoles) each tub, as well

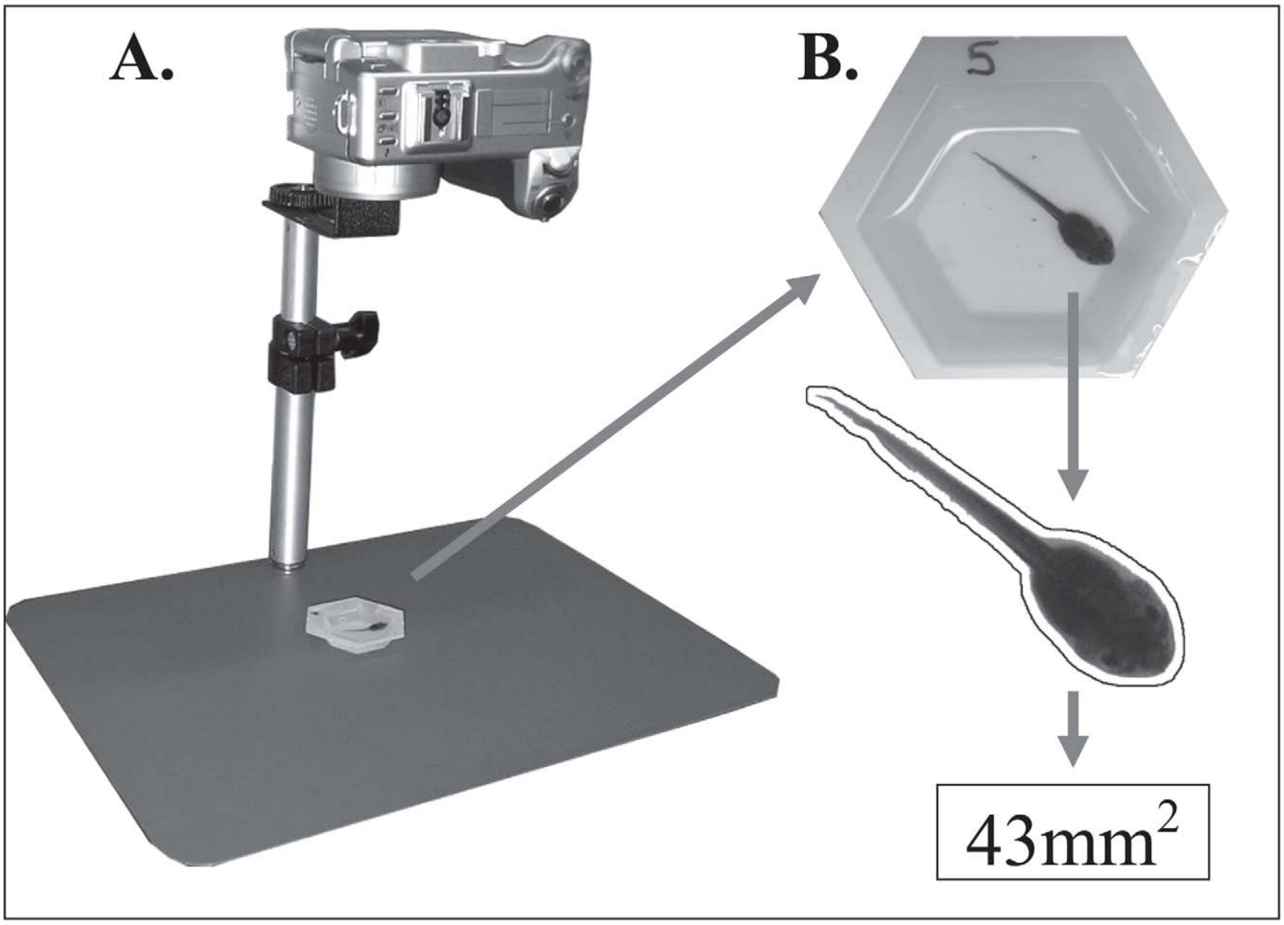


FIG. 1. Camera and photography stand used to photograph tadpoles in the lab and field (A). Tadpoles were held in white plastic weighing boats filled with water during photography. Later, digital images of tadpoles (B) were examined using Fovea Pro software. In the program, the entire tadpole was selected and its dorsal surface area measured in mm^2 based on a previously defined pixel-to-mm ratio obtained from a ruler image.

as the start and end times for the entire session. On average it took us approximately 2 min to sample the ten tadpoles per tub, with all 18 tubs finished in 48 min. In the lab, the same procedures were used to obtain size data from all individual tadpoles in the images as described earlier.

In the procedure described here, tadpoles are dipnetted and immediately placed in a dish of water for photography, they remain in this dish for no more than 2 min, and then are released back into their original water. Thus, tadpoles are subjected to minimal stress because they are never long out of water during the procedure. This technique can be used in the lab or field, and in both situations, large numbers of tadpoles can be quickly and non-invasively sampled, and sampling can be done multiple times over the course of experiments. Further, the measure of size we obtained for each tadpole (its surface area) was nearly perfectly correlated with tadpole mass (Fig. 2). Moreover, although not explored here, it is also possible to generate measures of body and tail length from the tadpole images (or the ratio of both). We also note that while the software used here costs ~ $ US800, there is a freely available image analysis program (ImageJ — http://rsb.info.nih.gov/ij/) that can perform the same tasks as outlined here. Thus, the method we describe is arguably faster and safer than mass measurements, and

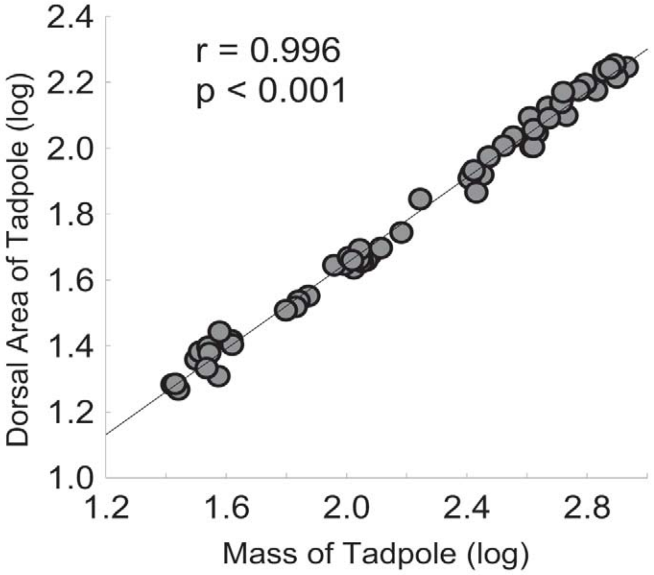


FIG. 2. Comparison of dorsal surface area measurement (log-transformed), obtained from image analysis procedure, and log-transformed mass of 58 lab-reared *Rana sylvatica* tadpoles.

allows for a wider range of data to be measured during experiments.

Acknowledgments.—We thank Daniel van Dijk, Alina Ruiz, and members of the University of Georgia 2006 Herpetology class for field and lab assistance. Tadpoles were captured and raised with approval of the Institutional Animal Care and Use Committee of the University of Georgia.

Literature Cited

Brown, C. J., B. Blossey, J. C. Maerz, and S. J. Joule. 2006. Invasive plant and experimental venue affect tadpole performance. Biol. Invasions 8:327–338.

Davis, A. K., N. Cope, A. Smith, and M. J. Solensky. 2007. Wing color predicts future mating success in male monarch butterflies. Ann. Entomol. Soc. Amer. 100:339–344.

———, B. Farrey, and S. Altizer. 2004. Quantifying monarch butterfly larval pigmentation using digital image analysis. Entomol. Exp. Appl. 113:145–147.

———, and K. L. Grayson. 2007. Improving natural history research with image analysis: the relationship between skin color, sex, size and stage in adult red-spotted newts (*Notophthalmus viridescens viridescens*). Herpetol. Cons. Biol. 2:67–72.

———, and J. C. Maerz. 2007. Spot symmetry predicts body condition in spotted salamanders, *Ambystoma maculatum*. Appl. Herpetol. 4:195–205.

Holbrook, C. T., and J. W. Petranka. 2004. Ecological interactions between *Rana sylvatica* and *Ambystoma maculatum*: Evidence of interspecific competition and facultative intraguild predation. Copeia 2004:932–939.

Loman, J. 2004. Density regulation in tadpoles of *Rana temporaria*: a full pond field experiment. Ecology 85:1611–1618.

Pahkala, M., A. Laurila, and J. Merila. 2003. Effects of ultraviolet-B radiation on behaviour and growth of three species of amphibian larvae. Chemosphere 51:197–204.

Relyea, R. A. 2004. Fine-tuned phenotypes: tadpole plasticity under 16 combinations of predators and competitors. Ecology 85:172–179.

———. 2005. The heritability of inducible defences in tadpoles. J. Evol. Biol. 18:856–866.

———, and J. T. Hoverman. 2003. The impact of larval predators and competitors on the morphology and fitness of juvenile treefrogs. Oecologia 134:596–604.

Rosenberg, E. A., and B. A. Pierce. 1995. Effect of initial mass on growth and mortality at low pH in tadpoles of *Pseudacris clarkii* and *Bufo valliceps*. J. Herpetol. 29:181–185.

Van Buskirk, J. 2002. Phenotypic lability and the evolution of predator-induced plasticity in tadpoles. Evolution 56:361–370.

Wilson, R. S., P. G. Kraft, and R. Van Damme. 2005. Predator-specific changes in the morphology and swimming performance of larval *Rana lessonae*. Funct. Ecol. 19:238–244.

Herpetological Review, 2008, 39(1), 58–63.

Laboratory Protocols for Husbandry and Embryo Collection of *Anolis* Lizards

THOMAS J. SANGER*
PAUL M. HIME
MICHELE A. JOHNSON
JACK DIANI
Washington University in St. Louis, Department of Biology
Campus Box 1229, St. Louis, Missouri 63130, USA

and
JONATHAN B. LOSOS
Department of Organismic and Evolutionary Biology
Harvard University, Cambridge, Massachusetts 02138, USA

**Corresponding author; present address:*
Department of Organismic and Evolutionary Biology
Biological Laboratories, 16 Divinity Avenue, Office 4110
Harvard University, Cambridge, Massachusetts 02138, USA
e-mail: TSanger@oeb.harvard.edu

Anolis lizards, or anoles, are a model system for evolutionary biology (e.g., Emerson 2002; Losos 1994), behavioral and physiological ecology (e.g ., Huey et al. 2003; Irschick and Garland 2001; Lovern et al. 2004; Stamps 1983), community ecology (e.g., Pacala and Roughgarden 1985; Schoener 1968), toxicology (e.g., Burger et al. 2004), and physiology and neuroendocrinology (e.g., Greenberg 2003; Wade 2005). Because the genome of *Anolis carolinensis* has recently been sequenced (http://www.genome.gov/Pages/Research/Sequencing/SeqProposals/GreenAnoleLizardAmericanAlligatorSeq.pdf), work on this genus is expected to expand as it becomes more accessible to new disciplines such as developmental and statistical genetics, comparative genomics, and the biomedical sciences. For this reason it is imperative that methods for working with *Anolis* be developed to facilitate comparisons between studies and assure the ethical treatment of these animals as they are used by researchers not accustomed to working with reptiles.

The goal of this manuscript is to describe methods for the maintenance of captive breeding colonies of *Anolis* species. The general characteristics of a species that must be considered to maintain healthy breeding populations include its availability (in the wild or the pet trade), physiological needs (e.g., temperature, humidity, nutrition), and sociality (density of housed individuals) (Greenberg 1992). These factors also must be independently considered for the care of juveniles to assure their proper growth and development. In addition to species-specific factors, practical limitations such as available space, number of species to be housed together, and available resources (incubators, cage washers, climate control systems, etc.) must be considered when developing protocols for the maintenance of animals in captivity. These likely vary greatly from institution to institution. A broad perspective on the general use of reptiles for research can be found in Greenberg (1992) and Pough (1991) and on reptilian egg incubation in Deming (2004).

Here we describe the detailed methods we have found to be successful for the care and maintenance of 13 *Anolis* species from southern Florida and four Caribbean islands that inhabit a wide range of microhabitats. While we have worked primarily with *A. sagrei*, *A. carolinensis*, and *A. cristatellus*, we also have experienced success breeding *A. chlorocyanus, A. coelestinus, A. cybotes, A. distichus, A. evermanni, A. grahami, A. gundlachi, A. krugi, A. lineatopus*, and *A. valencienni*. Lovern et al. (2004) provided protocols for *A. carolinensis*, the northernmost species of the genus. We have modified these protocols to fit the needs of an additional 12 species. The methods we describe here are readily amenable to larger-scale laboratory settings where large numbers of animals from different species are maintained. We successfully raised all species through the F1 generation except for *A. cristatellus*, which we raised through the F2 generation. We developed these methods under the guidance of Washington University's animal facilities management (J. Diani), veterinary services, and the Institutional Animal Care and Use Committee. Based on explicit calculations of the effort and expense needed to care for these lizards (space, lighting, climate control, basic daily care, etc.), we were charged $0.16 per lizard per day to maintain these lizards in our animal care facility.

The key factor in developing protocols for the maintenance of captive breeding colonies is an understanding of a taxon's natural history. Anoles are remarkably variable in their natural history, ecology, behavior, and physiology. They inhabit a broad variety of habitats from hot xeric semi-deserts to cool montane rainforests, ranging from the equatorial forests of South America throughout the Caribbean Islands and into southeastern North America. Caribbean anoles, including populations found in the southern United States, breed from early spring through late-summer depending on the temperature and humidity of a given year (Licht and Gorman 1970). Mean field body temperatures range from 21.0–33.0°C (Clark and Kroll 1974). Some species narrowly regulate their body temperature by basking, whereas others are thermoconformers; furthermore, within a species, mean body temperature changes with altitude and season (Hertz 1981; Hertz and Huey 1981; Huey and Webster 1976; Ruibal 1961). Although fewer field data are available, similar observations are made regarding interspecific variation among anoles in their hydric environments (Hertz 1980, 1992), size at reproductive maturity, hatchling size, and growth rate (Kobayashi et al. 1983; Michaud and Echternacht 1995). In some cases, physiological differences observed among populations or even species may represent phenotypic plasticity, rather than genetic differences (e.g., Wilson and Echternacht 1987, 1990). Given these many and varied differences, there is no one-size-fits-all prescription for caring for and raising anoles; rather, to the extent possible, husbandry needs to be tailored to the biology of each species.

For the following discussion, it is important to note that many of the species we have maintained in captivity occur primarily in open, sunny habitats (but see Interspecific Variation below). Such species often thermoregulate actively and maintain relatively high body temperatures; moreover, they tend to be less sensitive to water loss than many other anoles (Hertz 1981, 1980, 1992). Because the species we have used in our research naturally occur in the Caribbean and southeastern United States, we chose mean conditions for humidity, moisture, and light based on climatic data in those regions for the months of peak breeding, April to June (for Florida and southeastern United States: Hamlett 1952, Lee et al. 1989, TJS pers. obs.; for the Caribbean: Jenssen and Nunez 1994).

HUSBANDRY AND EGG COLLECTION

We have collected adults of each species from the wild or, for species found within the United States, purchased them from professional reptile vendors. Select species are also available from the pet trade; however the history and genetics of these individuals are not typically known. Occasionally, new animals in the lab do not breed or deposit eggs at first, but after two weeks, individuals generally show no visible signs of distress (as described in Greenberg 2002, 2003) and breed regularly. Wild-caught anoles commonly host parasitic mites, often found on the dewlap, groin, base of the tail, or axillae. Although specific host-parasite relationships likely exist between mite and lizard species from the same island, lizards may be more severely affected by novel parasites introduced from other locations. Because we do not have space to house each lizard species in its own room, lizards from different islands must be kept together. Consequently, upon arrival to the animal care facility, we remove all mites using forceps and treat the lizards with commercial de-miting solution as described by the manufacturer (Reptile Relief, Natural Chemistry). Endoparasites may be problematic as well. For example, we have discovered infestations of endoparasitic worms as tumor-like growths on the joints and jaws in wild-caught *A. cybotes* and *A. cristatellus*. We immediately cull individuals from our population as soon as a growth is detected to limit the spread of this parasite between individuals. Sarcophagic fly infestations are also common in some populations of wild-caught anoles (Irschick et al. 2006). We do not prophylactically treat for viral or bacterial infections.

To establish breeding groups, we kept three or four adult females (depending on size) and one adult male together in a standard rat cage (42 cm length × 27.5 cm width × 21 cm height, Ancare Corp.) with oak sticks collected from the outdoors to provide perches (Fig. 1). Before using the sticks as perches, we sterilize them with one cycle of a standard cage washing machine at 180°C. These sex ratios are within the range of those found in the wild for *A. carolinensis* (Jenssen and Nunez 1998) and in many species, several female territories occur within the territory of an adult male. This creates a breeding regime in which juveniles from each cage are half-sibs in genetic analyses, with known father and unknown mother. However, anoles are known to store sperm (Fox 1963; Licht 1973) for many months, so if females were reproductively mature when captured, one cannot assume that the male in the cage fathered the offspring; molecular testing is needed to verify that conclusion.

To prevent escape, we place screen mesh inserts in the cage tops rather than the cloth filter tops typically used when housing mice (New York Wire, mesh 16 × 84 inches). We cover cage bottoms with synthetic cage carpet (ReptiCage Carpet, ZooMed). Cage carpet is readily available, can be easily cleaned and sterilized, and is reusable. The carpet also holds moisture raising the humidity within each cage. We have avoided the use of gravel or sand as a cage substrate because we found that smaller individuals ingest these substrates and become impacted. We discovered that wood chips and sphagnum moss promoted fungal growth after a short time, needed to be changed often, and are not reusable. We clean and sterilize cage carpet and perches approximately once each

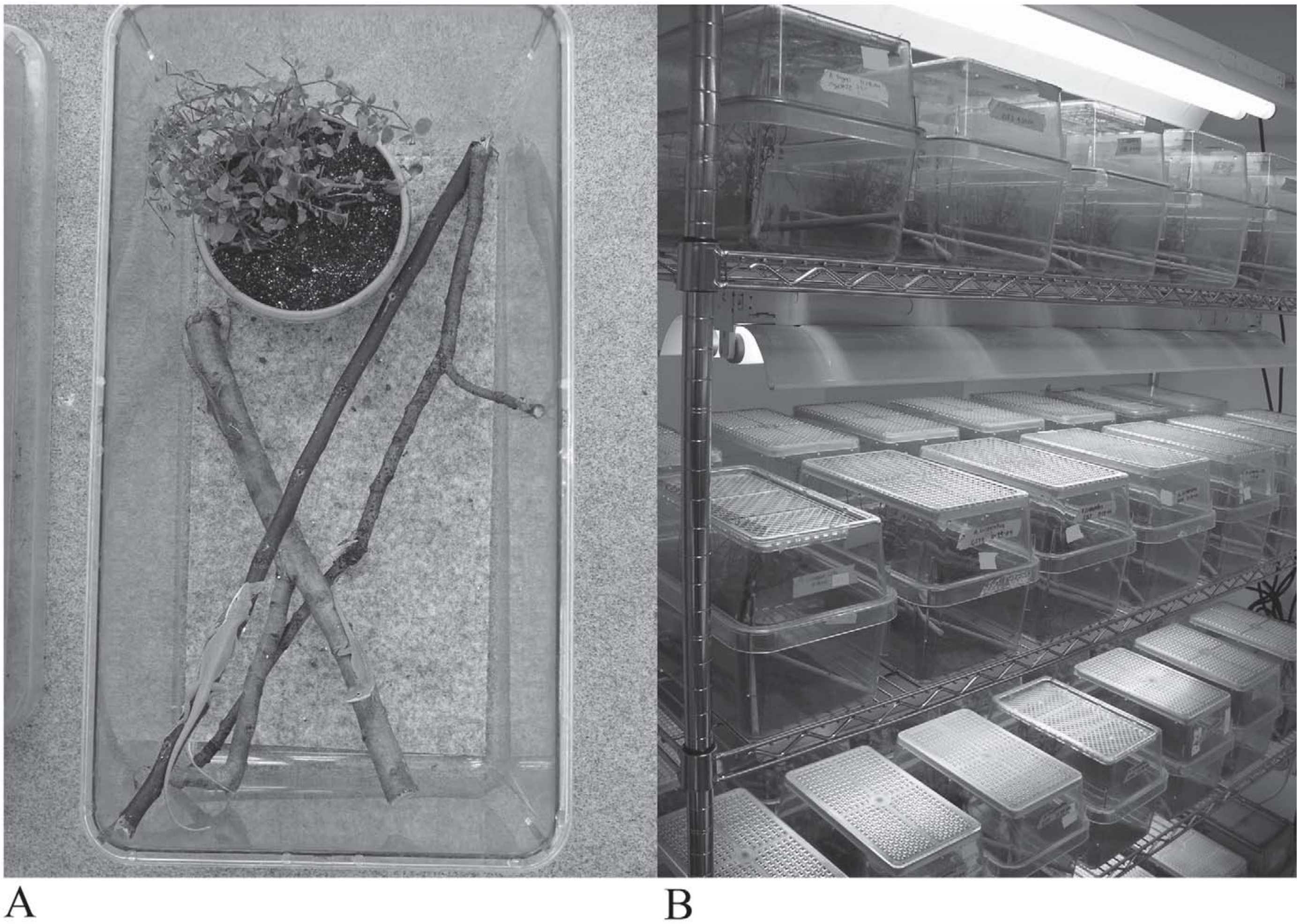


FIG. 1. A) A typical breeding cage for *Anolis*. Note the potted plant, cage carpet floor, sticks to act as perches, and several females held with one male. B) A typical rack of juvenile cages. Note the proximity of the UV lights over each row of cages.

month by soaking them in T.B.Q. disinfectant solution (Steris) while cages are washed using a cage washing machine. Anoles may exhibit "cage-novelty induced stress" when placed in unfamiliar conditions and thus perches and carpet should be exchanged separately so as not to return a lizard to a completely novel cage setting after cleaning (Greenberg 1992; Morgan and Tromborg 2007). Opaque cage dividers separate the cages to limit intraspecific male aggression. In cages not separated by cage dividers, males tend to develop open sores on their snout as they attempt to fight with males in adjacent cages. We recommend labeling each cage with the species name, locality of the collection, origin (wild caught or captive bred), date entered into the facility, number of individuals in the cage, and when applicable, age of the individuals and parentage information.

We maintain room temperature, humidity, and photoperiod using an automated climate control system (METASYS, Johnson Controls), cycled seasonally to mimic natural coastal Caribbean wet and dry season fluctuations. During the summer months room temperature cycles daily with a 28°C day, 25°C night and a constant 60–65% humidity. We mist each cage twice daily, raising the humidity within each cage to approximately 85% and supplying the animals with substantial drinking water. Relatively low humidity (less than 35%) is known to retard ovarian growth in *A. carolinensis* (Summers 1988). We also keep a small potted plant in each cage to help maintain relatively high levels of humidity between misting times. During the winter, we drop the room's humidity to approximately 50% and lower the temperatures by 1–2°C. The cages are still misted twice daily during this time. We keep light cycles at 13 h light/ 11 h dark cycles during the summer months and gradually shift to 11 h light/ 13 h dark cycles from early October though late March, approximately simulating the light cycles of the Caribbean. We light the room with both standard fluorescent ceiling lights and 15–20W fluorescent UV bulbs (ReptiSun 5.0) set directly over each cage. To simulate dawn and dusk we turn the ceiling lights on or off 30 minutes before or after the UV lights respectively. In the absence of an automated climate control system these conditions can be maintained with cost-effective, widely available alternatives such as electric timers, stand-alone humidifiers, and heat lamps (see Bartlett and Bartlett 2001; Hunziker 1994; Lovern 2004 for several more alternatives).

We feed adult anoles a diet of crickets twice weekly, sized appropriately for the species. Crickets are purchased from Timberline Fisheries Corp. (Marion, Illinois) sold in standard English units of 0.25," 0.5," or 0.75." To supplement the lizards' nutrition we dust crickets with a 1:1 mix of commercial vitamin supplement and a calcium additive (Rep-Cal brand Herpvitive and Calcium Supplement) every other feeding. To maintain our cricket colony, we feed the crickets a diet of Laena Poultry (Purina) *ad libitum*. Be aware that some cricket suppliers regularly feed their animals diets that will be transferred to the lizards and may influence a specimen's growth or experimental results. For example, some cricket suppliers prophylactically administer antibiotics to their crickets. During our study of skeletal development (TJS), we found signs of the calcium chelator, tetracycline, in developing long bones. Tetracyclines are known to retard bone growth in many mammals, and thus we may have inadvertently compromised our experiment on bone growth by indirectly feeding it to lizards. We recommend inquiring into the treatment of lizard food when it is purchased from a commercial supplier.

To facilitate the collection of eggs, we supply each cage with a small leafy plant potted in a disposable plastic container (*Ziplock Snap and Seal*™, medium bowl or equivalent). Anoles lay one egg from alternating oviducts every one to four weeks (Andrews 1985; Andrews and Rand 1974; Hamlett 1952). We collected eggs every one to three days by removing the plant from the cage, removing the plant from the pot and thoroughly sifting through the root ball and potting substrate. Female anoles lay eggs at all depths in the pot. To ensure that eggs are not missed during collections, we completely remove the potting substrate from the container and search it by hand. Because female anoles can retain their eggs during times of drought (Socci et al. 2005; Stamps 1976), we remoisten the soil after each egg collection. When females are first brought into captivity, they may take some time to begin laying eggs in the potted plant. Initially, eggs may be laid on the cage bottom where they rapidly desiccate.

Viable eggs are generally white and range 0.5–1.5 cm in length. We clean eggs of excess soil using forceps or by hand and transfer them to a standard tissue-culture dish (100 mm × 20 mm) packed with moist, coarse-grained vermiculite, 1:1 vermiculite to de-ionized water by weight. We then cover the dish with its lid, secure it with *Fisherbrand* labeling tape and incubate at 27°C and ~75% humidity for 10–14 days. At this time, we remove the lid and cover the dishes with fiberglass screening to facilitate gas exchange (New York Wire, mesh 16" × 84"). We cut the screening into 15 cm squares and secure it around the circumference of the dish with rubber bands. We restack the dishes in the incubator in such a way that each dish acts as the cover for the dish below it. The top dish is loosely covered with its original lid and the weight of each dish is recorded. We then reweigh the dishes every one to two days and rehydrate them with drops of de-ionized water until the dish is returned to its initial weight. Additionally, to limit the evaporation from the culture dishes, we keep a pan of de-ionized water in the incubator.

Eggs of many squamates are highly permeable and actively exchange water and gas throughout embryonic development. Eggs not relatively free of debris will often desiccate or the embryo will die leading to fungal growth on the egg's surface (PMH, pers. obs). A fine balance also exists between the water within the egg and that in the surrounding substrate (Ackerman and Lott 2004). If the substrate is drier than the egg, the egg will lose water and desiccate. Inversely, if the egg is drier than the substrate, water will flood the egg, possibly damaging the embryo due to increased pressure. While these scenarios represent the extremes of a vast continuum, water balance in reptilian eggs is known to influence many developmental processes (e.g., Packard et al. 1999, 2000) and care should be taken to keep these conditions stable between species and throughout the period of development. Anole species living in different environments probably have adaptively differentiated in their ability to withstand humidity and desiccation, but this has been little studied (Andrews and Sexton 1981).

Care for Juveniles

For the 13 *Anolis* species we have bred in the laboratory, eggs hatch between 25 and 42 days from the date of laying. Upon hatching, we remove juvenile lizards from the culture dish and place them in a standard mouse cage (26 × 20.5 × 15.5 cm, Ancare Corp.) with conditions as described above. We immediately mist hatchlings with water (for drinking and to remove vermiculite stuck to their bodies) and feed them flightless *Drosophila* maintained in a stock culture. As individuals mature, we gradually transfer them to a standard cricket diet. Crickets are chosen by size: smaller lizards, ~2.5cm to 4.0cm SVL, are fed smaller crickets, "pinhead" to 0.25". We later transfer juvenile anoles larger than ~4.0 cm to a diet of larger crickets, 0.5", by adding crickets of both sizes to the cage for approximately one month. We only feed the largest crickets, 0.75", to cages with adult males greater than 5.0 cm SVL. Because we occasionally observed crickets feeding on juvenile lizards, we remove uneaten crickets from the cages within several hours of feeding.

Lizards raised without substantial social interaction throughout juvenile development rarely breed during adulthood (MAJ, pers. obs.); however, housing juveniles individually may be desirable for some studies (e.g., tracking individual growth). In this case, juvenile lizards can be allowed limited social interaction by removing cage dividers to provide visual access to the neighboring cages. Preferably, juvenile lizards would be housed in groups of three to five individuals per rat cage when individual records are not necessary or when marking individuals (e.g., Ferner 1979; Fisher and Muth 1989; Johnson 2005) will not interfere with an experiment. In this case however, the more dominant lizards tend to capture more food, leading to size disparity within each cage (PMH, pers. obs). As animals become dominant, especially males, they can be removed from the group cage and housed separately.

Captive-bred juvenile lizards often face problems during shedding. Lizards often encounter the most difficulty shedding the skin around the head, legs, and cloacal regions. On the head, unshed skin can block eyes, cover nostrils and impede feeding. Around the legs, excess skin can constrict and reduce circulation leading to the occasional loss of limb elements. Unshed skin around the cloaca can cause complications during excretion and promote infection. To prevent these complications we often removed unshed skin on juvenile and adult lizards by moistening the skin with warm water and then gently removing it with fine forceps. Care should be taken not to injure sensitive areas while removing unshed skin.

Embryo Collection

Anolis embryos of all stages can be dissected from eggs under a dissecting microscope. To do so, we remove eggs from their culture dish and submerge them in phosphate buffered saline solution (PBS; Sambrook and Russell 2001). While illuminating the egg from the side, we then position the egg with the embryo upwards by locating its shadow beneath the shell. Using #5 watchmaker's forceps, we make a small incision outside of the darkened area and extend it, with shallow incisions, across the top of the embryo. Folding the shell away, the embryo is then visible lying on its left side within the yolk and can be easily removed from the yolk and surrounding amnionic membrane. For young embryos, adding a drop of 10% bovine serum albumin to the Petri dish may help in the removal of extra-embryonic membranes. We preserve young embryos by immediately submerging them in 4% paraformaldehyde PBS in a plastic centrifuge tube, incubating overnight at room temperature, and finally transferring them to 70% ethanol. This method of fixation is useful for many basic histological techniques such as gross examination of both soft and hard tissue. Additional measures may be needed for analysis of gene expression or other molecular or immunochemical assays. To assure the humane treatment of embryos approaching hatching, we euthanize these animals with an IP injection of xylazine (20 mg/ml) prior to fixing. Xylazine (Rompun**,** Bayer) is a nonnarcotic anesthetic commonly used on mammals. We fix late-stage embryos in 10% buffered formalin for 24 h, and then move them to 70% ethanol. We have observed that *Anolis* embryos tend to adhere to "soft" polystyrene tubes during fixation and cannot be removed without damage. We prefer to use common polypropylene 15 ml conical tubes (i.e., Corning, FisherBrand) when preserving embryos for use in histological preparations or 2 ml centrifuge tubes when preserving embryos for long term storage.

Interspecific Variation

Although the methods described above have been successful for the majority of *Anolis* species we have examined, interspecific differences have required minor adjustments in our protocol. For example, most well-studied species of Caribbean *Anolis*, including the majority of species for which these methods have been utilized, occur in sunny, open areas. The temperature and humidity simulated in our animal care facilities reflect these microclimatic conditions. However, *A. gundlachi* and *A. krugi* are two species that occur in the montane rainforest of Puerto Rico. *Anolis gundlachi* occurs in the cool, deep shade of the forest, and *A. krugi* is typically found in shaded habitats, moving into the sun at higher elevations (Hertz 1992; Rand 1964; Schoener and Schoener 1971; Williams 1972). These habitats offer a very different climate than sunny, open habitats. In our work, after a number of *A. gundlachi* eggs died before hatching when incubated at the standard 27°C (i.e., no eggs from this species survived to hatching), we placed 4 *A. gundlachi* eggs in a 25°C room immediately after being laid. Three of these eggs then produced live hatchlings one month later. In addition, only 17 of 64 *A. krugi* eggs (25%) survived to hatching, but incubating these eggs at a lower temperature may have increased survivorship substantially. For this reason, we recommend matching the cage and egg incubation temperatures and humidity as closely as possible to the natural microclimates of the species for optimal breeding results. Moreover, we note that a comparative study on the thermal biology of anole eggs could prove very interesting.

Additionally, not all species of *Anolis* lizards exhibit the same egg-laying behavior. *Anolis valencienni* is a species that, at least occasionally, lays eggs communally in sites such as tree holes or bromeliads well above the ground (Rand 1967). We have observed that almost all female *A. valencienni* held in captivity do not bury their eggs in the potted plants initially, but instead deposit them on the cage carpet, adhere them to perches, or leave them on the soil surface. To collect viable eggs from this species, we found it necessary to check for eggs every day, particularly in the late evening. However, individuals that have been in captivity for longer times, typically greater than nine months, do begin to deposit their eggs in the pots; after two years in captivity, all eggs are found buried in pots. In the future, we plan to examine whether this species will preferentially lay eggs in an artificial cavity if one is provided in the cage. Whereas the other *Anolis* species we have studied have a mean incubation time of about 29 days, *A. valencienni* also appears to have a considerably longer mean incubation time of about 42 days when incubated under similar conditions.

In conclusion, basic knowledge of the reproductive biology and ecology of a species is necessary to establish successful breeding colonies of lizards. Further consideration of variation among species' ecology may allow for optimization of these methods for species from different habitats. Fortunately for anoles, a substantial body of literature exists on the natural history of many species in this genus, further supporting the utility of these lizards as a model system for a great diversity of research questions.

We have successfully maintained breeding colonies and collected embryos from a diverse collection of *Anolis* species. The above protocols should allow researchers with even a basic animal care facility to breed *Anolis* and collect their embryos for use in standard analyses.

Acknowledgments.—We would like to thank the animal care technicians C. Deggendorf, M. Kwiatkowski, G. McMurray and D. Piatchek for their dedicated attention to these lizards. S. Echternacht, M. Lovern, and J. Stamps gave valuable recommendations for our protocols when beginning our research. J. Kolbe, M. Edgar, and P. Taylor also provided useful feedback on these protocols. Special thanks go to L. Revell and for his careful attention to the animals' well-being and many useful discussions regarding these protocols. We would also like to thank the many undergraduates who have helped optimize these protocols and have ensured the daily care of our lizards, especially S. Barrett, J. Bayers, C. Fresquez, P. Garner, J. Tarsi, and S. Wang. The Washington University Institutional Animal Care and Use Committee formally approved all of the methods described in this paper. Funding for the collection and maintenance of these colonies came from a generous grant from the David and Lucile Packard Foundation and NSF DEB0519777.

LITERATURE CITED

ACKERMAN, R. A., AND D. B. LOTT. 2004. Thermal, hydric, and respiratory climate of nests. *In* D. C. Deming (ed.), Reptilian Incubation: Environment, Evolution and Behavior, pp. 15–43. Nottingham University Press, Nottingham UK.

ANDREWS, R. M. 1985. Oviposition frequency of *Anolis carolinensis*. Copeia 1985:259–262.

———, AND S. RAND. 1974. Reproductive efforts in anoline lizards. Ecology 55:317–327.

———, AND O. J. SEXTON. 1981. Water relations of *Anolis auratus* and *Anolis limifrous*. Ecology 62:556–562.

BARTLETT, R., AND P. BARTLETT. 2001. Anoles: Facts & Advice on Care and Breeding. Barron's Educational Series, Hauppauge, New York.

BURGER, J., K. R. CAMPBELL, AND T. S. CAMPBELL. 2004. Gender and spatial patterns in metal concentrations in brown anoles (*Anolis sagrei*) in southern Florida, USA. Environ. Tox. Chem. 23:712–718.

Clark, D. R., and J. C. Kroll. 1974. Thermal ecology of anoline lizards: temperate vs. tropical strategies. Southwest. Nat. 19:9–19.

Deming, D. C. (ed.). 2004. Reptilian Incubation: Environment, Evolution and Behavior. Nottingham University Press, Nottingham UK.

Emerson, B. C. 2002. Evolution on oceanic islands: molecular phylogenetic approaches to understanding pattern and process. Mol. Ecol. 11:951–966.

Ferner, J. W. 1979. A review of marking techniques for amphibians and reptiles. SSAR Hepetol. Circ. No. 9:41.

Fisher, M., and A. Muth. 1989. A technique for permanently marking lizards. Herpetol. Rev. 20:45–46.

Fox, W. 1963. Special tubules for sperm storage in female lizards. Nature 198:500–501.

Greenberg, N. 1992. The saurian psyche revisited: Lizards in research. *In* D. O. Schaeffer, K. M. Kleinow, and L. Krulish (eds.), The Care and Use of Amphibians, Reptiles, and Fish in Research. Scientists Center for Animal Welfare, Bethesda, Maryland.

———. 2002. Ethological aspects of stress in a model lizard, *Anolis carolinensis*. Integ. Comp. Biol. 42:526–540.

———. 2003. Sociality, stress, and the corpus striatum of the green *Anolis* lizard. Physiol. Behav. 79:429–440

Hamlett, G. W. D. 1952. Notes on the breeding and reproduction in the lizard in the lizard *Anolis carolinensis*. Copeia 1952:183–185.

Hertz, P. E. 1980. Responses to dehydration in *Anolis* lizards sampled along altitudinal transects. Copeia 1980:440–446.

———. 1981. Adaptation to altitude in two West Indian anoles (Reptilia: Iguanidae): field thermal biology and physiological ecology. J. Zool., London 195:25–37.

———. 1992. Temperature regulation in Puerto-Rican *Anolis* lizards—a field-test using null hypotheses. Ecology 73:1405–1417.

Huey, R. B., P. E. Hertz, and B. Sinervo. 2003. Behavioral drive versus behavioral inertia in evolution: A null model approach. Am. Nat. 161:357–366.

———, and T. P. Webster. 1976. Thermal biology of *Anolis* lizards in a complex fauna: the *cristatellus* group on Puerto Rico. Copeia 1976:496–504.

Hunziker, R. 1994. Green Anole: Selection, Care and Breeding. TFH Publications, Neptune City, New Jersey.

Irschick, D. J., and T. Garland. 2001. Integrating function and ecology in studies of adaptation: Investigations of locomotor capacity as a model system. Ann. Rev. Ecol. Syst. 32:367–396.

———, G. Gentry, A. Herrel, and B. VanHooydonck. 2006. Effects of sarcophagid fly infestations on green anole lizards (*Anolis carolinensis*): An analysis across seasons and age/sex classes. J. Herpetol. 40:107–112.

Jenssen, T. A. and S. C. Nunez. 1994. Male and female reproductive cycles of the Jamaican lizard, *Anolis opalinus*. Copeia 1994:767–780.

———, and ———. 1998. Spatial and breeding relationships of the lizard *Anolis carolinensis*: Evidence of intrasexual selection. Behavior 135:981–1003.

Johnson, M. A. 2005. A new method of temporarily marking lizards. Herpetol. Rev. 36:277–279.

Kobayashi, D., W. J. Mautz, and K. A. Nagy. 1983. Evaporative water loss: humidity acclimation in *Anolis carolinensis* lizards. Copeia 1983:701–704.

Lee, J. C., D. Clayton, S. Eisenstein, and I. Perez. 1989. The reproductive cycle of *Anolis sagrei* in southern Florida. Copeia 1989:930–937.

Licht, P. 1973. Influence of temperature and photoperiod on the annual ovarian cycle in the lizard *Anolis carolinenesis*. Copeia 1973:465–472.

———, and G. C. Gorman. 1970. Reproductive and fat cycles in Caribbean *Anolis* lizards. Univ. California Publ. Zool. 95:1–52.

Losos, J. B. 1994. Integrative approaches to evolutionary ecology: *Anolis* lizards as model systems. Ann. Rev. Ecol. Syst. 25:467–493.

Lovern, M. B., M. M. Holmes, and J. Wade. 2004. The green anole (*Anolis carolinensis*): A reptilian model for laboratory studies of reproductive morphology and behavior. Ilar J. 45:54–64.

Michaud. E. J., and A. C. Echternacht. 1995. Geographic variation in the life history of the lizard *Anolis carolinensis* and support of the pelvic constraint model. J. Herpetol. 29:86–97.

Morgan, K. N., and C. T. Tromborg. 2007. Sources of stress in captivity. App. Anim. Behav. Sci. 102:262–304.

Pacala, S., and J. Roughgarden. 1985. Population experiments with the *Anolis* lizards of St. Maarten and St. Eustatius. Ecology 66:129–141.

Packard, G. C., K. Miller, M. J. Packard, and G. F. Birchard. 1999. A field study of environmentally induced variation in body size and condition in hatchling snapping turtles (*Chelydra serpentina*). Can. J. Zool. 77:278–289.

———, M. J. Packard, and G. F. Birchard. 2000. Availability of water affects organ growth in prenatal and neonatal snapping turtles (*Chelydra serpentina*). J. Comp. Phys. B170:69–74.

Pough, F. H. 1991. Recommendations for the Care of Amphibians and Reptiles in Academic Institutions. National Academy Press, New York.

Rand, A. S. 1964. Ecological distribution in anoline lizards of Puerto Rico. Ecology 45:745–752.

———. 1967. Communal egg laying in anoline lizards. Herpetologica 23:227–230.

Ruibal, R. 1961. Thermal relations of five species of tropical lizards. Evol. 15:98–111.

Sambrook J., and D. W. Russell. 2001. Molecular Cloning: A Laboratory Manual, 3rd ed. Cold Spring Harbor Laboratory Press, Cold Spring Harbor, New York.

Schoener, T. W. 1968. The *Anolis* lizards of Bimini: Resource partitioning in a complex fauna. Ecology 49:704–726.

———, and A. Schoener. 1971. Structural habitats of West Indian *Anolis* lizards II. Puerto Rican uplands. Breviora 375:1–39.

Socci, A. M., M. A. Schlaepfer, and T. A. Gavin. 2005. The importance of soil moisture and leaf cover in a female lizard's (*Norops polyepis*) evaluation of potential oviposition sites. Herpetologica 61:233–240.

Stamps, J. A. 1976. Egg retention, rainfall and egg laying in a tropical lizard *Anolis aeneus*. Copeia 1976:759–764.

———.1983. The relationship between ontogenetic habitat shifts, competition and predator avoidance in a juvenile lizard (*Anolis aeneus*). Behav. Ecol. Sociobiol. 12:19–33.

Summers, C. H. 1988. Chronic low humidity-stress in the lizard *Anolis carolinensis*, effects on ovarian and oviductal recrudescence. J. Exp. Zool. 248:192–198.

Wade, J. 2005. Current research on the behavioral neuroendocrinology of reptiles. Horm. Behav. 48:451–460.

Wilson, M. A., and A. C. Echternacht. 1987. Geographic variation in the critical thermal minimum of the green anole, *Anolis carolinensis* (Sauria: Iguanidae), along a latitudinal gradient. Comp. Biochem. Physiol. 87A:757–760.

———, and ———. 1990. A comparison of heat and cold tolerance of two morphs of *Anolis carolinensis* (Iguanidae) from southern Florida. J. Herpetol. 24:330–333.

Williams, E. E. 1972. The origin of faunas. Evolution of lizard congeners in a complex island fauna: A trial analysis. Evol. Biol. 6:47–89.

Herpetological Review, 2008, 39(1), 64–65.

A Notching System for Marking Softshell Turtles

MICHAEL V. PLUMMER
Department of Biology, Box 12251
Harding University, Searcy, Arkansas 72143, USA
e-mail: plummer@harding.edu

Beginning with Cagle (1939), the marginal scutes of the turtle carapace have often been used for various marking systems on hard-shelled turtles (reviewed in Ernst et al. 1974; Ferner 2007; Plummer and Ferner, *in press*). Because the carapacial margin of trionychid turtles lacks discrete scutes, these turtles present difficulties for unambiguously marking a large number of individuals; thus a systematic marking system for softshells is needed. Herein, I describe a system used in two softshell population studies (Plummer 1977a, 1979). Plummer (1977a) made at least 2300 captures of 1500 marked *Apalone mutica* over a 3-yr period. A long-term population study of *A. spinifera* by Plummer and Mills (*in press*) was based on 570 captures of 270 marked turtles from 1994–2003 and this study continues today (716 captures of 322 turtles as of 2007). Although the marking method was briefly mentioned by Plummer (1977a, 1979), details were not provided.

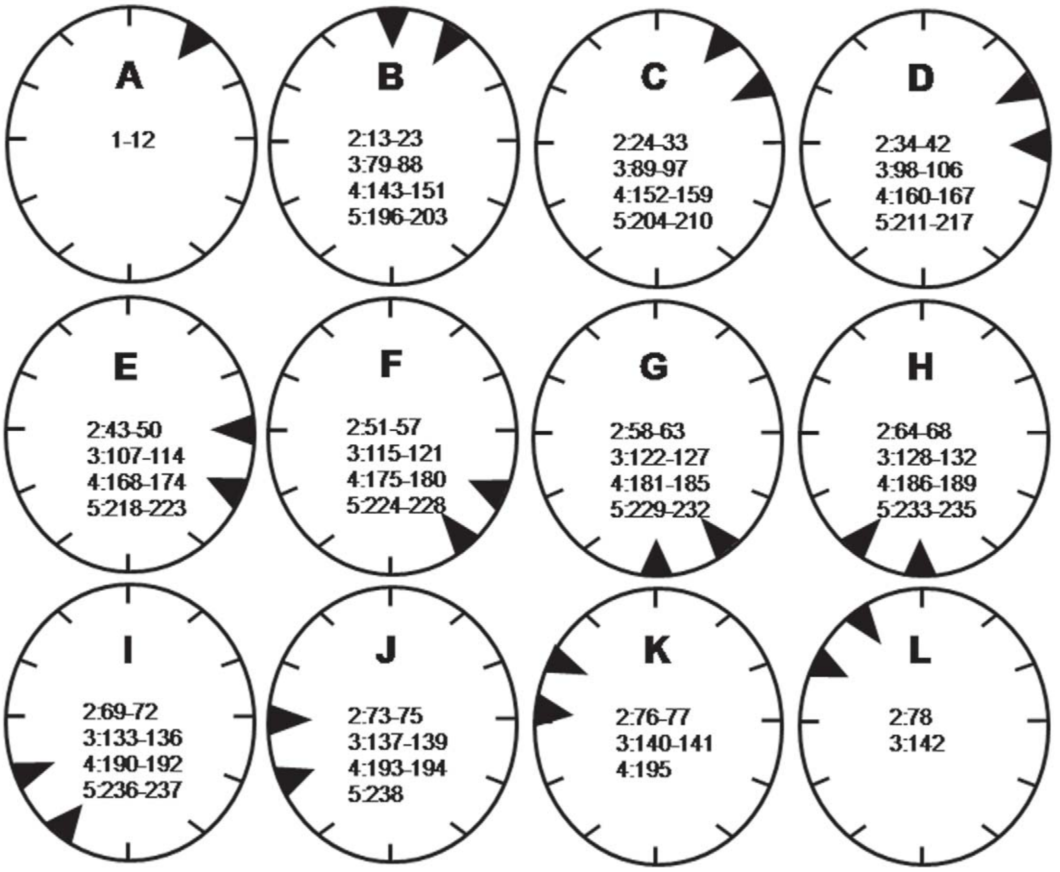


FIG. 1. Schematic of the proposed marking system for softshell turtles. Dark triangles represent notches in the carapacial margin corresponding to the hours of a 12 h clock. The first notch represents the starting position for numbering sequences in B (position 12) through L (position 10) for a two notch sequence. Numbers separated by a colon represent the number of notches to be cut and the range of turtle nos. that can be assigned from the starting position.

Marks are made by cutting triangular-shaped pieces from the edge of the carapace using a pocketknife for adults and juveniles and small scissors for hatchlings. Alternatively, a paper hole punch may be used to apply marks (Doody and Tamplin 1992). A small amount of bleeding may follow marking because the shell is vascularized. The depth of the notched cut varies with turtle size, approximately 15–25 mm for adults and 8–12 mm for juveniles. The notches eventually heal and fill to the original carapacial edge; however, the triangular pale scar tissue remains evident. The numerical marking scheme is based on 12 h clock positions. From a dorsal view, the face of a clock is superimposed on the carapace (Fig. 1). Distinct positions are 12 and 6 (opposite each other on the midline of the carapace), 2 and 10 (just anterior to the bridge), and 3 and 9 (opposite each other but perpendicular to the midline. Positions 4–5 and 7–8 are less precise but positioned between unambiguous positions (3, 6, and 9).

To mark a turtle, one to five notches (notching groups 1–5) are cut in the edge of the carapace. Turtle nos. 1–12 are identified by one notch; nos. 13–78 by two notches; nos. 79–142 by three notches; nos. 143–195 by four notches, and nos. 196–238 by five notches (Fig. 1). Within a notching group (Fig. 1 B–L), numbers are consecutive by progressing from last notch clockwise. For example, turtles no. 13–23 are marked as follows: turtle no. 13 has notches at the nos. 12 and 1 positions, turtle no. 14 has notches at the 12 and 2 positions, etc., to turtle no. 23, which has notches at the 12 and 11 positions (Fig. 1B). For a four notch example, turtle no. 181 has notches at the 5, 6, 7, and 8 positions, turtle no. 182 has notches at the 5, 6, 7, and 9 positions, etc., to turtle no. 185, which has notches at the 5, 6, 7, and 11 positions (Fig. 1G). A final example is turtle no. 238, which has notches at the 8, 9, 10, 11, and 12 positions (Fig. 1J). For numbers greater than 238, the 1-238 series may be repeated by coding with additional marks on the plastron or by toe clipping. For example, a notch in the anterior left quadrant of the plastron codes for turtle nos. 239–476, a notch in the anterior right quadrant codes for nos. 477–714, and notches in both the anterior left and right quadrants code for nos. 715–952. Using the 1–238 notching series and various combinations of just 1–2 notches in the four plastral quadrants would permit marking more than 2500 turtles.

Turtles unambiguously identified after extended periods of time between captures (Plummer and Mills, *in press*) demonstrate the long-term persistence of some marks. For example, 12 adult *A. spinifera* initially marked in 1994–95 were clearly identified 10–13 years later in 2005–07. In addition, five juvenile male *A. spinifera* initially marked at 30–60 mm plastron length (PL) were identified as adults 5–6 years later at 90–100 mm PL and 15 juvenile female *A. spinifera* initially marked at 30–60 mm PL were identified, some as adults, 7–13 years later at 140–215 mm PL. Whether marks on some individuals heal sufficiently to prevent identification is unknown; however, natural processes may occasionally alter marks. For example, the posterior most carapacial edge of adult males, which includes notching positions 5–7, is particularly susceptible to disfigurement resulting from aggressive intraspecific behavior (Doody and Tamplin 1992; Plummer 1977b). Growth from a small juvenile to a large adult may obscure marks such that periodic remarking is needed. Validation studies are needed to quantify the proportion of turtles that possibly "lose" their marks.

Several investigators have used shell marking in softshell mark-recapture studies (Breckenridge 1955; Doody and Tamplin 1992; Fitch and Plummer 1975; Graham and Graham 1997; Plummer 1977a; Plummer and Mills, *in press*); however, none described their marking methods in sufficient detail to be used by others. Thus, a systematic shell marking system designed specifically for the unique problems presented by marking large numbers of softshells for long-term studies is not available. The superiority of the system described herein may be open to question; however, its utility has been clearly demonstrated in population studies of two softshell species (Plummer 1979a; Plummer and Mills, *in press*). Whether the system would be suitable for other trionychid species is unknown. Various tagging techniques, such as PIT-tagging, offer advantages to shell marking and have been used on softshells (Galois et al. 2002), but they also have their own limitations (Gibbons and Andrews 2004).

Acknowledgments.—I thank Henry Fitch for challenging me in 1972 to devise a reliable marking system for softshells and Hunter Valls for reading the manuscript. The Arkansas Game and Fish Commission provided scientific collecting permits. This research was approved by the Harding University Animal Care Committee and was conducted following generally accepted guidelines for field research for reptiles (Anonymous 1987).

LITERATURE CITED

ANONYMOUS. 1987. Guidelines for use of live amphibians and reptiles in field research. American Society of Ichthyologists and Herpetologists, The Herpetologists' League, and Society for the Study of Amphibians and Reptiles. 13 pp.

BRECKENRIDGE, W. J. 1955. Observations on the life history of the soft-shelled turtle *Trionyx ferox*, with especial reference to growth. Copeia 1955:5–9.

CAGLE, F. R. 1939. A system of marking turtles for future identification. Copeia 1939:170–173.

DOODY, J. S., AND J. W. TAMPLIN. 1992. An efficient marking technique for softshelled turtles. Herpetol. Rev. 23:54–56.

ERNST, C. H., R. W. BARBOUR, AND M. F. HERSHEY. 1974. A new coding system for hardshelled turtles. Trans. Kentucky Acad. Sci. 35:27–28.

FERNER, J. W. 2007. A Review of Marking and Individual Recognition Techniques for Amphibians and Reptiles. Society for the Study of Amphibians and Reptiles Herpetol. Circ. No. 35. 72 pp.

FITCH, H. S., AND M. V. PLUMMER. 1975. A preliminary ecological study of the soft-shelled turtle, *Trionyx muticus*, in the Kansas River. Israel J. Zool. 24:1–15.

GALOIS, P., M. LEVEILLE, L. BOUTHILLIER, C. DAIGLE, AND S. PARREN. 2002. Movement patterns, activity, and home range of the eastern spiny softshell turtle (*Apalone spinifera*) in northern Lake Champlain, Quebec. Vermont J. Herpetol. 36:402–411.

GIBBONS, J. W., AND K. M. ANDREWS. 2004. PIT-tagging: simple technology at its best. Bioscience 54:447–454.

GRAHAM, T. E., AND A. A. GRAHAM. 1997. Ecology of the eastern spiny softshell, *Apalone spinifera spinifera*, in the Lamoille River, Vermont. Chelon. Cons. Biol. 2:363–369.

PLUMMER, M. V. 1977a. Activity, habitat, and population structure in the turtle, *Trionyx muticus*. Copeia 1977:431–440.

———. 1977b. Notes on the courtship and mating behavior of the softshell turtle, *Trionyx muticus* (Reptilia, Testudines, Trionychidae). J. Herpetol. 11:90–92.

———. 1979. Collecting and marking. *In* M. Harless and H. Morlock (eds.), Turtles: Perspectives and Research, pp. 45–60. John Wiley & Sons, New York.

———, AND J. W. FERNER. In press. Techniques for marking reptiles. *In* R. W. McDiarmid, M. S. Foster, J. W. Gibbons, C. Guyer, and N. Chernoff (eds.), Reptile Biodiversity: Standard Methods for Inventory and Monitoring, Appendix II. University of California Press, Berkeley.

———, AND N. E. MILLS. In press. Structure of an urban population of softshell turtles *(Apalone spinifera)* before and after stream alteration. *In* R. E. Jung and J. C. Mitchell (eds.), Urban Herpetology. Herpetol. Cons. Vol. 3. Society for the Study of Amphibians and Reptiles. Salt Lake City, Utah.

AMPHIBIAN CHYTRIDIOMYCOSIS GEOGRAPHIC DISTRIBUTION

This section offers a timely outlet for streamlined presentation of research exploring the distribution and prevalence of the amphibian chytrid fungus *Batrachochytrium dendrobatidis* (*Bd*). *Bd* is an emerging infectious disease linked to mass mortality of amphibians worldwide, yet *Bd* detections in amphibians with no symptoms also are known in many areas. To aid in our understanding of the scope of this issue, we encourage submission of studies on *Bd* geographic distribution, including research on individual species or groups of species, wild or captive animals, native or non-native species, live animals or museum specimens, environmental samples, and findings with no *Bd* detections. We ask authors to: 1) restrict the Introduction of their paper to a **maximum** of two paragraphs to highlight the context of their study; 2) briefly include both field and laboratory Methods; 3) present Results in a Table, although a map also might be very useful, and limited text; and 4) have a short discussion of a **maximum** of three paragraphs to touch upon key findings. Please consider including the following information in submissions: coordinates and description of sampling areas (or please note if locations are extremely sensitive to reveal, and provide general area instead); positive and negative results; disposition of voucher specimens; date of specimen collection; name of collaborative laboratory or researcher conducting histological sections or PCR analyses; and names of cooperative land owners or land management agencies. We aim to expedite the review and publication process! Please e-mail submissions directly to Associate Editor, Dr. Dede Olson: dedeolson@fs.fed.us.

Herpetological Review, 2008, 39(1), 66–68.

The Northern Leopard Frog *Rana pipiens* is a Widespread Reservoir Species Harboring *Batrachochytrium dendrobatidis* in North America

DOUGLAS C. WOODHAMS
Department of Biology, James Madison University
MSC 7801, Harrisonburg, Virginia 22807, USA
e-mail: dwoodhams@gmail.com

ALEX D. HYATT
DONNA G. BOYLE
Australian Animal Health Laboratory, CSIRO Livestock Industries
Private Bag 24, Geelong, Victoria 3220, Australia
e-mail: alex.hyatt@csiro.au, donna.boyle@csiro.au

and
LOUISE A. ROLLINS-SMITH
Departments of Microbiology and Immunology and of Pediatrics
Vanderbilt University, A5301 Medical Center North
Nashville, Tennessee 37232, USA
e-mail: louise.rollins-smith@vanderbilt.edu

The Northern Leopard Frog, *Rana pipiens*, is wide-ranging across North America and commonly harvested for research and teaching (Gibbs et al. 1971; Moriarty 1998). *Rana pipiens* were harvested on the order of 100,000 specimens per year in the early 1980s in Quebec (Gilbert et al. 1994) and 100,000 pounds per year in the early 1970s in Minnesota (Moriarty 1998). Carey et al. (1999) suggested that population declines observed in this species in the late 1960s or early 1970s in some areas (Gibbs et al. 1971; Rorabaugh 2005) may have been caused by the emergence of chytridiomycosis, a disease caused by infection with the fungus *Batrachochytrium dendrobatidis* (*Bd*). A histological survey of *Bd* infections in *R. pipiens* collected in Quebec province of Canada between 1960 and 2001 revealed a prevalence of 10–20% (Ouellet et al. 2004). Because of their widespread commercial use and transport, we sampled several populations of this species to test for the potential of adult *R. pipiens* to act as a reservoir for *Bd* that could potentially facilitate the spread of disease.

Rana pipiens from Minnesota and Vermont were obtained from commercial suppliers (Table 1) and sampled by swabbing 10 times on each of ventral surface, thighs, and feet upon receipt at Vanderbilt University or after several months in the laboratory. Frogs arrived in boxes with damp sphagnum moss and were in contact with one another. Frogs were then housed in groups of 4–5 in polypropylene (opaque) plastic tanks with Plexiglas® covers (45.7 × 25.4 × 20.3 cm), sterilized with bleach before use, and tilted to one side to allow access to both water and the dry bottom of the container. In addition, *R. pipiens* were sampled in the field at a site near Plainwell, Michigan (42°19'26.7"N, 85°47'01.2"W). At the field site, individuals of six other species were sampled concurrently. Diagnosis of infection status, including the number of zoospore equivalents per swab, was performed at the Australian Animal Health Laboratory, CSIRO, Geelong, Victoria, Australia. All samples were analyzed in triplicate by Taqman real-time PCR assay according to Boyle et al. (2004). Sampling was different to that described by Hyatt et al. (2007) for groups containing multiple tadpoles or juveniles. Animals were rinsed in 50 ml HPLC-grade water for 15 min and the solution absorbed onto swabs as described by Boyle et al. (2004), perhaps underestimating infection loads.

We found that *R. pipiens* obtained from Minnesota and Vermont by two commercial suppliers were positive for *Bd*, indicating infection, but clinical signs of disease were absent. It is unknown whether the frogs became infected in the wild or after collection by suppliers. In general, infection intensity was low, typically ranging from 0 to 651 zoospore equivalents. A swab from one frog contained 90,599 zoospore equivalents. Although some animals died in captivity, there were no significant die-offs observed after housing frogs in the laboratory for several years, indicating that some *R. pipiens* may have resistance to chytridiomycosis. All 55 individuals and seven species sampled in the field in Michigan in 2004 were negative for *Bd*, indicating infection prevalence at the site between 0 and 6.49% with 95% confidence. The data imply that the Michigan population was probably not infected in the field. After exposure to water housing *R. pipiens* from commercial suppliers, at least one and up to four of six frogs collected from Michigan developed low-level *Bd* infections (Table 1).

These results suggest that *R. pipiens* can function as a reservoir or carrier species for *Bd*. This places *R. pipiens* in a category with other wide-ranging and introduced species used for food, fish bait, research, teaching, and pets (Fisher and Garner 2007). Current regulations do not require amphibian suppliers or breeders to deliver uninfected frogs or to check infection status. However, unless frogs escape or are released into new habitats, or their waste water is not treated, they do not necessarily put susceptible amphibians at risk. Unfortunately, disease emergence caused by introduced species that are *Bd* reservoirs is increasingly detected (Fisher and Garner 2007), for example in Britain (Cunningham et al. 2005) and Uruguay (Mazzoni et al. 2003). We recommend increased monitoring and stricter biosecurity to prevent the spread of chytridiomycosis. The overall health of amphibians obtained from commercial suppliers has improved since the report by Gibbs et al. (1971), and more rigorous distribution procedures will surely benefit research and conservation.

It seems unlikely that all populations of *R. pipiens* carry *Bd*. Therefore, measures should be taken to limit human-mediated exposure to the pathogen. Populations that already carry *Bd* may still be at risk. Different strains of *Bd* may differ in pathogenicity (Berger et al. 2005; Retallick et al. 2007). Hence, exposure of susceptible amphibians to new strains of *Bd* carried by *R. pipiens* and other reservoir species may continue to threaten populations with disease.

Of interest for conservation management, population level variation in susceptibility may exist, and this awaits exposure experiments and immune defense characterization of different populations. Since carrier species are expected to survive *Bd* exposure, susceptible controls may be used in exposure experiments as in Daszak et al. (2004).

This study was not extensive and is comprised of information

TABLE 1. Amphibians sampled for diagnosis of infection with *Batrachochytrium dendrobatidis*. Frogs obtained by commercial suppliers were sampled in the laboratory at Vanderbilt University. Samples that returned a low number of zoospore equivalents in only one well* or two wells** (from a total of three) were defined as "indeterminate."

Date sampled (month/yr)	Location	Number swabbed	Species	Life-history stage	*Bd* diagnosis (no. zoospore equivalents)
3/04	BioCorporation, Alexandria, MN; frogs harvested in Minnesota, swabbed upon receipt	1	*Rana pipiens*	adult	0
		1	*Rana pipiens*	adult	0.2*
		1	*Rana pipiens*	adult	0.3*
		1	*Rana pipiens*	adult	1**
		1	*Rana pipiens*	adult	2**
		1	*Rana pipiens*	adult	6**
		2	*Rana pipiens*	adult	1
		1	*Rana pipiens*	adult	3
		3	*Rana pipiens*	adult	4
		1	*Rana pipiens*	adult	5
		1	*Rana pipiens*	adult	7
		1	*Rana pipiens*	adult	9
		2	*Rana pipiens*	adult	10
		1	*Rana pipiens*	adult	11
		1	*Rana pipiens*	adult	12
		2	*Rana pipiens*	adult	14
		1	*Rana pipiens*	adult	27
9/04	BioCorporation, Alexandria, MN; frogs harvested in Minnesota and kept in laboratory	3	*Rana pipiens*	adult	0
		1	*Rana pipiens*	adult	28
		1	*Rana pipiens*	adult	59
		1	*Rana pipiens*	adult	268
		1	*Rana pipiens*	adult	491
		1	*Rana pipiens*	adult	651
		1	*Rana pipiens*	adult	90599
12/04	BioCorporation, Alexandria, MN; frogs harvested in Minnesota and kept in laboratory	1	*Rana pipiens*	adult	0
		1	*Rana pipiens*	adult	0.46*
		1	*Rana pipiens*	adult	1*
		1	*Rana pipiens*	adult	13
		1	*Rana pipiens*	adult	18
		1	*Rana pipiens*	adult	288
12/04	Connecticut Valley Biological Supply Co., Southhampton, MA; frogs harvested in Vermont, swabbed upon receipt	2	*Rana pipiens*	adult	0
		2	*Rana pipiens*	adult	1
		1	*Rana pipiens*	adult	0.08*
		1	*Rana pipiens*	adult	3
		1	*Rana pipiens*	adult	248
7/04	Plainwell, Michigan	1	*Bufo americanus*	adult	0
		1 group of 5	*Bufo americanus*	juvenile	0
		2 groups of 5	*Hyla* sp.	tadpole	0
		1 group of 5	*Pseudacris crucifer*	juvenile	0
		1 group of 2	*Pseudacris triseriata*	juvenile	0
		1 group of 5	*Rana sylvatica*	juvenile	0
		1	*Rana* sp.	tadpole	0
		1 group of 2	*Rana clamitans*	juvenile	0
		2	*Rana clamitans*	juvenile	0
		3	*Rana clamitans*	adult male	0
		7	*Rana pipiens*	juvenile	0
9/04	Plainwell, Michigan	6	*Rana pipiens*	subadult	0
12/04	Laboratory (collected from Plainwell, Michigan, 9/04)	2	*Rana pipiens*	subadult	0
		1	*Rana pipiens*	subadult	0.13*
		1	*Rana pipiens*	subadult	0.73*
		1	*Rana pipiens*	subadult	1**
		1	*Rana pipiens*	subadult	17

accumulated while undertaking other studies. The data, however, are important because in addition to identifying *R. pipiens* as a carrier of *Bd*, they highlight the infectivity of *Bd* and the requirement for stringent biosecurity.

Acknowledgments.—We thank Jason Woodhams and Lawrence Woodhams for help with field collections, swamp property owners for access to field sites, and Louise Woodhams for hospitality. Scientific Collection Permits were provided by Michigan Department of Natural Resources. Subcontracts to L. A. Rollins-Smith and A. D. Hyatt from Integrated Research Challenges in Environmental Biology (IRCEB) grants IBN-9977063 and DEB-0213851 (James P. Collins, P.I.) and grant IOB-0520847 (L.A. Rollins-Smith, P.I.) from the U.S. National Science Foundation funded this research. D. Woodhams was partially supported by an NHBLI Immunology of Blood and Vascular Systems Training Grant 5T32 HL069765-05 (Jacek Hawiger, P.I.).

Literature Cited

Berger, L., G. Marantelli, L. F. Skerratt, and R. Speare. 2005. Virulence of the amphibian chytrid fungus, *Batrachochytrium dendrobatidis*, varies with the strain. Dis. Aquat. Org. 68:47–50.

Boyle, D. G., D. B. Boyle, V. Olsen, J. A. T. Morgan, and A. D. Hyatt. 2004. Rapid quantitative detection of chytridiomycosis (*Batrachochytrium dendrobatidis*) in amphibian samples using real-time Taqman PCR assay. Dis. Aquat. Org. 60:133–139.

Carey, C., N. Cohen, and L. Rollins-Smith. 1999. Amphibian declines: an immunological perspective. Dev. Comp. Immunol. 23:459–472.

Cunningham, A. A., T. W. J. Garner, V. Aguilar-Sanchez, B. Banks, J. Foster, A. W. Sainsbury, M. Perkins, S. F. Walker, A. D. Hyatt, and M. C. Fisher. 2005. Emergence of amphibian chytridiomycosis in Britain. Vet. Rec. 157:386–387.

Daszak, P., A. Strieby, A. A. Cunningham, J. E. Longcore, C. C. Brown, and D. Porter. 2004. Experimental evidence that the bullfrog (*Rana catesbeiana*) is a potential carrier of chytridiomycosis, an emerging fungal disease of amphibians. Herpetol. J. 14:201–207.

Fisher, M. C., and T. W. J. Garner. 2007. The relationship between the emergence of *Batrachochytrium dendrobatidis*, the international trade in amphibians and introduced amphibian species. Fung. Biol. Rev. 21:2–9.

Gibbs, E. L., G. W. Nace, and M. B. Emmons. 1971. The live frog is almost dead. BioSci. 21:1027–1034.

Gilbert, M., R. LeClair, and R. Fortin. 1994. Reproduction of the northern leopard frog (*Rana pipiens*) in floodplain habitat in the Richelieu River, P. Quebec, Canada. J. Herpetol. 28:465–470.

Hyatt, A.D., D. G. Boyle, V. Olsen, D. B. Boyle, L. Berger, D. Obendorf, A. Dalton, K. Kriger, M. Hero, H. Hines, R. Phillott, R. Campbell, G. Marantelli, F. Gleason, and A. Colling. 2007. Diagnostic assays and sampling protocols for the detection of *Batrachochytrium dendrobatidis*. Dis. Aquat. Org. 73:175–192.

Mazzoni, R., A. C. Cunningham, P. Daszak, A. Apolo, E. Perdomo, and G. Speranza. 2003. Emerging pathogen of wild amphibians in frogs (*Rana catesbeiana*) farmed for international trade. Emerg. Infect. Dis. 9:995–998.

Moriarty, J. J. 1998. Status of amphibians in Minnesota. *In* M. J. Lannoo (ed.), Status and Conservation of Midwestern Amphibians, pp. 166–168. University of Iowa Press, Iowa City, Iowa.

Ouellet, M., I. Mikaelian, B.D. Pauli, J. Rodrigue, and D.M. Green. 2004. Historical evidence of widespread chytrid infection in North American amphibian populations. Conserv. Biol. 19:1431–1440.

Retallick, R. W. R., and V. Miera. 2007. Strain differences in the amphibian chytrid *Batrachochytrium dendrobatidis* and non-permanent, sub-lethal effects of infection. Dis. Aquat. Org. 75:201–207.

Rorabaugh, J. C. 2005. *Rana pipiens* Schreber, 1782. *In* M. J. Lannoo (ed.), Amphibian Declines: The Conservation Status of United States Species, pp. 570–577. University of California Press, Berkeley, California.

Herpetological Review, 2008, 39(1), 68–70.

Batrachochytrium dendrobatidis in Wood Frogs (*Rana sylvatica*) from Three National Wildlife Refuges in Alaska, USA

MARI K. REEVES
U.S. Fish and Wildlife Service, Anchorage Fish and Wildlife Field Office
605 W. 4th Avenue, Room G-61, Anchorage, Alaska 99501, USA
e-mail: Mari_Reeves@fws.gov

Batrachochytrium dendrobatidis (*Bd*) is a fungal pathogen implicated in recent amphibian declines (Pounds et al. 2006). It was first documented in Alaska, USA, in 2002, in a single dead subadult Wood Frog (*Rana sylvatica*) in the Kenai National Wildlife Refuge (Reeves and Green 2006). Since then, there have only been two other studies of *Bd* in Alaska. In one, *Bd* was found in Boreal Toads (*Bufo boreas*) and Red-legged Frogs (*Rana aurora*) in western Canada and southeast Alaska (Adams et al. 2007). In the other, *Bd* was not found in wood frogs in Denali National Park (Chestnut et al. 2008). *Bd* distribution in other parts of Alaska is unknown. In summer of 2006, we sampled adult Wood Frogs from three Alaskan National Wildlife Refuges to screen them for *Bd*.

Methods.—Wood Frogs from the Innoko, Kenai, and Tetlin refuges were sampled (Fig. 1). From these refuges, we tested 48 opportunistically-encountered adult frogs from 29 breeding ponds between 11 May and 21 July 2006 (Table 1). At Kenai, four ponds were road-accessible and six were in remote areas, 1–10 km from any road (Fig. 1). All ponds at Innoko (N = 9) and Tetlin (N = 10) were in remote areas, 35–125 km from any road. All animals were alive when sampled and appeared healthy. Frogs were swabbed

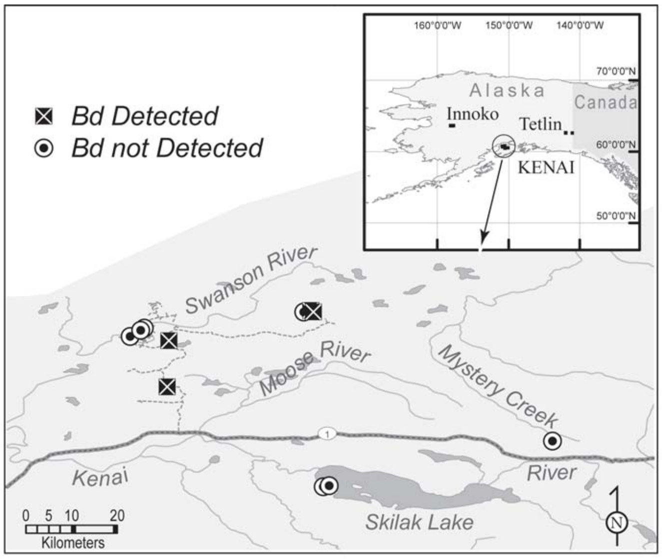


Fig. 1. Wood Frog (*Rana sylvatica*) *Bd* sampling locations in Alaska, USA, and Kenai Refuge sampling ponds and *Bd* detection locations. Dashed lines are gravel roads.

TABLE 1. Surveys for *Batrachochytrium dendrobatidis* (*Bd*) in Wood Frogs (*Rana sylvatica*) in three wildlife refuges in Alaska, USA. Prevalence indicates: number of frogs with *Bd* detected/total number of frogs sampled.

Date	Refuge	Latitude °N	Longitude °W	Prevalence
12 July 2006	Innoko	63.573080	-158.156880	0/1
6 July 2006	Innoko	63.599840	-158.136220	0/1
7 July 2006	Innoko	63.595970	-158.112640	0/1
14 July 2006	Innoko	63.615150	-158.081880	0/1
6 July 2006	Innoko	63.617460	-158.080380	0/1
13 July 2006	Innoko	63.618890	-158.020890	0/1
9 July 2006	Innoko	63.610790	-157.792040	0/1
15 July 2006	Innoko	63.648980	-157.750640	0/1
9 July 2006	Innoko	63.622410	-157.739620	0/1
23 June 2006	Kenai	60.725870	-150.889740	0/2
11 May 2006	Kenai	60.737920	-150.872070	0/8
11 May 2006	Kenai	60.743180	-150.863910	0/6
25 July 2002	Kenai	60.627410	-150.815570	1/1
20-29 June 2006	Kenai	60.714230	-150.815410	4/4
12 May 2006	Kenai	60.776810	-150.547290	0/1
28 June 2006	Kenai	60.780170	-150.543000	2/2
28 June 2006	Kenai	60.776550	-150.539150	1/1
24 May – 14 July 2006	Kenai	60.436260	-150.507710	0/3
14 July 2006	Kenai	60.439000	-150.500000	0/1
22 June 2006	Kenai	60.523690	-150.064090	0/1
18 July 2006	Tetlin	62.603330	-142.051030	0/1
13 July 2006	Tetlin	62.616140	-141.992900	0/1
15 July 2006	Tetlin	62.633420	-141.985200	0/1
12 July 2006	Tetlin	62.608420	-141.980030	0/1
12 July 2006	Tetlin	62.610000	-141.980000	0/1
21 July 2006	Tetlin	62.618210	-141.979860	0/1
17 July 2006	Tetlin	62.607280	-141.972820	0/1
13 July 2006	Tetlin	62.630000	-141.970000	0/1
17 July 2006	Tetlin	62.614990	-141.966540	0/1
17 July 2006	Tetlin	62.619140	-141.963100	0/1

30–35 times on the pelvic patch, inner thighs, and between toes with sterile, foam-tipped swabs (REF 25-1506 1PF: Puritan, Guilford ME). Swab tips were then placed in individual 1.5 ml microfuge tubes in reagent-grade ethanol and stored at room temperature until analysis. Samples were sent in late August 2006 to Pisces Molecular (J. Wood, Boulder, Colorado, USA) for analysis by polymerase chain reaction (Annis et al. 2004). Clean nitrile gloves were worn when handling each frog, and field equipment was decontaminated with 5% bleach solution between ponds. Pond coordinates were recorded with hand-held Garmin III GPS units and referenced to datum, WGS84.

Results.—*Bd* was detected in the Kenai refuge, but not in the Tetlin or Innoko refuges. In Kenai, *Bd* was found in one of four road-accessible ponds and in two of six remote ponds; in the three ponds where *Bd* was found, all sampled frogs tested positive (Table 1). All of the *Bd*-positive ponds were near the Swan Lake recreational canoe route or the gravel road used to access it (Fig. 1). One of the *Bd*-positive ponds was directly adjacent to the road, and the other two ponds were in a wilderness area within 3 km of the road and within 1 km of the canoe trails.

Discussion.—Wood Frogs are the only amphibian common in southcentral and interior Alaska (Wright and Wright 1995). In these regions, *Bd* has been found only on the Kenai refuge, in limited locations (1 pond in 2002 and 3 ponds in 2006). The dead Wood Frog found in 2002 (Reeves and Green 2006) was in a different pond along the same access road as our 2006 *Bd* detections; all positive detections in the Kenai refuge thus have been along this recreational access corridor. Of the four remote ponds that tested negative for *Bd* in 2006, two are in areas where human visitation is rare or nonexistent (access requires a motor boat and then a walk through trail-less forest), one is in a wilderness area near an established hiking trail, and one is near the recreational canoe route and the two ponds that tested positive. Amphibian researchers have visited all the remote ponds repeatedly since 2004, using hygiene protocols described above. The three road-accessible ponds that tested negative for *Bd* are on an operating oil field, where the public is not allowed. The oil field ponds have been visited by amphibian researchers since 2000, and access by other people is limited to oil field workers and other (non-amphibian) researchers. It is unlikely these other users enter the wetlands sampled in this study, although the roads pass within 100 m of each pond. The ponds in the Innoko and Tetlin refuges are all remote, requiring a combination of planes and watercraft to access, and it is unlikely that any people other than amphibian researchers enter the ponds sampled for this study. Although sample size is limited, the results from this study suggest *Bd* is not ubiquitous in southcentral and interior Alaska. More systematic research is needed on the distribution and abundance of *Bd* in Alaskan refuges, especially as it relates to recreational use.

Acknowledgments.—Funding was provided by the U.S. Fish and Wildlife Service, Division of Environmental Quality. The following refuge staff provided logistical support: S. Kovach, J. Morton, J. Hall, C. Caldes, R. West, and B. Johnson. Insightful comments on the manuscript were given by M. Nemeth, D. Olson, and an anonymous reviewer. Field assistants were M. Purdue, S. Jensen, P. Jensen, N. Maxon, T. Hahka, and C. Wall. We thank K. Christiansen with the U.S. Forest Service, Pacific Northwest Research Station, Corvallis, Oregon, for creating Fig. 1. All animals in this study were treated humanely in accordance with institutional Animal Care guidelines, and required federal and state permits were obtained.

LITERATURE CITED

ADAMS, M. J., S. GALVAN, D. REINITZ, R. A. COLE, AND S. PYARE. 2007. Incidence of the fungus *Batrachochytrium dendrobatidis*, in amphibian populations along the Northwest Coast of North America. Herpetol. Rev. 38:430–431.

ANNIS, S. L., F. DASTOOR, H. ZIEL, P. DASZAK, AND J. E. LONGCORE. 2004. A DNA-based assay identifies *Batrachochytrium dendrobatidis* in amphibians. J.Wildl. Dis. 40:420–428.

CHESTNUT, T., J. E. JOHNSON, AND R. S. WAGNER. 2008. Results of amphibian chytrid sampling in Denali National Park, Alaska, USA. Herpetol. Rev. 39: (in press).

POUNDS, J. A., M. R. BUSTAMANTE, L. A. COLOMA, J. A. CONSUEGRA, M. P. L. FOGDEN, P. N. FOSTER, E. LA MARCA, K. L. MASTERS, A. MERINO-VITERI, R. PUSCHENDORF, S. R. RON, G. A. SANCHEZ-AZOFEIFA, C. J. STILL, B. E. YOUNG. 2006. Widespread amphibian extinctions from epidemic disease driven by global warming. Nature. 439:161–167.

REEVES M. K., AND D. E. GREEN. 2006. *Rana sylvatica* wood frog chytridiomycosis. Herpetol. Rev. 37:450.

WRIGHT, A. H., AND A. A. WRIGHT. 1995. Handbook of Frogs and Toads of the United States and Canada. Cornell University Press, Ithaca, New York.

Herpetological Review, 2008, 39(1), 70–73.

Amphibian Chytrid Fungus Infections in *Hyperolius* (Anura: Hyperoliidae) from Eastern Democratic Republic of Congo

ELI GREENBAUM
Department of Biology, Villanova University
800 Lancaster Avenue, Villanova, Pennsylvania 19085, USA
e-mail: eli.greenbaum@villanova.edu

CHIFUNDERA KUSAMBA
Laboratoire d'Erpétologie, Département de Biologie
Centre de Recherche en Sciences Naturelles
Lwiro, République Démocratique du Congo
e-mail: chifundera@yahoo.co.uk

MWENEBATU M. ARISTOTE
Institut Superieur d'Ecologie pour la Conservation de la Nature
Katana Campus, Sud Kivu, République Démocratique du Congo
e-mail: aristote_mwenebatu@yahoo.fr

and
KURT D. REED
Emerging Infectious Disease Laboratory
Marshfield Clinic Research Foundation
1000 North Oak Avenue, Marshfield, Wisconsin 54449, USA
e-mail: reed.kurt@mcrf.mfldclin.edu

Amphibian chytrid fungus (*Batrachochytrium dendrobatidis*) infection has been well-studied in Australia and the New World, where species seem to be especially susceptible to infection in montane, stream habitats (e.g., Carnaval et al. 2006; Hero and Morrison 2004; Lips et al. 2004; McDonald et al. 2005). The destructive fungus also has been associated with frog die-offs and mortality in similar habitats in East and South Africa (e.g., Channing et al. 2006; Hopkins and Channing 2003; Smith et al. 2007), and it is likely that the fungus is killing frogs in other highland regions throughout Africa. If present, the fungus could be potentially catastrophic in Central Africa where the species richness, endemism, and numbers of threatened amphibians are among the highest in continental Africa (Burgess et al. 2004; IUCN et al. 2006). Moreover, the amphibians of Central Africa are poorly surveyed or unknown in many areas (Broadley and Cotterill 2004; Channing and Howell 2006; Laurent 1983; Plumptre et al. 2003; Schmidt and Noble 1919), and thus, infections could wipe out species before they are identified by science. Remarkably, no published study has examined amphibians in Central Africa for the presence of chytrid fungus.

We (EG, CK, and MA) conducted a preliminary survey of the herpetofauna at sites in and near Kahuzi Biega National Park (a UNESCO World Heritage Site in Danger), South Kivu Province, Democratic Republic of Congo between 14 August and 2 September 2007. Habitats ranged from high-elevation bamboo forest to lowland rainforest, and although the timing of our collecting corresponded to the dry season for the eastern Congo highlands (Chapin 1932) we observed some rainfall almost daily. Amphibians were collected by hand, euthanized via cutaneous contact with Orajel®, and preserved in 10% formalin solution; after a 24 h rinse in water, specimens were transferred to 75% ethanol and 1–4 mm toe clippings of 24 selected specimens (Table 1) were prepared for histological examination. Tissues were dehydrated in graded concentrations of ethanol and then xylene, paraffin-embedded, sectioned at 4 microns and stained with hematoxylin and eosin. To avoid delays from cataloging backlogs, field numbers are provided for voucher specimens, but these specimens will be deposited in an American natural history museum collection in the future. Abbreviations are as follows: EBG = Eli Greenbaum field series; SVL = snout–vent length.

Two of 24 specimens (EBG 1087 and EBG 1307) showed evidence of chytridiomycosis. The former specimen is a subadult *Hyperolius kivuensis* (19.7 mm SVL; adult size 22–39 mm according to Schiøtz 1999) with no evidence of lesions. A juvenile *H. kuligae* (EBG 1307; 10.1 mm SVL; adult size 20–31 mm according to Schiøtz 1999) has multiple small, white lesions on the venter of the hind limbs and abdomen. Infections were characterized by thickening of the superficial keratinized layers of the epidermis due to the presence of smooth-walled sporangia of *Batrachochytrium* that ranged in diameter from 10–25 microns. Most sporangia were empty, but several contained five to ten zoospores (Figs. 1A, B). No hyphae were present and there was no inflammatory cell response in the deeper layers of the epidermis and dermis. Twenty-two additional specimens representing 17 additional anuran species were negative for chytrid infection (Table 1).

To the best of our knowledge, the chytrid infections reported herein are the first positive results for any amphibian in Central Africa, where the fungus is present in both lowland (primary rainforest) and highland (secondary montane forest) habitats. Both infected individuals were collected in (EBG 1087) or near (EBG 1307) streams in close proximity to (< 1 km) human habitations and agricultural fields. The subadult and juvenile ages of the infected frogs are consistent with the high rate of infection and mortality reported for postmetamorphic frogs in Africa and Australia (Berger et al. 1999; Smith et al. 2007).

Weldon et al. (2004) hypothesized that *Batrachochytrium*

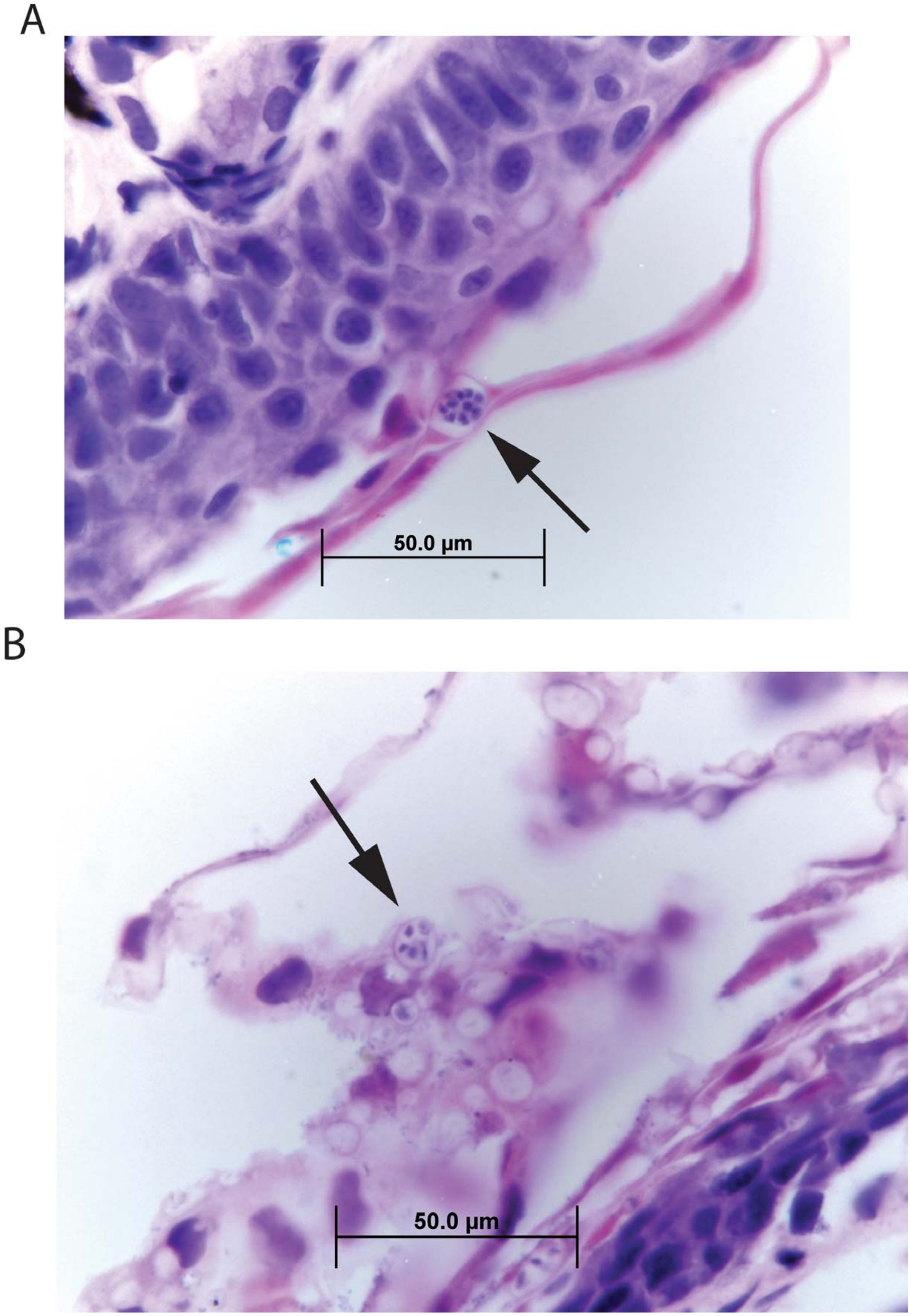


Fig. 1. Chytridiomycosis was detected in two frogs from the eastern Democratic Republic of Congo in Summer, 2007. (A) Sporangia (arrow) containing 10 zoospores from the toe of a subadult *Hyperolius kivuensis* (EBG 1087) from Tshivanga; (B) skin from the toe of a juvenile *Hyperolius kuligae* (EBG 1307) from Irangi. The superficial keratinzed layer of epidermis is markedly thickened from numerous round-to-oval sporangia of *Batrachochytrium dendrobatidis*. Most of the sporangia are empty, but one contains five zoospores (arrow).

TABLE 1. List of amphibian species tested for chytrid infection, including locality information. EBG = Eli Greenbaum field series. All localities are in South Kivu Province, Democratic Republic of Congo. Coordinates are from the WGS 84 datum.

Field Number	Species	Locality	Date of Collection	Habitat	Chytrid Infection
EBG 1316	*Afrixalus laevis*	Forest near Irangi, -1.8873055, 28.4495, 820 m	28 Aug 07	Primary rainforest	Negative
EBG 1336	*Afrixalus osorioi*	Irangi, -1.8743611, 28.4523611, 806 m	29 Aug 07	Secondary rainforest	Negative
EBG 1331	*Afrixalus quadrivittatus*	Irangi, -1.8743611, 28.4523611, 806 m	29 Aug 07	Secondary rainforest	Negative
EBG 1178	*Amietophrynus* sp.	Lwiro, -2.2383611, 28.8051944, 1750 m	18 Aug 07	Agricultural pond in cleared montane forest	Negative
EBG 1255	*Arthroleptis* cf. *adolfifriederici*	Mugaba, -2.2750278, 28.6631111, 2333 m	26 Aug 07	Bamboo forest	Negative
EBG 1111	*Hyperolius castaneus*	Mbayo, -2.2545833, 28.7680556, 2146 m	15 Aug 07	Stream near edge of montane forest	Negative
EBG 1253	*Hyperolius castaneus*	Mugaba, -2.2750278, 28.6631111, 2298 m	25 Aug 07	Montane forest	Negative
EBG 1372	*Hyperolius cinnamomeoventris*	Catena near Irangi, -1.8655, 28.4526944, 805 m	30 Aug 07	Primary rainforest	Negative
EBG 1087	*Hyperolius kivuensis*	Maziba village, near Tshivanga, -2.3128056, 28.7551944, 2200 m	14 Aug 07	Stream near edge of montane forest	Positive
EBG 1160	*Hyperolius kivuensis*	Lwiro, -2.2383611, 28.8051944, 1750 m	18 Aug 07	Agricultural pond in cleared montane forest	Negative
EBG 1307	*Hyperolius kuligae*	Forest near Irangi, -1.8873056, 28.4495, 820 m	28 Aug 07	Vegetation 2 m above a stream in primary rainforest	Positive
EBG 1110	*Hyperolius nasutus*	Mbayo, -2.2545833, 28.7680556, 2146 m	15 Aug 07	Stream near edge of montane forest	Negative
EBG 1226	*Hyperolius nasutus*	Nyakasaz Swamp near Lwiro, -2.2278889, 28.7793333, 1991 m	23 Aug 07	Swamp near secondary montane forest	Negative
EBG 1120	*Hyperolius* sp.	Mbayo, -2.2783611, 28.77175, 2146 m	15 Aug 07	Flooded reeds adjacent to montane forest	Negative
EBG 1344	*Leptopelis christyi*	Irangi, -1.8743611, 28.4523611, 806 m	29 Aug 07	Secondary rainforest	Negative
EBG 1116	*Leptopelis* cf. *kivuensis*	Mbayo, -2.2783611, 28.77175, 2146 m	15 Aug 07	Flooded reeds adjacent to montane forest	Negative
EBG 1282	*Leptopelis* cf. *kivuensis*	Mugaba, -2.2671389, 28.6455, 2267 m	26 Aug 07	Montane forest	Negative
EBG 1127	*Ptychadena* cf. *chrysogaster*	Kayumaga stream near Mbayo, -2.2663056, 28.7838056, 1943 m	17 Aug 07	Agricultural stream in cleared montane forest	Negative
EBG 1213	*Ptychadena* cf. *chrysogaster*	vicinity of Lwiro, -2.2333333, 28.8, 1750 m	23 Aug 07	Agricultural pond in cleared montane forest	Negative
EBG 1292	*Ptychadena mascareniensis*	Irangi, -1.8746667, 28.4523889, 793 m	27 Aug 07	Secondary rainforest edge puddles	Negative
EBG 1142	*Schoutedenella* cf. *schubotzi*	Mbayo, -2.2590278, 28.7683056, 2156 m	17 Aug 07	Road in cleared montane forest	Negative
EBG 1294	*Xenopus pygmaeus*	Irangi, -1.8743611, 28.4523611, 806 m	27 Aug 07	Roadside ditch near secondary rainforest	Negative
EBG 1169	*Xenopus victorianus*	Lwiro, -2.2383611, 28.8051944, 1750 m	18 Aug 07	Agricultural pond in cleared montane forest	Negative
EBG 1105	*Xenopus wittei*	Mbayo, -2.2783611, 28.77175, 2150 m	15 Aug 07	Agricultural pond near secondary montane forest	Negative

dendrobatidis originated in Southern Africa and spread to other regions of the world through commercial frog trade. Based on this premise, Smith et al. (2007:163) suggested that the relative rarity of amphibian die-offs associated with *B. dendrobatidis* in southern Africa reflects regional differences either in the pathogenicity or the host response to chytrid infection. Because we observed large, reproducing populations of *Hyperolius kivuensis* and *H. kuligae* in several localities in and near Kahuzi Biega, it is possible that chytrid infections in these species are not causing significant mortality rates.

Our results are likely an underestimate of the true incidence of chytrid infection in Kahuzi Biega for four reasons: 1) to minimize destructive sampling, we examined toe clippings from only one specimen per species in a given locality; 2) we sampled a small portion of the epidermis, and sampling from more areas of the body could have identified additional, positive infections; 3) we did not want to engage in destructive sampling of several unique specimens from species that are rare (e.g., *Arthroleptis* sp., *Hyperolius ocellatus*, *Leptopelis modestus*) or potentially new; and 4) because of recent warfare and ongoing rebel militia activity (Afoaku 2004; Barnes and Lahm 1997; Edgerton 2002; Hart and Liengola 2005), it was not possible to sample amphibians in many unique habitats in the park (Fischer 1996). Further testing is needed for additional species and localities throughout the Albertine Rift to assess potential threats to scores of amphibians (including several monotypic genera) with limited distributions in the highlands (IUCN et al. 2006; Laurent 1983; Plumptre et al. 2003). The presence of the fungus in lowland habitats of eastern Congo also underscores the need for testing in multiple elevations and habitats to improve understanding of the distribution and niche requirements of the fungus.

Acknowledgments.—We thank Ben Evans for assistance with *Xenopus* frog identification and Patrick Kraus for preparing the histology specimens. We appreciate our field companions Maurice L. Mutwa and Celestin B. Chimanuka, and Baluku Bajope of CRSN for project support. Kevin G. Smith and one anonymous reviewer provided helpful comments on an earlier version of the manuscript. This project was supported by an IUCN/SSC Amphibian Specialist Group Seed Grant to the senior author. Additional thanks to Aaron Bauer and Russell Gardner for project support and research funds from the Department of Biology at Villanova University.

LITERATURE CITED

AFOAKU, O. 2004. Congo's rebels: their origins, motivations, and strategies. *In* J. F. Clark (ed.), The African Stakes of the Congo War, pp. 109–128. Palgrave Macmillan, New York, New York.

BARNES, R. F. W., AND S. A. LAHM. 1997. An ecological perspective on human densities in the central African forests. J. Appl. Ecol. 34:245–260.

BERGER, L., R. SPEARE, AND AND A. D. HYATT. 1999. Chytrid fungi and amphibian declines: overview, implications and future directions. *In* A. Campbell (ed.), Declines and Disappearances of Australian Frogs, pp. 23–33. Environment Australia, Canberra, Australia.

BROADLEY, D. G., AND F. P. D. COTTERILL. 2004. The reptiles of southeast Katanga, an overlooked 'hot spot'. Afr. J. Herpetol. 53:35–61.

BURGESS, N., J. D'AMICO HALES, E. UNDERWOOD, E. DINERSTEIN, D. OLSON, I. ITOUA, J. SCHIPPER, T. RICKETTS, AND K. NEWMAN. 2004. Terrestrial Ecoregions of Africa and Madagascar: A Conservation Assessment. Island Press, Washington, DC. xxiii + 501 pp.

CARNAVAL, A. C. O. Q., R. PUSCHENDORF, O. L. PEIXOTO, V. K. VERDADE, AND M. T. RODRIGUES. 2006. Amphibian chytrid fungus broadly distributed in the Brazilian Atlantic rain forest. EcoHealth 3:41–48.

CHANNING, A., K. S. FINLOW-BATES, S. E. HAARKLAU, AND P. G. HAWKES. 2006. The biology and recent history of the critically endangered kihansi spray toad *Nectophrynoides asperginis* in Tanzania. J. East Afric. Nat. Hist. 95:117–138.

———, AND K. M. HOWELL. 2006. Amphibians of East Africa. Cornell University Press, Ithaca, New York. xi + 418 pp.

CHAPIN, J. P. 1932. The Birds of the Belgian Congo. Part I, Section A. Reprint from Bull. Amer. Mus. Nat. Hist. Vol. LXV. viii + 391 pp.

EDGERTON, R. B. 2002. The Troubled Heart of Africa: A History of the Congo. St. Martin's Press, New York, New York. xiv + 288 pp.

FISCHER, E. 1996. Die vegetation des Parc National de Kahuzi-Biega, Sud Kivu, Zaire. Unpubl. Ph.D. Diss., Franz Steiner Verlag, Stuttgart, Germany. 240 pp.

HART, J., AND I. LIENGOLA. 2005. Post-conflict inventory of Kahuzi Biega National Park. Gorilla Journal 30:3–5.

HERO, J.-M., AND C. MORRISON. 2004. Frog declines in Australia: global implications. Herpetol. J. 14:175–186.

HOPKINS, S., AND A. CHANNING. 2003. Chytrid fungus in Northern and Western Cape frog populations, South Africa. Herpetol. Rev. 34:334–336.

IUCN, CONSERVATION INTERNATIONAL, AND NATURESERVE. 2006. Global Amphibian Assessment. <www.globalamphibians.org>. Accessed on 31 October 2007.

LAURENT, R. F. 1983. About the herpetofauna of Central African montane forest. *In* A. G. J. Rhodin and K. Miyata (eds.), Advances in Herpetology and Evolutionary Biology: Essays in Honor of Ernest E. Williams, pp. 350–358. Museum of Comparative Zoology, Cambridge, Massachusetts.

LIPS, K. R., J. R. MENDELSON, III., A. MUNOZ-ALONSO, L. CANSECO-MARQUEZ, AND D. G. MULCAHY. 2004. Amphibian population declines in montane southern Mexico: resurveys of historical localities. Biol. Cons. 119:555–564.

MCDONALD, K. R., D. MENDEZ, R. MUELLER, A. B. FREEMAN, AND R. SPEARE. 2005. Decline in the prevalence of chytridiomycosis in frog populations in north Queensland, Australia. Pacific Cons. Biol. 11:114–120.

PLUMPTRE, A. J., M. BEHANGANA, T. R. B. DAVENPORT, C. KAHINDO, R. KITYO, E. NDOMBA, D. NKUUTU, I. OWIUNJI, P. SSEGAWA, AND G. EILU. 2003. The Biodiversity of the Albertine Rift. Albertine Rift Technical Reports No 3. Wildlife Conservation Society. 107 pp.

SCHIØTZ, A. 1999. Treefrogs of Africa. Edition Chimaira. Frankfurt am Main, Germany. 351 pp.

SCHMIDT, K. P., AND G. K. NOBLE. 1919–1923. Contributions to the Herpetology of the Belgian Congo. Facsimile Reprints in Herpetology, Society for the Study of Amphibians and Reptiles, St. Louis, Missouri. 780 pp.

SMITH, K. G., C. WELDON, W. CONRADIE, AND L. H. DU PREEZ. 2007. Relationships among size, development, and *Batrachochytrium dendrobatidis* infection in African tadpoles. Dis. Aquat. Org. 74:159–164.

WELDON, C., L. H. DU PREEZ, A. D. HYATT, R. MULLER, AND R. SPEARE. 2004. Origin of the amphibian chytrid fungus. Emerg. Infect. Dis. 10:2100–2105.

NATURAL HISTORY NOTES

The Natural History Notes section is analogous to Geographic Distribution. Preferred notes should 1) focus on observations in the field, with little human intrusion; 2) represent more than the isolated documentation of developmental aberrations; and 3) possess a natural history perspective. Individual notes should, with few exceptions, concern only one species, and authors are requested to choose a keyword or short phrase which best describes the nature of their note (e.g., Reproduction, Morphology, Habitat, etc.). Use of figures to illustrate any data is encouraged, but should replace words rather than embellish them. The section's intent is to convey information rather than demonstrate prose. Articles submitted to this section will be reviewed and edited prior to acceptance.

Electronic submission of manuscripts is requested (as Microsoft Word or Rich Text format [rtf] files, as e-mail attachments). Figures can be submitted electronically as JPG files, although higher resolution TIFF or PDF files will be requested for publication. Please DO NOT send graphic files as imbedded figures within a text file. Additional information concerning preparation and submission of graphics files is available on the SSAR web site at: http://www.ssarherps.org/HRinfo.html. Manuscripts should be sent to the appropriate section editor: **Marc P. Hayes** (crocodilians, lizards, and *Sphenodon*; mhayesrana@aol.com); **Charles W. Painter** (amphibians; charles.painter@state.nm.us); **Andrew T. Holycross** (snakes; holycross@asu.edu); and **James Harding** (turtles; hardingj@pilot.msu.edu).

Standard format for this section is as follows: SCIENTIFIC NAME, COMMON NAME (for the United States and Canada as it appears in Crother [ed.] 2008. *Scientific and Standard English Names of Amphibians and Reptiles of North America North of Mexico*. SSAR Herpetol. Circ. 37:1–84, available from SSAR Publications Secretary, ssar@herplit.com; for Mexico as it appears in Liner 1994, *Scientific and Common Names for the Amphibians and Reptiles of Mexico in English and Spanish*. Herpetol. Circ. 23:1–113), KEYWORD. DATA on the animal. Place of deposition or intended deposition of specimen(s), and catalog number(s). Then skip a line and close with SUBMITTED BY (give name and address in full—spell out state names—no abbreviations). (NCN) should be used for common name where none is recognized. References may be briefly cited in text (refer to this issue for citation format).

Recommended citation for notes appearing in this section is: Lemos-Espinal, J., and R. E. Ballinger. 1994. *Rhyacosiredon leorae*. Size. Herpetol. Rev. 25:22.

CAUDATA – SALAMANDERS

AMBYSTOMA CALIFORNIENSE (California Tiger Salamander). **PREDATION**. *Ambystoma californiense* is an endemic species listed as threatened by the Department of the Interior (U.S. Fish Wild. Serv. 2004. Fed. Reg. 69:47212–47248) and as a species of special concern by the State of California (Jennings 2004. California Fish Game 90:161–213). Although many factors are stated for the decline of this salamander, wild pigs (*Sus scrofa*)—a game species first introduced into California in the 1920s—are suspected to eat juvenile and adult salamanders and also negatively impact salamander habitats (U.S. Fish Wild. Serv. 2003. Fed. Reg. 68:28648–28670). Here we report on the first documented incident of wild pigs negatively effecting *A. californiense* eggs.

On Blue Oak Ranch in Santa Clara County, California (USA), an early storm brought ca. 7.6 cm of rain between 7 and 9 Nov 2003, saturating the soil. The next storm delivered just over 2.5 cm of rain overnight on 14–15 Nov, causing Junction Pond (37.375556°N, 121.724444°W; elev. 599 m) to fill to a maximum depth of 33 cm in a basin ca. 4 m in diameter. On 20 Nov, we discovered 60 *A. californiense* eggs attached to the substrate of the pond, primarily on Dove Weed (*Eremocarpus setigerus*). No precipitation events occurred over the next 5 days. Humidity was quite low because of offshore winds, with high temperatures reaching almost 29°C and nighttime lows dipping to 2°C. As a result, the water level in Junction Pond dropped to ca. 15 cm, with the diameter of the pond basin shrinking to ca. 2 m. While checking on the *A. californiense* eggs on the morning of 25 Nov, we discovered that wild pigs had used the pond basin as a wallow, crushing most of the eggs and detaching the remainder from their substrates. Those that were not crushed, eaten, or buried in the mud were frozen in the thin ice that covered the surface of the small area of remaining water. Pigs continued to wallow in Junction Pond until rains filled it completely in mid-December.

Submitted by **JEFFERY T WILCOX**, Wildlands Consultants, 1474 Rose Street, Berkeley, California 94702, USA (e-mail: jtwilcox@comcast.net); **BRIAN D. GIBESON**, 36 San Marin Drive, Novato, California 94945, USA (e-mail: bdgibeson@comcast.net); and **MARK R. JENNINGS**, Rana Resources, PO Box 2185, Davis, California 95617 USA, and Research Associate, Department of Herpetology, California Academy of Sciences, 875 Howard Street, San Francisco, California 94103, USA (e-mail: RanaResources@aol.com).

ANURA – FROGS

ARGENTEOHYLA SIEMERSI PEDERSENI (Rana Tractor or Rana de Pedersen). **REPRODUCTION**. *Argenteohyla siemersi* is an uncommon Neotropical hylid frog listed as Endangered by IUCN (2006. Conservation International and NatureServe. Global Amphibians Assessment. http://www.globalamphibians.org. Accessed 01 Mar 2007). The main threat is habitat destruction caused by fires to expand/maintain pastures and pine forestry plantations. *Argenteohyla siemersi pederseni* occurs in northwest Corrientes Province (Argentina), inhabiting the leaf axils of terrestrial *Aechmae* bromeliads in gallery forests along main river systems. Its population appears to be stable (IUCN 2006, *op. cit*).

Reproduction in *A. siemersi pederseni* takes place in temporary pools close to bromeliads. The larva was described by Céspedez (2000. Bol. Asoc. Herpetol. Esp. 11:75–80), who counted 1020 ova (1–1.5 mm diam) in a gravid female. Barrio (1969. Physis 26[71]:225–228) reported diurnal choruses of *A s. siemersi*, from noon to late afternoon.

Here we provide data on clutch size and characteristics of the reproductive behavior in *Argenteohyla siemersi pederseni*. On 28 Sept 2006, at 2330 h after heavy rains (air temp 14.5°C; water temp 17.5°C; humidity 74%), we observed several calling males and two pairs in axillary amplexus in a semi-permanent pond in the Reserva Provincial Iberá (28.68°S, 57.43°W, WGS 84; elev. 65 m), near Colonia Carlos Pellegrini, Corrientes, Argentina. We did not observe bromeliads near the pond. The males had paired vocal sacs and called from the water, floating in open spaces, or near of the edge of the pond among aquatic vegetation (Fig. 1). The vocalization occurred from 1800 h to past midnight in choruses of 3–4 males.

The next day a clutch with ca. 2464 eggs was found in the same pond. The eggs were dark at the animal pole and creamy yellow at the vegetal pole. Mean diameter was 1.73 mm (SD = 0.86, range 1.5–1.9 mm, N = 109). They were laid in gelatinous ribbons floating at the surface of the water or among aquatic vegetation (Fig. 2). Other species reproducing in the same pond were *Scinax squalirostris*, *Chaunus fernandezae*, *Pseudopaludicola falcipes*, and *Elachistocleis bicolor*.

Fig. 1. Calling male *Argenteohyla siemersi pederseni*. Photograph by Víctor Zaracho.

Fig. 2. Egg clutch of *Argenteohyla siemersi pederseni*. Photograph by Víctor Zaracho.

This is the first description of an egg clutch of the genus *Argenteohyla*.

Submitted by **MARIA CECILIA DIMINICH**, División Herpetología, Museo Argentino de Ciencias Naturales "Bernardino Rivadavia," Av. Ángel Gallardo 490 (1405), Ciudad Autónoma de Buenos Aires, Argentina (e-mail: cecitamandua@yahoo.com.ar); and **VICTOR HUGO ZARACHO**, Laboratorio de Herpetología, Facultad de Ciencias Exactas y Naturales y Agrimensura, Universidad Nacional del Nordeste, Av. Libertad 5470, (3400) Corrientes, Argentina (e-mail: victorzaracho@yahoo.com.ar).

BUFO AMERICANUS (American Toad). **REPRODUCTION**. Herein I report an instance of prolonged amplectant behavior with an inanimate object in the bufonid *Bufo americanus*. On 22 April 2007 at ca. 2200 h, I visited South Run District Park (Fairfax Co., Virginia, USA) and observed a male *B. americanus* in amplexus with a tennis ball (Fig. 1). The individual manipulated the tennis ball for ca. 2 min until he finally released his misguided embrace. Although a breeding chorus was well established, the sex ratios appeared to be skewed with males outnumbering females 30:1 in this particular vernal pool. Given the importance of release and advertisement calls for mate recognition in *B. americanus* (Leary 2001. Can. J. Zool. 79[9]:1577–1585), it is not surprising that this instance of amplectant behavior was prolonged. While some bufonid studies have incorporated mating interactions with inanimate objects (Hinsche 1926. Zeitschr. Vergl. Physiol. 4:564–606) few have given an example of this behavior in a natural setting. Despite this, it is well known among field workers that breeding male *B. americanus* will readily grasp objects placed between their front limbs (e.g., worker's fingers) (Tupper, pers. comm.). As suggested by Höbel (2005. Herpetol. Rev. 36:55–56; 2005. Herpetol. Rev. 36:439–440), it is likely that male anurans are more prone to mating mistakes when, as in this scenario, there is high competition for conspecific females.

Fig. 1. Male *Bufo americanus* in amplexus with a tennis ball, found in northern Virginia, USA.

Submitted by **JEFFREY W. STREICHER**, Department of Molecular and Microbiology, George Mason University, 4400 University Drive, Fairfax, Virginia 22030, USA; e-mail: jstreic1@gmu.edu.

BUFO PUNCTATUS (Red-spotted Toad). **PREDATION**. Adult *Bufo punctatus* are predated by some vertebrates, mainly aquatic snakes but also, turtles (Stone et al. 2005. Herpetol. Rev. 36:312), and birds and mammals (Stebbins 2003. A Field Guide to Western Reptiles and Amphibians. Houghton Mifflin Co., Boston, Massachusetts, 533 pp.); tadpoles are predated by gartersnakes and aquatic beetle larvae (Coleoptera) (Livo and Kondratieff 2000. Herpetol. Rev. 31:168–169). In Baja California Sur (BCS), adult *B. punctatus* are part of the diet of gartersnakes (Blázquez 1996. Herpetol. Rev. 27:83–84; Rodríguez-Robles and Galina-Tessaro 2006. Herpetol. Rev. 37:335. Herein, we report predation of an adult *B. punctatus* by the giant aquatic bug, *Lethocerus angustipes* (Heteroptera: Belostomatidae).

At 2138 h on 30 Aug 2006, we found an adult male (60.6 mm SVL, 17 g) *B. punctatus* held by the left hind limb by an adult female *L. angustipes* (60 mm TL). The pair was floating in a small temporary pond near Rancho El Álamo, northern BCS, México (27.108083°N, 112.927306°W), ambient air temperature was 27.1°C and water temperature was 29.4°C. The toad made erratic movements with its forelimbs, but the posterior part of its body seemed paralyzed by the toxins of the *L. angustipes* (Smith 1997. Cambridge University Press). After an hour of observation the toad stopped moving and died.

This observation confirms that *L. angustipes* are voracious predators in freshwater systems and, in addition to aquatic invertebrates and tadpoles (Kehr and Schnack 1991. Alytes 9[3]:61–69), can predate adult toads.

Bufo punctatus and *L. angustipes* were deposited in herpetological and entomological collection respectively, of the Centro de Investigaciones Biológicas del Noroeste (CIBNOR), La Paz, Baja California Sur, México. We thank to A. Cota for assistance with fieldwork and C. Palacios for assistance identifying *L. angustipes*.

Submitted by **VICTOR H. LUJA** (e-mail: lujastro@yahoo.com), **M. CARMEN BLÁZQUEZ**, and **RICARDO RODRÍGUEZ-ESTRELLA**, Centro de Investigaciones Biológicas del Noroeste (CIBNOR), Mar Bermejo #195 Colonia Playa Palo de Santa Rita, La Paz, Baja California Sur, 23090, México.

CRAUGASTOR TALAMANCAE (NCN). **REPRODUCTION AND NEST ATTENDANCE.** The genus *Craugastor* contains 116 species of poorly known tropical direct-developing frogs. While most of the species are characterized by small clutch size, large egg size, and attendance of terrestrial nests by one of the parents, descriptions of reproductive behavior are lacking for the majority of species (Townsend 1996. *In* Powell and Henderson [eds.], Contributions to West Indian Herpetology, a Tribute to Albert Schwartz, pp. 229–239. SSAR Conrib. Herpetol. Vol. 12. Ithaca, New York). Herein, we provide what we believe to be the first report of reproduction in the Central American frog *Craugastor talamancae*, including descriptions of the clutch, the nesting site, and attendance of the nest by the female parent.

On 31 Jan 2007 at ca. 1300 h, we encountered an adult *C. talamancae* with a clutch of 36 eggs in an old-growth lowland tropical wet forest at La Selva Biological Station, Heredia Province, Costa Rica (10.41°N, 84.00°W, ca. 60 m elev.). The nest was located on the ground, under leaf litter, and consisted of a round depression in the soil ca. 50 mm diameter and ca. 25 mm deep. The egg mass was ca. 40 × 30 mm, and ca. 15 mm high. The eggs were ca. 6 mm in diameter. The attending frog was first noted leaping from the vicinity the clutch. We determined the attending frog to be a female by comparison of the relative size of the tympanum to the eye (Savage 2002. The Amphibians and Reptiles of Costa Rica. Univ. Chicago Press. Chicago, Illinois. 934 pp.). The female measured 43.5 mm SUL and 7.75 g. Embryos were stages 5–7 (Townsend and Stewart 1985. Copeia 1985:423–436) because the yolk was cream white, eyes were prominent and darkly pigmented, pupils were clear, and embryos were pink with nascent hind limbs and a short tail, but no evident forelimbs. We collected five eggs from the clutch and kept them in a small container containing moist paper towels at ambient temperature.

FIG. 1. A female *Craugastor talamancae* perched upon a clutch of eggs in leaf litter at La Selva Biological Station, Costa Rica.

We checked the status of the nest in the field on 08 Feb 2007 at ca. 1300 h. At this time, the nest was intact, and the female was perched directly on top of the clutch of eggs. When we checked the nest on 14 Feb 2007 at ca. 1300 h there were no eggs in the nest cavity and the female was not present nearby. However, one egg was still present in the vicinity of the nest, ca 15 cm from the nest cavity, and appeared intact. On 20 Feb 2007, all five of the artificially incubated eggs hatched; the froglets measured 8.8, 8.8, 9.0, 9.0, and 9.2 mm SUL.

To our knowledge this is the first report of reproduction and nest attendance in *Craugastor talamancae*. Nest attendance is common in *Craugastor*, although some species lack this or other parental behavior. While we observed no active parental behavior (such as hydrating eggs or defending the nest from predators), we believe that the occurrence of the female perched upon the clutch containing eggs at such a late developmental stage, on two occasions, to be strong evidence of active nest attendance. Further, while we are unable to determine the development time for these eggs, we presume that hatching must occur significantly longer than 21 days after oviposition because we found these eggs at an intermediate developmental stage. Finally, because the eggs in the field disappeared before the eggs in the lab hatched, we presume that these eggs failed to hatch, likely due to nest predation.

Submitted by **STEVEN M. WHITFIELD**, Florida International University, Department of Biological Sciences, U.P. Campus, OE 167, Miami, Florida 33199, USA (e-mail: steven.whitfield@fiu.edu); **SASHA GREENSPAN**, 685 Greystone Park, Atlanta, Georgia 30324, USA (e-mail: sasha.greenspan@gmail.com); and **SHANNAN MILLER,** U.S. Forest Service, Black Hills National Forest, 330 Mt. Rushmore Rd, Custer, South Dakota 57730, USA (e-mail: shannanmiller78@hotmail.com).

FEJERVARYA LIMNOCHARIS (Indian Cricket Frog). **MORPHOLOGY.** In agriculture fields, amphibian populations are affected by chemical contaminants and managerial practices. Earlier reports have shown that exposure to these contaminants can result in expression of behavioral and morphological abnormalities and contribute to the decline of amphibian populations in agro ecosystems (Daniels 2003. Curr. Sci. 85:1415–1422). Similar incidences have been reported from various habitats of central Western Ghats (Gurushankara et al. 2007. Appl. Herpetol. 4:39–45). Ouellet (2000. *In* Sparling [ed.], Ecotoxicology of Amphibians and Reptiles, pp. 617–661) reported that abnormalities of amphibians in tropical habitats ranged between 0 and 2%. In the following report, we present information on abnormalities of adult *Fejervarya limnocharis* recorded in paddy fields of central Western Ghats. In these fields, regular application of pesticides (organochlorine and organophosphate, range 300–700 mL ha^{-1} per crop) and synthetic manures (nitrophosphate and potash, range 200–300 kg ha^{-1} per crop) is being practiced.

Fejervarya limnocharis is a common inhabitant of all agro ecosystems in central Western Ghats, where it breeds in the shallow water of paddy fields. A survey of *F. limnocharis* was made at ten different plots, each 2.5 ha, in paddy fields located (13.53°–1373°N and 75.63°–75.66°E), near the Bhadra River Dam in Karnataka State of India. Surveys were conducted during January–March 2007 after harvest. During these surveys 259 adult frogs (mean SVL 21.39 mm, SD 4.951 mm) were collected and visually examined for abnormalities following the field keys of Meteyer (2000. Biological Science Report USGS/BRD/BSR-2000-0005). Of the 259 individuals examined, 28 (10.8%) were abnormal. Abnormalities recorded were tumors with disfigured hind limb bones (39.29%), short-toe/ brachydactyly (35.71%), missing digit/Ectrodactyly (21%), and incomplete limb/Ectromelia (3.6%).

Becon et al. (2000. Appl. Herpetol. 3:39–65) observed 5% abnormality of frogs living in contaminated agro ecosystems of Asia, some parts of Europe, and America. Compared to these observations, the occurrence of 10.8% abnormality in our area is very high. Given that pesticides and chemical manures are added to every crop, the likelihood that contaminants cause abnormalities in *F. limnocharis* is high.

Submitted by **A. M. PATEL**, **PRAVEEN A. KULKARNI**, **K. G. GIRISH**, **H. P. GURUSHANKARA,** and **S.V. KRISHNAMURTHY**, Department of Environmental Science, Kuvempu University, Jnana Sahyadri, Shankaraghatta 577 451, Karnataka, India (e-mail: svkrishnamurthy@yahoo.co.in).

HYLARANA RANICEPS (White-lipped Frog). **PREDATION.** We relate herein an observation of the predation of a *Hylarana raniceps* metamorph by a spider. *H. raniceps* is a common species in the lowlands of Bornean dipterocarp forests. In March 2007, we observed a "fishing spider" of the species *Thalassius* cf. *albocinctus* (Doleschall, 1859) at a alluvial pond off Headhunters' Trail, near Camp 5 (04.139056°N, 114.899944°E), Gunung Mulu National Park, Sarawak, East Malaysia (Borneo), with a metamorph of *H. raniceps* as prey. The spider was sitting on a vertical trunk in the pond, about 20 cm above the pond surface and was grasping the right thigh of the metamorph (Fig. 1). The spider was disturbed by photography and ran down the trunk with the prey, entered into the water and actively dove to the bottom at about 1 m below surface. Shortly afterwards, it surfaced again and released the dead metamorph. *Thalassius albocinctus* is well known for fishing small sized fishes from the surface layer of standing or slow running waters.

FIG. 1. *Hylarana raniceps* metamorph captured by a "fishing spider" (Pisauridae: *Thalassius* cf. *albocinctus*), Gunung Mulu National Park, Malaysia.

At Gunung Mulu National Park, we frequently saw *T.* cf. *albocinctus* hunting on the surface of ponds with an abundance of breeding frogs and therefore we assume it to be a common amphibian predator. Toledo (2005 Herpetol. Rev. 36:395–400) reported that risk of invertebrate predation on amphibians is proportionately high during the breeding season and in recently metamorphosed frogs, compared to other life history stages, and we speculate that predation by fishing spiders might have an important impact on larval and metamorph survival.

We thank Hubert Höfer, State Museum of Natural History, Karlsruhe, for identification of the spider via photo and for advice on the manuscript; Volkswagen Stiftung for financial support; the Sarawak Forest Department for research permit (Number 38/2006, dated 3.8.2006.) and the staff of Gunung Mulu National Park, for support with logistics.

Submitted by **TORBEN RIEHL, ALEXANDER HAAS**, Biozentrum Grindel und Zoologisches Museum, Universität Hamburg, Martin-Luther-King-Platz, 3, 20146 Hamburg, Germany (e-mail: alexander.haas@uni-hamburg.de); and **INDRANEIL DAS**, Institute of Biodiversity and Environmental Conservation,

Universiti Malaysia Sarawak, 94300, Kota Samarahan, Sarawak, Malaysia.

HYLA CHRYSOCELIS (Cope's Gray Treefrog). **OCULAR PATHOLOGY**. Ophthalmological disorders have been recorded in most vertebrates. Often, the animals in which these lesions have been described have been in captivity for varying lengths of time. However, it is unusual for such lesions to be discovered in wild populations. This disparity in discovery (not necessarily *incidence*) is probably because the condition may predispose the subject to predation or make it unable to compete successfully for resources. Captive animals are subject to observation and, thus, increased detection of ocular abnormalities. Intraocular inflammation, including panophthalmitis and uveitis have been reported in amphibians and reptiles (Zwart 1985. *In* R. Ippen et al. [eds.], Sinnesorgane: Erkrankungen der augen; Haut und anhangs-organe, pp. 250–269. Akadamie Verlag, Berlin (DDR); Millichamp and Jacobson 1986. *In* R. N. Kirk [ed.], Current Veterinary Therapy IX. Small Animal Practice, Ophthalmic Diseases of Reptiles, pp. 621–623, W. B. Saunders Company, Philadelphia, Pennsylvania; Frye 1991. *In* F. L. Frye [ed.], Husbandry, Medicine and Surgery in Captive Reptiles, Ophthalmic Conditions, 2nd Edition, Vol. 2, pp. 326–244. Krieger Publ. Co., Malabar, Florida; Lawton 1993. *In* S. M. Peterson-Jones and S. M. Crispin [eds.], Manual of Small Animal Ophthalmology, Ophthalmology of Exotic Species, British Small Animal Veterinary Association, Cheltenham, United Kingdom; Frye and Williams 1995. Self-Assessment Colour Review of Reptiles and Amphibians. Manson Publ., Ltd., London, United Kingdom, 192 pp.). The alteration of the crystalline lens, in the form of subcapsular lenticular disorganization, is well known in human ophthalmology (Hogan and Zimmerman 1962. Ophthalmic Pathology, 2nd Edition. W.B. Saunders Company, Philadelphia, Pennsylvania, pp. 344–468) but less often encountered (or recognized) in herpetological medicine.

An adult male *Hyla chrysoscelis* (50 mm SVL) was collected alive on 16 September 2000 by CTM in the South Texarkana/Liberty-Eylau area of Bowie County, Texas, USA (33.3716°N, 94.0733°W, elev. 92.1 m). The left eye was grossly enlarged and discolored. After obtaining essential measurements, the frog was photographed (Fig. 1), and euthanized by immersion in a satu-

FIG. 1. Pre-euthanasia photograph of *Hyla chrysoscelis* with panopthalmitis; note the massively swollen and enlarged left eye.

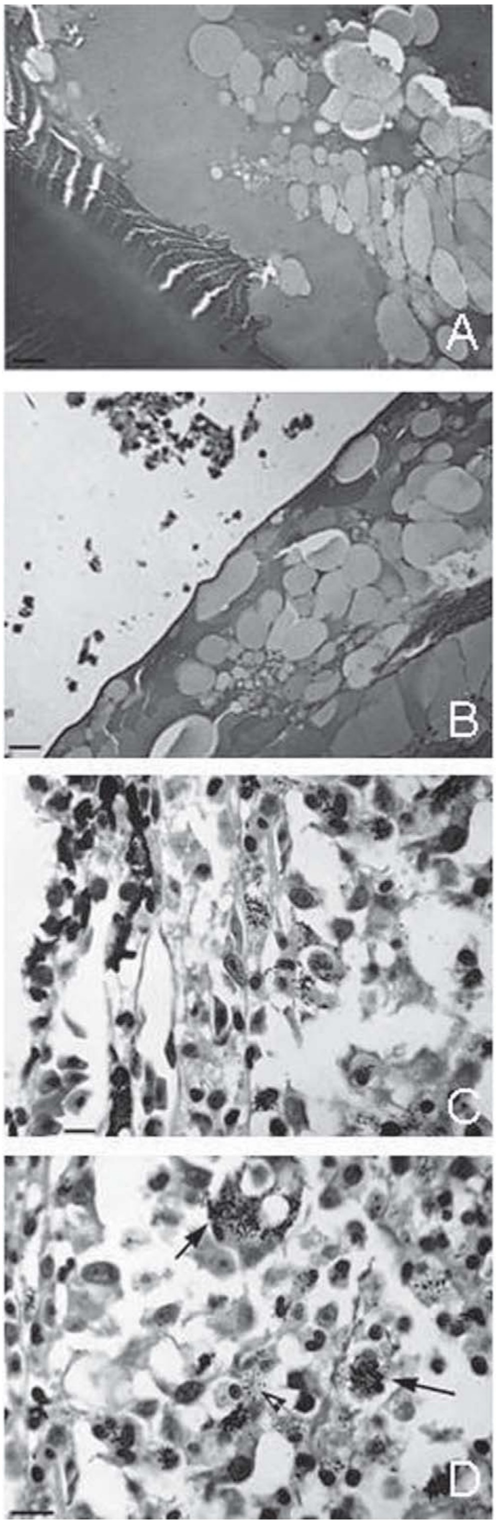


FIG. 2. A. Low power photomicrograph of the crystalline lens and intraocular fibrinous exudate adhering to a portion of the lens capsule. H & E. Bar = 20 µm. B. Photomicrograph of the affected lens. Note the degeneration and disruption of the normally lamellated concentric ring pattern and the pale-staining altered foamy areas. H & E. Bar = 20 µm. C. Photomicrograph of the intraocular exudate. Brown & Brenn. Bar = 30 µm. D. Another view of intraocular exudate; note large macrophages with engulfed bacteria and melanin pigment (solid black arrows) and individual bacteria (open black and white arrow). Brown & Brenn. Bar = 30 µm.

rated chlorobutanol (Chloretone®, Ward's Natural Science, Rochester, New York) solution (Etheridge 1958. Methods for Preserving Amphibians and Reptiles for Scientific Study. Mus. Zool., Univ. Michigan, Ann Arbor, Michigan, 18 pp.). The eye was enucleated, preserved in 10% neutral buffered formalin, and submitted for routine histopathological processing. A single hematoxylin and eosin stained section was examined by light microscopy and photographed by digital photomicroscopy. Supplemental sections were cut on a rotary microtome at 7 µm and stained with Brown and Brenn's modification of Gram's stain (Leaver et al. 1977. J. Clin. Pathol. 30:290–291). The frog voucher is deposited in the Arkansas State University, Herpetological Museum as ASUMZ 25326.

The anterior and posterior chambers of the eye were filled with a highly cellular exudate consisting of predominately mixed mononuclear inflammatory leukocytes comprised of lymphoplasmacytes, large histiocytic macrophages, a few heterophil granulocytes, and tenuous strands of fibrin that often adhered to the lens capsule (Figs. 2A–B). The crystalline lens had lost much of its normal concentric onion-like lammelated architectural pattern; the affected lens substance was lighter staining and possessed a patchy and foamy appearance (Figs. 2A–B). Small mononuclear cells were found adjacent to the lens capsule and numerous large macrophages with engulfed numerous Gram-negative and fewer numbers of Gram-positive bacteria were identified (Figs. 2C–D). In addition, engulfed melanin pigment granules were observed (Fig. 2D).

Ophthalmologic disorders have been recorded for many vertebrate species. Often, the animal involved is a captive and, thus, its ocular disease is more readily observed during daily animal care and maintenance. In this case, the affected frog was wild-caught, having already developed its severe ophthalmologic condition. In addition, 20 additional *H. chrysocelis* from the same site were not found to have this condition or any other noticeable gross pathology. Except for the finding of intra- and extracellular coccobacillary bacteria within the uvietic exudate, the etiology of the severe inflammation and subsequent lens disruption is unknown but, it is noteworthy because of its severity. Certainly, trauma associated with the introduction of pathogenic bacteria is a strong possibility. Had the frog escaped septicemic dissemination of the infection, it is conjectural as to whether the frog could have survived after having lost vision in one eye; however, there have been recorded instances of other wild-caught frogs after having lost the sight of an eye due to trauma, corneal, and lenticular opacities (Frye and Williams, *op. cit*.) Therefore, it can be surmised that in these cases binocular vision was not essential for prey capture and survival under wild conditions.

CTM thanks the Texas Parks and Wildlife Department for Scientific Collecting Permit No. 42-02 and S. E. Trauth (ASUMZ) for curatorial assistance.

Submitted by **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA (e-mail: cmcallister@csc.edu); **FREDRIC L. FRYE**, La Primavera Farm, 33422 Highway 128, Cloverdale, California 95425-9428, USA (e-mail: FredFrye@aol.com); and **STEPHEN R. GOLDBERG**, Department of Biology, Whittier College, Whittier, California 90608, USA (e-mail: sgoldberg@whittier.edu).

LEPTODACTYLUS MELANONOTUS (Sabinal Frog). **DIET**. Some species of the Leptodactylidae are known for diverse diets (Savage 2002. Amphibians and Reptiles of Costa Rica. Univ. Chicago Press, Chicago, Illinois) and have tadpoles known for their intraspecific cannibalism (Silval et al. 2005. J. Nat. Hist. 39[7]:555–566). Herein we document a previously unknown scavenging habit of *Leptodactylus melanonotus*. In November 2005 in a Lowland Tropical Wet Forest in the northeastern coast of Costa Rica near Barra del Colorado, an adult male *L. melanonotus* was found advertising on the periphery of a desiccated ephemeral pool. The pool contained 48 dead, newly hatched *Rana vaillanti* tadpoles. Upon capture the *L. melanonotus* exhibited signs of a recent meal, and while being measured regurgitated 17 newly hatched *R. vaillanti* tadpoles. Among the stomach contents was an assortment of small springtail (Collembola) body parts and an intact aquatic Belostomatidae (Hemiptera). The adult frog weighed 3.5 g after regurgitating the tadpoles, and the regurgitated tadpoles totaled 2.3 g wet weight (the frog consumed 65% of its own body weight in tadpoles). The specimen and its stomach contents were deposited in the University of Costa Rica (UCR) collections within the Cãno Palma Biological Station specimens. To our knowledge this is the first record of consumption of tadpoles of another anuran species by adult *L. melanonotus*.

We thank M. Springer (University of Costa Rica) and F. St. Ours (University of Massachusetts, Boston) for help in invertebrate identification, Xavier Guevara of the Ministerio de Recursos Naturales Energia y Minas for permits, and the Canadian Organization for Tropical Education and Rainforest Conservation for permission to study at Caño Palma Biological Station.

Submitted by **TODD R. LEWIS,** 4 Worgret Road, Wareham, Dorset, BH20 4PJ, United Kingdom (e-mail: biotropical@gawab.com); **COLIN RYALL**, Farnborough College of Science and Technology, Boundary Road, Farnborough, Hampshire, GU14 6SB, United Kingdom; and **THOMAS C. LADUKE,** East Stroudsburg University, 200 Prospect St., East Stroudsburg, Pennsylvania 18301-2999, USA.

RANA CASCADAE (Cascades Frog). **ALBINISM.** On 5 Aug 2002, we observed eight albino *Rana cascadae* larvae at a pond (65 m × 100 m; < 1 m deep) located 1.2 km NNW of Three Creek Lake, Deschutes County, Oregon, USA (44.1119444°N, 121.6313889°W; elev. 1960 m). Five albino larvae were captured and SVL measured (average SVL 25.4 mm, range 24–27 mm) and developmental stage estimated (stage 28–39; Gosner 1960. Herpetologica 16:183–190). During the survey, we observed 10 *R. cascadae* larvae with normal pigmentation that were of similar size and stage as the albino larvae. Albino larvae had a slight orangish hue in their eyes and throughout the body, typical of albinos of other species (Dyrkacz 1981. SSAR Herpetol. Circ. No. 11, 31 pp.). The brain, spinal column, internal organs, and almost completely developed forelegs were clearly visible through nearly transparent skin. We collected one albino larva (JC Braly Vertebrate Collection, Oregon State University 01550). We returned to the site on 16 Aug 2002 and collected a second albino larva and one non-albino larva. These two larvae transformed in the lab and died after 13 months.

In the only record of albinism in *R. cascadae* we were able to find, Altig and Brodie (1968. Wasmann J. Biol. 26:241–242) reported 13 albino larvae from 1965 and five from 1966 at a small pond "near Three Creek Lake, Deschutes County, Oregon." We were unable to confirm the exact pond through review of the museum records or communication with the authors. *Rana cascadae* breed in at least three lentic sites within 1.5 km of our site (C. Brown, unpubl. data: BM, pers. obs.). Nonetheless, our observations suggest the presence of albinos within a frog population > 35 years after original description. Our literature survey revealed only one report of albinism in the same anuran population in > 1 year (*Pseudacris triseriata* in two consecutive years; Corn 1986. J. Hered. 77:164–168).

This work was supported by the USGS State Partnership Program and the Amphibian Research and Monitoring Initiative (ARMI). We thank S. Borrego for field assistance, and two anonymous reviewers for their comments. All animals were handled under an Oregon Dept. of Fish and Wildlife scientific collecting permit.

Submitted by **BROME McCREARY** (e-mail: brome_mccreary@usgs.gov), **CHRISTOPHER A. PEARL**, U.S. Geological Survey, Forest and Rangeland Ecosystem Science Center, 3200 SW Jefferson Way, Corvallis, Oregon 97331, USA.

RANA CATESBEIANA (American Bullfrog). **LITHOPHAGY**. Gravel, sand, and plant matter have been documented in *R. catesbeiana* stomachs during numerous diet studies. Korschgen and Moyle (1955. Amer. Midl. Nat. 54[2]:332–341) documented a variety of plant material and a small amount of gravel in *R. catesbeiana*. In Arkansas, McKamie and Heidt (1974. Southwest. Nat. 19:107–111) found a 15.9 g rock and plant matter in 28% of the stomachs examined. Plant material, rocks, and gravel are likely ingested accidentally.

On 8 June 2006, an adult *R. catesbeiana* was collected ca. 15 mi. SE of Mena, Arkansas, USA in a mineshaft ca. 18 m from the entrance. Upon collection it was apparent that it had a full stomach. Dissection revealed a single salamander skeleton (presumably *Plethodon caddoensis*), a small piece of wood, and inorganic matter including rocks and grit. The wood's mass was 0.07 g. The inorganic material ranged from tiny grains to large pebbles. There were 34 rocks totaling 12.27 g. Average mass was 0.36 g/rock (range 0.05–1.63 g) not including the fine grit.

This frog likely ingested the gravel while attempting to feed on salamanders that frequent the mineshaft. The specimen is deposited in the herpetology collection at the Arkansas State University Museum of Zoology (ASUMZ 30143).

Submitted by **JOSH ENGELBERT** (e-mail: josh.engelbert@smail.astate.edu), **MELISSA PATRICK,** and **STANLEY E. TRAUTH,** Department of Biological Sciences, Arkansas State University, P.O. Box 599, State University, Arkansas 72467-0599, USA.

RANA CLAMITANS (Bronze Frog). **RANID AGGRESSION AND INTERSPECIES AMPLEXUS**. On 3 July 2007, we observed and video-recorded inter- and intra-specific aggressive behavior of two male Bronze Frogs (*Rana clamitans clamitans*) and a male-female pair of Southern Leopard Frogs (*R. sphenocephalus*) in amplexus. The interactions occurred in the shallows of a slow moving creek in Santa Rosa County, Florida, USA, at ca. 1300 h and lasted for > 45 min. We first observed the behavior when the two Bronze Frogs began calling with increased frequency and then noticed one Bronze Frog aggressively moving (i.e., chasing) towards the pair of leopard frogs. The pair of leopard frogs submerged and crawled along the bottom of the creek, apparently to avoid the male Bronze Frog. However, each time they emerged, the Bronze Frog would reinitiate aggressive chasing and calling. After 12 min, the second male Bronze Frog moved toward the attacker and both frogs began grappling and ramming their heads together. Every 1–10 sec they would vocalize and jump between aggressive behaviors. After ca. 46 sec, the first Bronze Frog (and the larger of the two) retreated. The second, smaller Bronze Frog then chased the leopard frogs, which continued submerging and emerging to evade the aggressor. After another 50 sec of chase, the Bronze Frog amplexed the pair of leopard frogs (i.e., the male) using axillary amplexus. Next, the male leopard frog emitted a release call while the female leopard frog crawled along the bottom of the creek, carrying both males. The Bronze Frog remained amplexed for 96 sec before the first Bronze Frog returned and physically removed the amplexed Bronze Frog from the leopard frogs. Finally, the leopard frogs traveled a few more feet and the female slowly began depositing her eggs while the Bronze Frogs resumed their intra-specific aggression. After another 120 sec, the egg deposition ceased, and the larger Bronze Frog again chased the pair of leopard frogs. The leopard frogs quickly separated and retreated in different directions and the Bronze Frog eventually abandoned his pursuit and left the area.

Submitted by **STEPHEN C. RITCHIE**, **BRANDON K. RINCON**, and **THOMAS A. GORMAN** (e-mail: gormant@vt.edu), Department of Fisheries and Wildlife Sciences, Virginia Tech, 100 Cheatham Hall, Blacksburg, Virginia 24061, USA.

SPEA HAMMONDII (Western Spadefoot). **PREDATION AND USE AS BURROW DECORATIONS**. During 19–22 April 2004, I observed two breeding pairs of Western Burrowing Owls (*Athene cunicularia hypugaea*) located 20 m apart within a vernal pool system near Goshen, Tulare Co., California, USA (36.3451°N, 119.3991°W, NAD83/WGS84, 88 m elev.). Each pair used a network of burrows clustered 1–3 m apart. The primary burrow used by the pair at each cluster was decorated with domestic sheep dung, skunk (*Mephitis mephitis*) fur, and desiccated remains of *Spea hammondii*. The entrances of the burrows in each cluster exhibited 3–5 toads in various states of disarray. Hindquarters were present without the head or thoracic cavity, and nearby were the heads and upper bodies either still attached or in separate pieces. The soft organs of the thoracic cavity were missing. Both the Western Spadefoot and Western Burrowing Owl are species of special concern in California.

Burrowing Owls commonly decorate their burrows with a variety of items, such as dung, grass, paper, cotton, and dried moss (Levey et al. 2004. Nature 431:39; Smith and Conway 2007. Anim. Behav. 73:65–73). Flattened mummified remains of the Southern

Toad (*Bufo terrestris*), assumed to be roadkills, have been found at Burrowing Owl burrows as well (D. Levey, pers. comm.). The *S. hammondii* were readily available to the Burrowing Owls because their burrows were within a vernal pool system, a habitat where spadefoots commonly occur.

The Burrowing Owl has been infrequently reported to prey on toad species (Haug et al. 1993. Burrowing Owl. *Athene cunicularia*. *In* A. Poole and F. Gill [eds.], The Birds of North America, No. 61. Academy of Natural Sciences, Philadelphia, and American Ornithologists' Union, Washington, D.C.). Although toads commonly have toxins in their dorsal surfaces, owls and other predatory birds are able to avoid these toxins by consuming the vulnerable ventral portions (Olson 1989. Copeia 1989:391–397). Ervin et al. (2007. Herpetol. Rev. 38:197–198) were the first to report Burrowing Owl predation on Western Spadefoot adults, however, only the tongues were consumed in those cases. In this instance, the Burrowing Owl may have captured and consumed portions of *S. hammondii* and then used their remains as burrow decorations.

Submitted by **HOWARD O. CLARK, JR**., H.T. Harvey and Associates, 423 Fallbrook Avenue, Suite 202, Fresno, California 93711, USA; e-mail: hclark@harveyecology.com.

TESTUDINES – TURTLES

DERMOCHELYS CORIACEA (Leatherback Sea Turtle). **NESTING**. *Dermochelys coriacea* is currently classified by the Brazilian Ministry of the Environment and the IUCN (World Conservation Union) as critically endangered. Although occasional nesting has been observed on the Brazilian coast (see Barata and Fabiano 2002. Marine Turtle News. 96:13–16), the only site reporting regular Leatherback nesting is located in a restricted area on the northern coast of the state of Espírito Santo, with an extent of ca. 200 km (18.35°S, 39.67°W and 20.07°S, 40.17°W) (Marcovaldi and Marcovaldi 1999. Biol. Conserv. 91:35–41). Here, we present the first record of *D. coriacea* nesting on the Brazilian northern coast. On 17 July 2004 at about 1800 h, within the limits of the Environmental Protection Area of the Delta do Parnaíba, Arrombado beach, Luís Correia city, state of Piauí (02.9097°S, 41.5325°W; 3 m elev.; Datum SAD69 IBGE/BR), we observed one *D. coriacea* deposit a clutch of 108 eggs. The nest was monitored after oviposition; 96 hatchlings emerged after an incubation period of 58 days (Fig. 1). Local fishermen also have reported the sporadic presence of *D. coriacea* nesting along Piaui's beaches. Although this species may be expected in Piauí state, considering its distribution in the neighboring states of Ceará and Maranhão (Barata et al. 2004. J. Mar. Biol. Assoc. U.K. 84:1233–1240), this is the first documented record of this species nesting in Piauí state.

FIG. 1. Infrared photography of newborns of *Dermochelys coriacea* leaving the nest after sunset at about 1800 h. Photo by Daniel Loebmann.

Submitted by **DANIEL LOEBMANN,** Departamento de Zoologia, Instituto de Biociências, Universidade Estadual Paulista, Rio Claro, São Paulo, Brazil, Caixa Postal 199, CEP 13506-970 (e-mail: contato@danielloebmann.com); **JEFFERSON FRANCISCO ALVES LEGAT**, **ANGELA PUCHNICK LEGAT**, Embrapa Meio – Norte, BR-343, Km 35, Parnaíba, Piauí, Brazil, Caixa Postal 341, CEP 64200-97; **RICARDO COSTA RODRIGUES DE CAMARGO**, Embrapa Meio – Norte, Av. Duque de Caxias, 5650, Teresina, PI – Brazil, Caixa Postal 001, CEP 64006-220; **SILMARA ERTHAL, MAGNUS MACHADO SEVERO**, Ibama, Unidade de Conservação APA Delta do Parnaíba, Rua Merval Veras nº 80, Parnaíba, Piauí, Brazil, Bairro do Carmo, CEP 64.200-300; and **JOÃO MARCO DE GÓES**, UESPI Parnaíba, Av. Nossa Sra. de Fátima, s/n, Parnaíba, Piauí, Brazil, Bairro de Fátima, CEP: 64202-220.

GLYPTEMYS INSCULPTA (Wood Turtle). **HATCHLING BEHAVIOR**. Hatchlings of *Glyptemys insculpta* typically emerge from the nest in late summer and early fall (Buech et al. 2004. Herpetol. Rev. 35:54; Harding and Bloomer 1979. Bull. New York Herpetol. Soc. 15:9–26; Tuttle and Carroll 2005. Northeast. Nat. 12:331–348), even at the northern limits of their range (Brooks et al. 1992. Can. J. Zool. 70:462–469; Walde et al. 2007. Herpetol. Conserv. Biol. 2:49–60). In New Hampshire, newly-emerged hatchling *G. insculpta* were observed to migrate long distances (up to 445 m), spending several days and nights on land before reaching aquatic habitats where they are presumed to hibernate (Tuttle and Carroll, *op. cit.*). Parren and Rice (2004. Northeast. Nat. 11:229–233) reported a suspected terrestrial overwintering by a neonatal *G. insculpta* in Vermont.

As a follow up to a study of the nesting ecology of *G. insculpta* (Walde et al. 2007, *op. cit.*), hatchlings were tracked using fluorescent pigments (Butler and Graham 1993. Herpetol. Rev. 24:21–22) as they dispersed from nests in August through October 1997.

Hatchlings often used forms as resting places during periods of inactivity. Typically, forms consisted of a small excavation into which the turtle could fit; often under vegetation, rocks, or other objects. When in a form, hatchlings were observed to withdraw their head and limbs into their shells. Seven hatchlings emerged from one nest on 23 September 1997 and all hatchlings were treated with fluorescent pigments and subsequently released. Nocturnal tracking revealed five of the seven hatchlings were still on land, but not in forms or under any insulative material. At 0730 h on 24 September 1997, there was frost on the ground, ice on puddles, and the air temperature, measured with a handheld thermometer at the nesting site was -3°C. The five turtles were in the same position as the previous night, and all of them had their limbs and necks extended and were covered in ice crystals. Upon handling, two individuals were rigid, as if they were frozen solid, with their limbs rigid and extended. Both turtles were placed back where they were found. By 1100 h the temperature was well above freezing and four of the five hatchlings had moved; whereas, the fifth hatchling was alive but had not moved. Air temperature remained above 0°C the following two nights, when four of the five hatchlings were still on land and dug into forms; a fifth hatchling was not re-located.

Natural freeze tolerance, defined as the ability to survive freezing of body tissues under ecologically-relevant conditions, has been reported for several species of frogs, lizards, snakes, and turtles (Storey and Storey 1996. Ann. Rev. Ecol. Syst. 27:365–386). Hatchling turtles of several emydid species survive freezing to temperatures -3°C for at least 72 h (Costanzo et al. 2006. J. Comp. Physiol. B 176:697–707). Our observations suggest that hatchling *G. insculpta* can endure freezing at similar subzero temperatures, although we were not able to record actual body temperatures. In regions where overnight frosts occur during the fall emergence period, a tolerance for freezing of body tissues may increase survival of terrestrially migrating hatchling turtles.

Funding, equipment, and personnel for this research were generously provided to the St. Lawrence Valley Natural History Society by Parks Canada, Fondation de la Faune du Québec, and Société de la faune et des parcs du Québec.

Submitted by **ANDREW D. WALDE**, 7686 SVL Box, Victorville, California 92395, USA (e-mail: awalde@hotmail.com); **PATRICK J. BAKER**, Department of Zoology, Miami University, Oxford, Ohio 45056, USA (e-mail: bakerpj@muohio.edu); and **RAYMOND A. SAUMURE**, Research Division, The Springs Preserve, 1001 South Valley View Boulevard, Las Vegas, Nevada 89107, USA (e-mail: insculpta@gmail.com).

GLYPTEMYS INSCULPTA (Wood Turtle). **MAXIMUM CLUTCH SIZE.** Gammons (1871. Amer. Nat. 4:53) reported a clutch size of 18 eggs for *Glyptemys insculpta*. Carr (1952. Handbook of Turtles: The Turtles of the United States, Canada, and Baja California. Cornell Univ. Press. Ithaca, New York) rejected the account however, assuming it to be some sort of "hallucination" because of the odd nesting behaviors described by Gammons. Nevertheless, Harding (1977. Herpetol. Rev. 8:34) subsequently reported a clutch size of 18 eggs for *G. insculpta* in Michigan.

During studies of the nesting ecology of *G. insculpta* in Québec, Canada (Walde et al. 2007. Herpetol. Conserv. Biol. 2:49–60), we observed a record clutch for this species. On 17 June 1997 at 0930 h, a *G. insculpta* was observed constructing a nest. It was not possible to count the number of eggs, as approaching would have disturbed the other Wood Turtles present in the nesting area (Walde et al. 2007, *op. cit.*). Seventeen hatchlings emerged from this nest cavity; 16 on 27 August 1997, and an additional one on 30 August 1997. When the nest was excavated on 9 September 1997, there were 17 egg shells and an additional three eggs with dead embryos that were putrid and covered in fly larvae. Thus, the total clutch size was 20 eggs. The female that laid this clutch had a carapace length (CL) of 224 mm, plastron length (PL) of 214 mm, and mass of 1475 g. She was the largest of 39 females observed to nest during the two-year study, and was the second largest observed in the population of 83 females (Walde et al. 2003. Can Field-Nat. 117:377–388). Walde et al. (2007, *op. cit.*) reported that clutch size within this population was significantly positively correlated with CL, PL, and mass. There appears to be some level of geographic variation present, as maximal clutch sizes from northern populations tend to be larger than those from southern ones (Walde et al. 2007, *op. cit.*).

We acknowledge Parks Canada, Fondation de la Faune du Québec, and Société de la faune et des parcs du Québec for provided funding, equipment and personnel to the St. Lawrence Valley Natural History Society for this study. Many individuals assisted with this project, to them we are grateful. We specifically acknowledge Denis Masse of Parks Canada for logistical support while in the field.

Submitted by **ANDREW D. WALDE**, 7686 SVL Box, Victorville, California 92395, USA (e-mail: awalde@hotmail.com); and **RAYMOND A. SAUMURE**, Research Division, The Springs Preserve, 1001 South Valley View Boulevard, Las Vegas, Nevada 89107, USA (e-mail: insculpta@gmail.com).

GRAPTEMYS GEOGRAPHICA (Northern Map Turtle). **HABITAT.** *Graptemys geographica* is typically described as inhabiting larger, slow-moving rivers, and lakes with mud bottoms rather than small creeks and ponds (Conant and Collins 1998. A Field Guide to Amphibians and Reptiles of Eastern and Central North America. 3rd Edition. Houghton Mifflin Co., Boston, Massachusetts. 616 pp.; Ernst et al. 1994. Turtles of the United States and Canada. Smithsonian Inst. Press, Washington D.C. 578 pp.) Here I report two encounters of *G. geographica* in Marshall Co., Tennessee, in 2006 and 2007, in or near Bear Creek. These encounters happened at least 20 miles from the nearest large river (Duck River) and both encounters were with large females.

A mature female *G. geographica* was found during the morning of 23 April 2005 crossing Mooresville Highway (Highway 373) at a bridge (35.4545°N, 86.9304°W). This occurrence was after a heavy rain and water levels in Bear Creek were elevated. The reason for the turtle crossing the bridge was unclear; other than the creek, there are no other water sources in the direction from which she had presumably traveled, or in the direction she was moving. This large (though unmeasured) female was retrieved from the bridge, photographed, and released back into Bear Creek in the location that she was apparently headed.

The second *G. geographica* female was encountered in the afternoon of 1 June 2006 in the same general area, but was further away from the road and was further upstream (35.45005°N, 86.93107°W). This female, encountered while constructing a nest, was photographed but not otherwise disturbed. The water level in Bear Creek at the time was not abnormally elevated. The width of the creek nearest to where this female was found does not exceed 1.8 m and the depth does not exceed 0.3 m. The success of the nest was not determined because of time constraints.

Photo vouchers are deposited at Austin Peay State University Museum of Zoology: APSU 18305 (23 April 2005 record), APSU 18306 (1 June 2006 record).

Submitted by **NATHAN HAISLIP**, 3817 Mooresville Highway. Culleoka, Tennessee 38451, USA; e-mail: nhaislip@utk.edu.

MANOURIA IMPRESSA (Impressed Tortoise). **ENDOPARASITES.** *Manouria impressa*, a medium-sized, forest dwelling tortoise, ranges from Myanmar and Malaysia to Vietnam and China (Iverson 1992. A Revised Checklist with Distribution Maps of the Turtles of the World. Privately printed, Richmond, Indiana. 278 pp). To our knowledge, no endoparasites have been reported from *M. impressa*. The purpose of this note is to report an occurrence of the nematode *Cissophyllus roseus* in *M. impressa*.

Wild-caught *Manouria impressa* confiscated by the US Fish and Wildlife Service were sent to Zoo Atlanta for triage and veterinary care. *Manouria impressa* is a particularly difficult species to keep in captivity. Wild-caught tortoises often succumb to a myriad of illnesses (Innis C et al. 2006. J. Herpetol. Med. Surg. 16:49–52). The confiscated tortoises appeared in good health and had adequate body weight. They were kept in quarantine for 45 days. On a routine fecal examination, during the first week in quarantine, we found two male and four gravid female nematodes. The nematodes were fixed in alcohol and later cleared in lactophenol for microscopic examination during which they were identified as *Cissophyllus roseus*.

Railliet and Henry (1912. Bull. Soc. Path. exot., Paris. 5:251–259) described the nematode *Cissophyllus laverani* from an Indomalaysian tortoise, *Testudo emys* (currently *Manouria emys*) and in a second paper (Railliet and Henry 1915. Bull. Soc. Path. exot., Paris. 8:446–452) mentioned that the nematode *Cucullanus roseus* described by Leidy (1851. Proc. Acad. Nat. Sci., Philadelphia 5:205–210) from the tortoise *Manouria fusca* (currently *Manouria emys*) had greater affinity for the genus *Cissophyllus* than for the genus *Cucullanus*; however, synonymy was not established. Walton (1927. Proc. Acad. Nat. Sci., Philadelphia 79:49–163) revised the Leidy collection and did synonymize *Cucullanus roseus* with *Cissophyllus laverani*, the name becoming *Cissophyllus roseus*.

Prior to our observation, *Manouria emys* was the only known host for *C. roseus*. *Manouria impressa* represents a new host record and only the second host known for *C. roseus*. To our knowledge there are no life history studies for this nematode, but zoological institutions and those in the private sector should be aware of the possibility of infection when introducing wild-caught specimens into established collections.

Submitted by **SAM RIVERA**, Department of Veterinary Services, Zoo Atlanta, 800 Cherokee Avenue S.E. Atlanta, Georgia 30315, USA (e-mail: srivera@zooatlanta.org); **DANA AMBROSE**, Athens Diagnostic Laboratory, College of Veterinary Medicine, University of Georgia, Athens, Georgia, USA; and **CHARLES R. BURSEY,** Department of Biology, Pennsylvania State University, Shenango Campus, Sharon, Pennsylvania 16146, USA (e-mail: cxb13@psu.edu).

STERNOTHERUS ODORATUS (Stinkpot) **MINIMUM SIZE / GROWTH**. On 26 April 2007 one of us (JKT) collected a juvenile *Sternotherus odoratus* using a dip net at Stump Lake, Jersey County, Illinois, USA (38.9912°N, 90.5554°W; NAD27 datum). This turtle had presumably hatched in 2006, and likely emerged from the nest in the fall of 2006, as this species typically does not overwinter in the nest (Ernst et al. 1994. Turtles of the United States and Canada. Smithsonian Institution Press, Washington, D.C. 578 pp.). Most reports on turtle size present record maximum lengths (e.g., Blasus et al. 2004. Herpetol. Rev. 34:54; Lubcke and Wilson 2006. Herpetol. Rev. 37:339; Tucker et al. 2006. Herpetol. Rev. 36:453–455). Here we report on the smallest non-hatchling turtle. Despite the possibility that the turtle was active for some weeks or months, it was remarkably tiny (Fig. 1). It weighed 1.54 g and had a carapace length of 18.8 mm, a carapace width of 17.7 mm, a carapace height of 11.5 mm, and a plastron length of 13.8 mm (all measurements made using methods of Tucker et al. 2006, *ibid*.). This individual was much smaller than eight recently emerged hatchlings (= those with no scute annuli on the carapace scutes) caught in Stump Lake during fall of 2005 (29 September and 5 October) using minnow traps. These eight averaged 4.58 g in wet mass (SD = 0.74 g, range = 3.70–5.90g), 27.4 mm carapace length (SD = 1.92 mm, range = 25–30 mm), 24.0 mm in carapace width (SD = 2.14 mm, range = 21–27 mm), 14.5 mm in carapace height (SD = 1.31 mm, range = 12–16 mm), and 19.4 mm in plastron length (SD = 1.51 mm, range = 17–22 mm). This species grows rapidly after hatching. For instance, nine individuals in their first full season of growth (= those with one

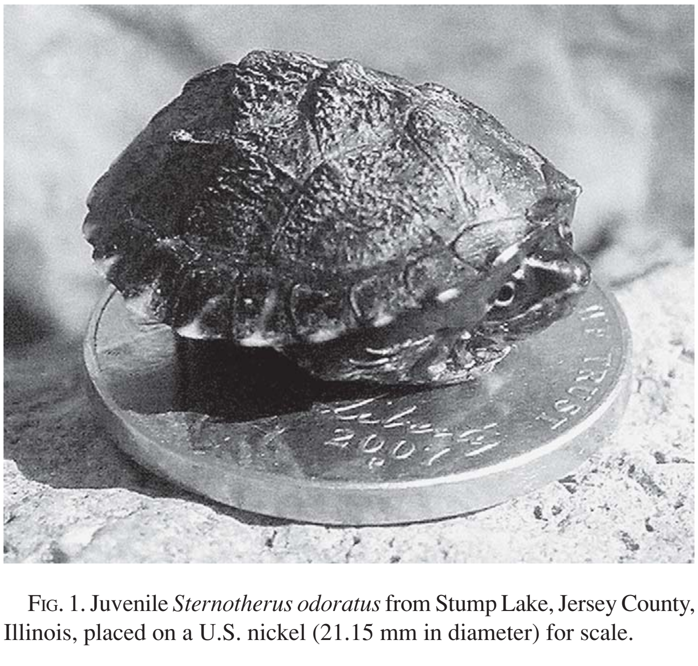


FIG. 1. Juvenile *Sternotherus odoratus* from Stump Lake, Jersey County, Illinois, placed on a U.S. nickel (21.15 mm in diameter) for scale.

scute annulus on the carapace scutes) from Stump Lake were caught between 30 June and 29 July 2005 with minnow traps. These juveniles had emerged from eggs laid in 2004. They averaged 10 g (SD = 0.80 g, range = 8.9–11.1 g) in wet mass, 34.3 mm in carapace length (SD = 1.22 mm, range = 32–36 mm), 30.2 mm in carapace width (SD = 1.20 mm, range = 29–32 mm), 19.2 mm in carapace height (SD = 0.67 mm, range = 18–20 m), and 24.8 mm in plastron length (SD = 1.09 mm, range = 24–27 mm). Thus juveniles from Illinois nearly double their size in the first full year after hatching. Reports on first year growth are few for this species (see Ernst et al. 1994, *op. cit.*, for a review). Nonetheless, Risley (1933. Pap. Michigan Acad. Sci. Arts Lett. 17:685–711) found that yearling Stinkpots averaged 32.5 mm in carapace length, similar to the 34.3 mm size we found.

The Stump Lake juvenile was smaller than 95 hatchlings from eggs induced from Jersey and Calhoun county females. These 95 eggs averaged 3.94 g in wet mass (SD = 0.47 g, range = 2.85–5.67 g) (see Tucker 1999. Bull. Maryland Herpetol. Soc. 35:61–75 for details on egg collection). The 95 hatchlings averaged 2.74 g in weight (SD = 0.32 g, range = 1.94–3.56 g), 22.8 mm in carapace length (SD = 1.15 mm, range = 19.6–25.2 mm), 16.7 mm in carapace width (SD = 1.44 mm, range = 12.8–20 mm), and 17.5 mm in plastron length (SD = 0.96 mm, range = 15.2–19.8 mm). These hatchlings were weighed and measured the day they left the egg (methods of Tucker et al. 2006, *op. cit.*). Carapace height was not measured for these hatchlings to avoid interference with residual yolk retraction.

Nonetheless, hatchlings of several species of Kinosternidae may leave the egg even smaller than the juvenile that we caught. Few published records of kinosternid hatchlings report mass, and the next smallest individual by weight was a hatchling Stinkpot that weighed 1.6 g (Mitchell 1994. The Reptiles of Virginia. Smithsonian Institution Press, Washington, D.C. 352 pp.). Many authorities report linear shell dimensions and several have reported hatchlings with carapace lengths smaller than 18.8 mm. These include *Sternotherus odoratus* with carapace length of 18.5 mm (Ernst et al. 1994, *op. cit.*; Mitchell 1985. Copeia 1985:941–949), *Kinosternon subrubrum* with carapace length of 16.7 mm, *K. baurii* with carapace length of 16.5 mm, and *K. flavescens* with carapace length of 17.7 mm (all from Ernst et al. 1994, *op. cit.*). These are, however, measurements of hatchlings fresh from the eggs and not of free ranging turtles such as the one we report. It is interesting that such genuinely minute juveniles only occur in North American Kinosternidae and apparently not in any non-North American turtles (see reviews by Pritchard 1979. Encyclopedia of Turtles. T.F.H. Publications, Inc. Ltd., Neptune, New Jersey, 895 pp.; Ernst and Barbour 1989. Turtles of the World. Smithsonian Institution Press, Washington, D.C., 313 pp.). Hatchlings tend to be much larger (25 mm + carapace length) in other turtle species (for a review of many worldwide species see Table 2 in Ewert 1979 (*In* Harless and Morlock [eds.], Turtles: Perspectives and Research. The embryo and its egg: development and natural history, pp. 333–413, John Wiley and Sons, New York). The adaptive significance for such small hatchlings has never been investigated and it may be simply a reflection of the small adult sizes of many kinosternids. Egg size can be strongly constrained in such small turtles (e.g. Tucker et al. 1978. Bull. Maryland Herpetol. Soc. 14:10–22; Congdon and Gibbons 1987. Proc. Natl. Acad. Sci. 84:4145–4147).

All hatchlings and juveniles that we examined were or will be released at the original collecting sites or at the collecting sites of the female parent. We thank Emily Dustman, Michael Avara, and Megan Dooling for help in collecting turtles. This research was partially funded by a grant to JKT from the National Great Rivers Research and Education Center, Richard E. Sparks, director of research and by support from the Illinois Natural History Survey, Great Rivers Field Station, John H. Chick, Director.

Submitted by **JOHN K. TUCKER** (e-mail: jktucker@inhs.uiuc.edu) and **CHAD R. DOLAN** Great Rivers Field Station, Illinois Natural History Survey, 8450 Montclaire Avenue, Brighton, Illinois 62012, USA; and **JAMES T. LAMER**, Department of Biology, Western Illinois University, 1 University Circle, Macomb, Illinois 61455, USA.

SQUAMATA – LIZARDS

ANOLIS AENEUS (Grenadian Bush Anole), ***ANOLIS RICHARDII*** (Grenadian Tree Anole). **NECTARIVORY / POLLINATION**. Recent observations (e.g., Perry and Lazell 1997. Herpetol. Rev. 28:150–151) have contributed to the general notion that island-dwelling lizards may both feed on nectar and act as pollinators (Olesen and Valido 2003. Trends Ecol. Evol. 18:177–181). Here, we report accounts of nectarivory and presumptive pollinating behavior on two native plant species, *Charianthus grenadensis* and *Marcgravia umbellata*, by *Anolis aeneus* and *Anolis richardii*, in Grenada, Lesser Antilles. Our observations were made in the elfin forest along the Mt. Qua Qua trail within Grand Etang National Park in the montane interior (elev. 690–710 m) of Grenada.

Between 0828 and 0830 h on 22 June 2006, we observed *A. aeneus* visiting two flowers of *C. grenadensis*. This generalized anole (Germano et al. 2003. Caribb. J. Sci. 39:68–76) was slowly sliding its head into the broad opening of the flowers and licking nectar from the bottom of these. In the process, the anole touched both anthers and stigma with its head. *Charianthus grenadensis*, endemic to montane Grenada, is believed to be adapted for hummingbird pollination (Penneys and Judd 2005. Syst. Bot. 30:559–584) as it displays several classic ornithophilous features, e.g., bright red corolla, no obvious scent, and a large amount of diluted nectar (24-h sample of bagged flowers; volume: = 53.0 ± 24.8 µl; concentration: = 11.6 ± 0.9% weight/weight sucrose equivalents, N = 13; AT and LHA, unpubl. data).

On 22 June 2006, we observed two *A. richardii* visiting a single inflorescence of *M. umbellata*. The length of the visits was 15 and 6 min (1135–1150 and 1242–1248 h). Both anoles exhibited pollinating behavior, i.e., the head and dorsal part of the body brushed against the anthers and stigma of the flowers as they licked nectar from several of the pendulous tubular nectaries of the inflorescence (Fig. 1). *Marcgravia umbellata*, restricted to the Lesser Antilles, is adapted for bird and/or bat pollination (Dressler 1997. Curtis's Bot. Mag. 14:130–136; Zusi and Hamas 2001. Caribb. J. Sci 37:274–278). Its flowers have no scent, a large amount of dilute nectar (24-h sample of bagged flower; volume = 119.5 µl; concentration = 9% weight/weight sucrose equivalents; N = 1; AT and LHA, unpubl. data) and dull brown-green colors.

Our observations reveal that *Anolis* lizards have the potential to pollinate both *C. grenadensis* and *M. umbellata*. Nectar-feeding

FIG. 1. *Anolis richardii* visiting *Marcgravia umbellata*. The lizard was feeding on nectar and touching both anthers and stigma. Photo by Allan Timmermann.

lizards acting as pollinators on islands may result from reduced insect abundance, fewer predators and less interspecific competition than on comparable mainland environments, allowing density compensation and niche broadening by island lizards (Olesen and Valido 2003. Trends Ecol. Evol. 18:177–181). In the Caribbean, hurricanes may be an additional factor offering lizards the opportunity to expand their diet to include energy-rich nectar from typically bird-pollinated flowers. Hurricanes can cause nectarivorous bird populations to crash, subsequently limiting pollination and resulting in surplus nectar in bird-pollinated flowers (Rathcke 2000. Ecology 81:1951–1958). Whether lizards are important backup pollinators of otherwise bird-pollinated plants in the aftermath of hurricanes needs investigation.

We are grateful to the Department of Forestry and National Parks, Ministry of Agriculture, Grenada for help throughout our fieldwork in Grenada. We thank A. Valido for comments and R. Powell for kindly identifying the *Anolis* lizards. Funding was provided by Aarhus University Faculty of Natural Science, Knud Højgaard, Augustinus and Carlsberg Foundations, the Danish National Science Research Council and WWF-Denmark/Novozymes.

Submitted by **ALLAN TIMMERMANN**, **BO DALSGAARD**, **JENS M. OLESEN**, **LAILA H. ANDERSEN**, and **ANA M. MARTÍN GONZÁLEZ**, Department of Ecology & Genetics, University of Aarhus, Ny Munkegade, Building 1540, DK-8000 Aarhus C, Denmark; e-mail (AT): allantimmermann@yahoo.dk.

ASPIDOSCELIS COSTATA HUICO (Blue-chested Whiptail). **REPRODUCTION**. *Aspidoscelis costata huico* (*sensu* Reeder et al. 2002. Am. Mus. Novit. 3365:1–61) is among many named, but biologically poorly known reptiles from remote areas of México. Zweifel (1959. Bull. Am. Mus. Nat. Hist. 117:57–116) described the subspecies based on 193 specimens from the type locality (Peñitas, 19.2 km SE of Rosamorada, Nayarit) and other mainland sites in the states of Sinaloa, Nayarit, Jalisco, and Zacatecas; and Isla Isabel, Nayarit. Here, I provide reproductive data (clutch ranges and mean ± SE, SVL ranges and mean ± SE) for a subspecies which Zweifel (*op. cit.*) noted only maximum sizes of males and females (both 106 mm SVL) and described a mating incident observed at La Vuelta, Nayarit, on 25 August 1957 by T. H. Lewis.

I examined 255 *A. c. huico* from the California Academy of Sciences (CAS) collection obtained by A. E. Greer and F. W. Burley in two Mexican states: Sinaloa, 45.4 km E of jct Hwy 40 (Durango Road) and Hwy 15 on Hwy 40 [10 June 1963 (CAS 114865–114876, N = 12); Hwy 40, 17.0 km E of jct Hwy 15 [30 June 1963 (CAS 115230–115256, N = 27)]; 9.9 km E of jct Hwys 15 and 40 on Hwy 40 [30 June 1963 (CAS 115210–115221, N = 12)]; Hwy 40 at 39.7 km E of jct Hwy 15 [10 June 1963 (CAS 114877–114833, N = 7)]; 5.3 km W of Hwy 15 on dirt road 13.3 km N of Escuinapa [3 July 1963 (CAS 115295–115301, N = 7)]; 11.1 km E of jct 15 and Hwy 40 on Hwy 40 [14 June 1963 (CAS 114961–114991, N = 31)]; 25.9 km S of Escuinapa on Hwy 15 [2 July 1963 (CAS 115269–115287, N = 19)]; Nayarit, Arroyo de Rifilion, 9.0 km N of Compostela [13 May 1963 (CAS 114391–114416, N = 26)]; Laguna Santa Maria Road, 6.6 km NE of jct Hwy 15 and Laguna Santa Maria Road [28 May 1963 (CAS 114664–14669, N = 6)]; 1.3 km N of Navarette on Sauta Road [27 June 1963 (CAS 115154–115174, N = 21)]; 32.3 km S of Río Acoponeta on Hwy 15 [23 June 1963 (CAS 115036–115071, N = 36)]; 1.8 km S of Río Canas on Hwy 15 [2 July 1963 (CAS 115262–115267, N = 6); 23.0 km W of jct Mexcaltitan and Hwy 15 on Mexcaltitan Road [28 June 1963 (CAS 115194–115199, N = 6)]; end of Santiago Ixcuintla Road, 45.6 km W of jct of Santiago Road and Hwy 15 [7 July 1964 (CAS 95372–95389, N = 18)]; Laguna Santa Maria [26 June 1964 (CAS 95326–95346, N = 21). I obtained size (as SVL to the nearest 1 mm) for all except 41 males in the CAS sample. Females were dissected to identify the number and measure oviductals eggs and ovarian follicles.

The ca. 3:1 sex ratio (187 males:68 females) in *A. c. huico* was on the order of the ca. 2:1 (129 males:64 females) observed in *A. c. nigrigularis* (Walker 2008. Herpetol. Rev., *in press*). Only 10 (21.7%, 6 measurable clutches) of 46 gravid females contained oviductal eggs; all others had yolked ovarian follicles of 3.5–10.0 mm in diameter. Bias in the sex ratio of both subspecies may pertain to secretive female behavior during egg maturation and oviposition that make them less susceptible to collection than other adults. Alternatively, shorter life expectancy in females is a non-mutually exclusive explanation that may also contribute to the male-biased sex ratio. The latter may be reflected by the relative male-female numbers, respectively (in parens), by sizes (as 10-mm SVL increments where applicable) in the CAS sample: 50–59 mm (2:0); 60–69 mm (9:4); 70–79 mm (23:23); 80–89 mm (37:24); 90–99 mm (32:13); 100–109 mm (4:4); 110–118 mm (39:0). Assuming any secretive behavior by adult females is unrelated to their size, *A. c. huico* appears strongly sexually dimorphic in favor of males.

Females of *A. c. huico* appear to attain reproductive maturity at a small size (< 70 mm SVL) similar to *A. c. barrancarum* in southwestern Chihuahua (Walker et al. 2004. Herpetol. Rev. 34:366) and *A. c. nigrigularis* in Sinaloa (Walker 2008, Herpetol. Rev., *in press*). In *A. c. huico*, 2 females 67–69 mm SVL had clutches of 2–3 (mean = 2.5). Among females 70–79 mm SVL, 13 (56.5%) were gravid; clutch size was 2–5 (mean = 3.2, N = 13). Among females 80–89 mm SVL, 17 (70.8%) were gravid; clutch size was 2–6 (mean = 4.4, N = 17). Among females 90–99 mm SVL, 12 (92.3%) were gravid; clutch size was 3–8 (mean = 5.6, N = 12). Among females 100–105 mm SVL 2 (50%) were gravid; clutch size was 5–7 (mean = 6.0, N = 2). For the period 13 May–7 July 1964, 46 females with data had clutches (sample size in each category in parens) of 2 (4), 3 (11), 4 (13), 5 (6), 6 (7), 7 (4), and 8 (1) eggs; mean clutch size and SVL for these females were 4.4 ± 0.23 (2-8) eggs and 83.5 ± 1.27 (67–105) mm, respectively. A moderately strong relationship exists between clutch size and SVL (adjusted $r^2 = 0.50$). Data for the CAS sample established these additional reproductive characteristics for *A. c. huico*: SVL of all adult males, 92.7 ± 1.21 (67–118, N = 131) mm; SVL of all adult females, 83.6 ± 1.42 (62–105, N = 66); oviductal egg dimensions in 6 (60%) females with oviductal eggs, 8.2 ± 0.44 × 14.4 ± 0.45 (7.5–10.0 × 13–16) mm; reproductive season, minimally May–July (this study) into late August (Zweifel, *op. cit.*); and reproductive potential, multiple clutches based on maturation of both sexes at small body sizes within the long activity cycle near the northern fringe of the tropics in Sinaloa, Nayarit, and Jalisco. Collecting data for the CAS sample also revealed that *A. c. huico* achieves high population densities at numerous mainland sites and on Isla Isabel (based on CAS 59136–59295, N = 160) within an area occupied by a congener (*A. lineatissima duodecemlineata*; Zweifel, *op. cit.*) only at San Blas, Nayarit.

I thank Robert C. Drewes (CAS) for the opportunity to examine specimens in his care.

Submitted by **JAMES M. WALKER**, Department of Biological Sciences, University of Arkansas, Fayetteville, Arkansas 72701, USA; e-mail: jmwalker@uark.edu.

ASPIDOSCELIS COSTATA NIGRIGULARIS (Sinaloa Black-throated Whiptail Lizard). **REPRODUCTION**. *Aspidoscelis costata nigrigularis* (*sensu* Reeder et al. 2002. Am. Mus. Novit. 3365:1–61) is among many named, but biologically poorly known reptiles from remote areas of México. Zweifel (1959. Bull. Am. Mus. Nat. Hist. 117:57–116) described the subspecies based on 47 specimens from the type locality (16.8 km NW of Culiacán) and other sites in western Sinaloa. Here, I provide reproductive data (clutch ranges and ± SE; SVL ranges and ± SE) for a subspecies which Zweifel (1959, *op. cit.*) noted only minimum size at maturity for females (62 mm SVL), maximum sizes for females (84 mm SVL) and males (114 mm SVL), and reproductive season between 18 July and August 20 (based on 3 gravid females).

I examined 196 *A. c. nigrigularis* from the California Academy of Sciences (CAS) collection obtained by A. E. Greer and F. W. Burley at 12 sites in Sinaloa over 13 days in 1963: 1.9 km E of La Cruz [6 July (CAS 115304–115313, N = 10)]; 8.5 km E of La Cruz [6 July (CAS 115314–115327, N = 14)]; 21.4 km S of San Lorenzo, Hwy 15 [10 July (CAS 115365–115384, N = 20)]; 14.4 km N of San Lorenzo, Hwy 15 [11 July (CAS 115385–115410, N = 26)]; 12.5 km S of Río San Lorenzo, Hwy 15 [11 July (CAS 115417–115435, N = 19)]; 12.8 km S of Culiacán, Hwy 15 [12 July (CAS 115440–115456, N = 17)]; 1.9 km N of El Dorado on El Dorado Road [13 July (CAS 115460–115482, N = 23)]; 3.5 km W of jct of road 4.5 km S of Navolato [14 July 1963 (CAS 115509–115523, N = 15)]; 22.9 km S of jct of El Dorado and Navolato Roads [14 July (CAS 115524, N = 1)]; 33.6 km N of Río Culiacán, Hwy 15 [17 July (CAS 115528–115546, N = 19)]; 11.4 km N of Río Evora Nocorito, Hwy 15 [18 July (CAS 115584–115596, N = 13)]; 85.8 km N of Río Culiacán, Hwy 15 [18 July (CAS 115611–115629, N = 19)]. I obtained size (as SVL to the nearest 1 mm) and gender on all except 3 animals. Females were dissected to identify the number and measure oviductal eggs and ovarian follicles; 5 females were too damaged internally to permit this assessment.

The ca. 2:1 sex ratio (129 males:64 females) in *A. c. nigrigularis* was on the order of the ca. 3:1 (187 males:68 females) observed in *A. c. huico* (Walker 2008. Herpetol. Rev., *in press*). Only 9 (20.5%) of 44 undamaged gravid females of *A. c. nigrigularis* contained oviductal eggs; all others had yolked ovarian follicles 3.5–9.5 mm in diameter. Bias in the sex ratio in both subspecies may pertain to secretive female behavior during egg maturation and oviposition that make them less susceptible to collection by humans than other adults in the population. Alternatively, shorter life expectancy in females is a non-mutually exclusive explanation that may also contribute to the male-biased sex ratio. The latter may be reflected by the relative male-female numbers, respectively (in parens), by sizes (as 10 mm SVL increments where applicable) in the CAS sample: 50–59 mm (3:2); 60–69 mm (15:12); 70–79 mm (25:13); 80–89 mm (18:20); 90–99 mm (20:14); 100–109 mm (26:3); 110–118 mm (22:0). Assuming any secretive behavior by adult females is unrelated to their size, *A. c. nigrigularis* appears strongly sexually dimorphic in favor of of males.

Females of *A. c. nigrigularis* appear to attain reproductive maturity at a small body size similar to *A. c. barrancarum* in southwestern Chihuahua (Walker et al. 2003. Herpetol. Rev. 34:366). In *A. c. nigrigularis*, 5 females 62–68 mm SVL had clutches of 1–2 (mean = 1.8) eggs. Among females 70–79 mm SVL, 11 (85%) were gravid; clutch size was 2–4 (mean = 2.9, N = 10). Among females of 80–89 mm SVL, 18 (95%) were gravid; clutch size was 3–7 (mean = 4.8, N = 14). Among females 90 mm SVL, all were gravid; clutch size of females 90–99 mm SVL was 4–7 (mean = 5.8, N = 12), but was 6–7 (mean = 6.7, N = 3) for females 100–105 mm SVL. For the period 6–18 July 1963, 44 females with data had clutches (sample size in each category in parens) of 1 (1), 2 (8), 3 (4), 4 (10), 5 (6), 6 (10), and 7 (5) eggs; mean clutch size and SVL for these females were 4.4 ± 0.26 (1–7) eggs and 83.6 ± 1.63 (62–105) mm, respectively. Clutch size was strongly correlated with SVL (adjusted $r^2 = 0.71$). Data for the CAS sample established these additional reproductive characteristics: SVL of all adult males, 90.8 ± 1.45 mm (range: 62–118, N = 123) mm; SVL of all adult females, 83.6 ± 1.42 mm (range: 62–105, N = 54) mm; oviductal egg dimensions in 9 females with oviductal eggs, 8.0 ± 0.17 × 14.5 ± 0.44 (7.5–9.0 × 13.0-17.0) mm; reproductive season, minimally July (this study)–August (Zweifel 1959, *op. cit.*) and probably much longer; and reproductive potential, multiple clutches based on maturation of both sexes at a small body size within the long activity cycle in subtropical regions of Sinaloa.

Collecting data for the CAS sample further indicated that *A. c. nigrigularis* achieves high population densities at numerous sites within a distribution area that is inhabited by no other species in the genus *Aspidoscelis.*

I thank Robert C. Drewes (CAS) for the opportunity to examine specimens in his care.

Submitted by **JAMES M. WALKER**, Department of Biological Sciences, University of Arkansas, Fayetteville, Arkansas 72701, USA; e-mail: jmwalker@uark.edu.

ASPIDOSCELIS LAREDOENSIS (Laredo Striped Whiptail) × ***ASPIDOSCELIS GULARIS GULARIS*** (Texas Spotted Whiptail). **REPRODUCTIVE POTENTIAL**. Clonal complex A (Walker 1987a. Texas J. Sci. 39:313–334) of diploid (Bickham et al. 1976. Herpetologica 32:395–399) parthenogenetic *Aspidoscelis laredoensis* (nomenclature based on Reeder et al. 2002. Am. Mus. Novit. 3365:1–61), derived from a single hybrid (Abuhteba et al. 2000. Can. J. Zool. 78:895–904) of *A. gularis gularis* × *A. sexlineata* (McKinney et al. 1973. Herpetologica 29:361–366), is syntopic with *A. gularis gularis* at Los Ebanos (between the town and the Rio Grande), Hidalgo County, Texas (26.2341667°N, 98.5605556°W, datum WGS84; elev. 26 m; Walker 1987b. Am. Midl. Nat. 117:319–332). On 25 May 1986, JMW, JEC, and MAP collected 3 male lizards (University of Arkansas Department of Zoology, UADZ 1948–1950) at this site characterized by the basic striped and spotted adult dorsal pattern of all-female *A. laredoensis* and a ventral pattern intermediate between this species and *A. gularis gularis*. Each of these males is a triploid hybrid resulting from a haploid sperm of *A. gularis gularis* (1n = 23) fertilizing the unreduced egg of *A. laredoensis* (2n = 46). *Aspidoscelis laredoensis* A has been identified as a participant in numerous other hybridizations. Walker et al. (1989a. J. Herpetol. 23:119–130) listed 23 hybrids (including UADZ 1948–1950) from 8 sites in Texas and 3 in México and Walker et al. (1989b. Copeia 1989:1059–1064) described the triploid karyotype of a male *A. laredoensis* × *A. gularis gularis*. Here, we detail the morphological and histological examination of a male hybrid (UADZ 1948) from Los Ebanos.

Based on Walker et al. (1989b, *op. cit.*), Taylor et al. (2001. Am. Mus. Novit. 3345:1–65), and Walker et al. (2006. Herpetol. Rev. 37:344–345), JMW described the gross reproductive morphology of the preserved hybrid. Based on Goldberg and Beaman (2003. Herpetol. Rev. 34:143), SRG histologically examined the left testis and left epididymis of the hybrid and assessed its previous reproductive functionality.

The preserved hybrid was 62 mm SVL (mass unavailable). Compared to male *A. gularis gularis* of similar size from Los Ebanos, its testes (mean dimensions of both = ca. 4 × 6 mm), epididymides, and hemipenes were of expected size and structure. Histology of the left testis indicated that the hybrid was undergoing early spermiogenesis. As reported for other teiid lizards (Lowe and Goldberg 1966. J. Morphol. 119:277–281), a circumtesticular tunic 1–3 Leydig cells thick surrounded the left testis. Clusters of metamorphosing spermatids were present and tails of spermatozoa projected into the lumina of the seminiferous tubules. Sperm were also present in the sectioned epididymis.

This *A. laredoensis* × *A. gularis gularis* hybrid would have been deemed fertile based on cytological characteristic of testes in species such as *A. tigris* (Lowe and Goldberg 1966, *op. cit.*). However, though it seemed capable of producing sperm and inseminating females of *A. laredoensis* and *A. gularis gularis* at Los Ebanos, its ability to produce a stable haploid chromosome complement from triploid number of 69 is dubious. Nevertheless, the hybrid likely would have interfered with normal reproduction in some females of these two species should any of them have been inseminated. Taylor et al. (2001, *op. cit.*) provided indirect histological evidence that the more numerous triploid hybrid males of *A. tesselata* × *A. marmorata* (N = 12) collected at Arroyo del Macho, Chaves County, New Mexico, may have inseminated congeneric females at this site.

Collection of UADZ 1948–1950 was made under authority of Permit No. 61 issued to JEC in 1986 by the Texas Parks and Wildlife Department. Field studies at the Los Ebanos site in the Lower Rio Grande Valley National Wildlife Refuge, under the protectorate of the U.S. Fish and Wildlife Service, were conducted under Permit 21552-07-09-ms granted to JEC.

Submitted by **JAMES M. WALKER**, Department of Biological Sciences, University of Arkansas, Fayetteville, Arkansas 72701, USA (e-mail: jmwalker@uark.edu); **STEPHEN R. GOLDBERG**, Department of Biology, Whittier College, Whittier, California 90608, USA (e-mail: sgoldberg@whittier.edu); **JAMES E. CORDES**, Division of Sciences. Louisiana State University at Eunice, Louisiana 70535, USA (e-mail: jcordes@lsue.edu); and **MARK A. PAULISSEN**, Department of Natural Sciences, Northeastern State University, Tahlequah, Oklahoma 74464, USA (e-mail: paulisse@nsuok.edu).

CHAMELEO RUDIS (Ruwenzori Side-striped Chameleon). **REPRODUCTION.** Few data exist on clutch size and sex ratio, and neonate size for most viviparous chameleons. Recently, many species have been described from the tropical and montane forests of East Africa (Neças et al. 2003. Trop. Zool. 16:1–12; Menegon et al. 2002. J. Herpetol. 36:51–57; Largen 1995. Trop. Zool. 8:333–339; Tilbury 1991. Trop. Zool. 4:159-165; Tilbury 1992. Trop. Zool. 5:1–9; Klaver and Bohme 1988. Bonn. Zool. Beitr. 39:381–393), and additional cryptic or semi-cryptic species have been distinguished from previously described species (Neças 1994. Herpetozoa 7[3/4]:95–108). Thus, even existing data addressing clutch characteristics may be questionable unless linked to voucher specimens that confirm species identity.

In mid-January 2007, a recently deceased gravid female chameleon, included in a shipment form Tanzania, was received from an animal dealer in Calgary. PADW identified it as *Chamaeleo rudis* (Spawls et al. 2002. A Field Guide to the Reptiles of East Africa. Academic Press, San Diego, California. 543 pp.) based on the enlarged granules on the median line of the throat extending onto the belly; no cranial horns or sail-like dorsal crest; heterogeneous body scalation with scattered large tubercles; head less than twice as long as broad; nostrils laterally directed; and body squat. Snout–vent length (SVL) and tail length were measured to the nearest 1 mm, head length (HL) and head width (HW) were measured using a Mitutoyo Digital Caliper Model 500-322 to the near-

TABLE 1. Data on 12 *Chameleo rudis* embryos; mass in grams, all other measurements in millimeters.

	Individuals	SVL	Tail	HL	HW	Mass
Females	1	30.7	24.8	9.9	6.2	0.61
	2	26.7	21.0	10.0	5.9	0.53
	3	24.7	23.0	9.6	5.8	0.47
	4	26.0	23.8	9.5	5.8	0.50
	5	26.3	24.3	9.8	5.8	0.48
	6	27.5	23.5	9.9	5.9	0.51
	mean	27.0	23.4	9.8	5.9	0.52
Males	1	26.7	21.0	10.2	5.9	0.57
	2	27.2	22.7	9.9	5.9	0.51
	3	27.0	22.3	10.1	6.2	0.46
	4	26.3	22.3	9.6	5.9	0.51
	5	27.7	22.0	10.0	6.0	0.49
	6	27.5	20.7	9.7	6.0	0.49
	mean	27.1	21.8	9.9	6.0	0.51

est 0.1 mm; each measurement was taken three times, then averaged. Measurements are: 81 mm SVL, 82 mm tail, 25.7 mm HL, 14.8 mm HW.

Twelve near full-term embryos were found *in utero,* six in each oviduct. The sex ratio was six males, six females (4.2 in one oviduct and 2.4 in the other). Gender was determined either by direct observation of at least one everted hemipenes in males (three specimens) or by observation of two circular openings into the retracted hemipenes at the lateral corners of the vent. Accuracy of the latter approach was verified by making a medial incision on the ventral side of the tail immediately posterior to the vent in two suspected males and two suspected females. Presence of a cone-shaped connective tissue sheath overlying the conical muscle, transversus penis, which surrounds the hemipene, confirmed male gender. These structures were absent in the suspected females. All embryos were measured and weighed (Table 1).

Female embryos had longer tails than male embryos (Mann Whitney U test: $P = 0.03$) but it is unknown whether this sexual dimorphism carries into adulthood with this species, however, in dwarf chameleons some ecomorphological sex-specific variation has been documented (Stuart-Fox and Moussalli 2007. J. Evol. Biol. 20:1073–1081). SVL, HL, HW, mass, ratio of tail length to SVL, and ratio of HW to HL did not differ between genders (Mann-Whitney U test: $P > 0.11$ for these 6 comparisons). However, variation in SVL for female embryos was significantly greater than variation in SVL for male embryos (Variance ratio test: F = 15.4, $P < 0.01$).

The adult female (L07.15.1) and male embryo 1 (L07.15.2; see Table 1) were deposited in the Royal Alberta Museum, Edmonton, Alberta, Canada.

Submitted by **PATRICK A. D. WISE**, Vertebrate Morphology and Palaeontology Research Group, Department of Biological Sciences, University of Calgary, Calgary, Alberta T2N 1N4, Canada (e-mail: padwise@ucalgary.ca); and **ANTHONY P. RUSSELL**, Vertebrate Morphology and Palaeontology Research Group, Department of Biological Sciences, University of Calgary, Calgary, Alberta T2N 1N4, Canada (e-mail: aprussell@ucalgary.ca).

CROTAPHYTUS COLLARIS (Eastern Collared Lizard). **HOMING.** Previously, I reported an instance of homing by a male collared lizard that became separated from his territory by the outflow of the Arcadia Lake Reservoir (Baird 2004. Herpetol. Rev. 35:167). In that instance, possible routes for homing to its territory were either to travel an indirect and lengthy (> 800 m) overland route through the territories of several other *C. collaris* males, or to swim across the outflow channel (9.5 m). On 20 June 2006, I observed another territorial *C. collaris* male, a paint-marked subject in my space-use studies, attempting to swim across the channel. This male's territory was on the north side of the channel, but because no water was released during most of May and June 2006, he made occasionally forays into the dry channel and onto its south side. The outflow was opened at 0530 h on 19 June and remained open for three days. From 1000 to 1020 h on 20 June, I made a focal observation of this male, then on the south side of the now flowing channel that separated him from his north-side territory. Throughout the entire 20-min observation, this male moved back and forth along the bank while peering across to the north side. Thirty min after the end of the focal observation, I sighted this male swimming in the midst of the outflow, presumably in an attempt to return to his territory. I followed him for 120 m as the current carried him downstream. When it became apparent that he would not reach the rocks on the other side before being swept over the end of the spillway, I waded into the water, caught the lizard, and returned him to his territory. Upon capture, the male's lungs were completely inflated, allowing floatation. This observation implies that my 2002 report of homing also involved swimming across the flowing channel, and that the drive to home is strong enough in territorial *C. collaris* males that fast-flowing water does not appear to be a deterrent. The spillway at the Arcadia Lake Reservoir is human constructed, but *C. collaris* may face similar flowing aquatic barriers during flash flooding events.

This study was conducted under Oklahoma Department Wildlife Conservation permit 4097 to Troy A. Baird.

Submitted by **TROY A. BAIRD**, Department of Biology, University of Central Oklahoma, Edmond, Oklahoma, 73034-5209, USA; e-mail: tbaird@ucok.edu.

HOPLOCERCUS SPINOSUS (NCN). **ENDOPARASITES.** *Hoplocercus spinosus* is a poorly known lizard distributed from Mato Grosso and Mato Grosso do Sul States, Brazil to adjacent Bolivia (Wiens and Etheridge 2003. Herpetologica 59:375–398). To our knowledge, no parasite records were published for *H. spinosus*. Hence, we report the nematode, *Cruzia rudolphii,* infecting *H. spinosus*.

Five adult *H. spinosus* (mean = 80.4 mm SVL, SD = 12.3 mm) deposited in the Coleção Zoológica de Referência, Laboratory of Zoology, *Campus* of Corumbá, Universidade Federal de Mato Grosso do Sul (UFMS; CEUCH 2888, 2892, 3374, 3375, and 3460) were examined for endoparasites. Lizards were captured in November 2003 and February 2004 in pitfall traps with drift fences in a remnant patch of Cerradão (savanna-like vegetation) of Dois Irmãos do Buriti and Terenos (55.30°W, 20.50°S, datum: WGS84) between the municipality of Aquidauna and Campo Grande, Mato Grosso do Sul State, Brazil. Endoparasites were cleared in phenol, identified and deposited in the Coleção Helmintológica do Instituto de Biociências da Unesp de Botucatu as *Cruzia rudolphii* (CHIBB 1052). Three adult females of *C. rudolphii* were recovered and the observed prevalence (infected lizards / examined lizards × 100) was 20%.

Species of *Cruzia* have been reported in mammals (Quintão e Silva and Costa 1999. J. Wildl. Dis. 35:371–374) and reptiles (Vicente et al. 1993. Rev. Bras. Zool. 10:1–183). *Cruzia rudolphii* was described from the colubrid snake *Erythrolamprus aesculapii* in southeastern Brazil (Ruiz [1947] in Vicente et al. 1993, *op. cit.*). *Hoplocercus spinosus* represents a new host record for *C. rudolphii* and Mato Grosso do Sul, Brazil is a new locality for *C. rudolphii*.

We thank Marc P. Hayes for comments in the manuscript. This study had financial support by FAPESP (processes 04/03628-1; 06/59692-5) and RWA thanks CAPES for a grant.

Submitted by **ROBSON W. ÁVILA,** Programa de Pós-Graduação em Biologia Geral e Aplicada, Departamento de Parasitologia, Instituto de Biociências, UNESP, Distrito de Rubião Jr., s/nº, CEP 18618-000, Botucatu, SP, Brazil (e-mail: robsonavila@gmail.com); **NILTON C. CACERES,** Universidade Federal de Santa Maria, Centro de Ciência Naturais e Exatas, Departamento de Biologia; **VANDA L. FERREIRA**, Departamento de Ciências do Ambiente, Laboratório de Zoologia, *Campus* de Corumbá, Universidade Federal de Mato Grosso do Sul. Av. Rio Branco, 1270. Caixa Postal 252, CEP 79301-970, Corumbá, MS, Brazil; and **REINALDO JOSÉ DA SILVA,** Departamento de Parasitologia, Instituto de Biociências, UNESP, Distrito de Rubião Jr., s/nº, CEP 18618-000, Botucatu, SP, Brazil.

PHRYNOSOMA CORNUTUM (Texas Horned Lizard). **MORTALITY.** Literature on horned lizards (*Phrynosoma*) reveals that horn length is an anti-predator device whereby individuals with larger horns are less susceptible to mortality from predation (Young et al. 2004. Science 304:65). Behaviors where horned lizards thrust their heads back to inflict puncture wounds into would-be predators support this idea (Brodie et al. 2004. Science 306:230b; Sherbrooke 2003. Introduction to Horned Lizards of North America. University of California Press, Berkeley. 177 pp.). However, elongated cranial horns have not been implicated as contributing to mortality. We report mortality of *Phrynosoma cornutum* resulting from entrapment of the horns in a hardware cloth fence.

FIG. 1. Texas Horned Lizard, *Phrynosoma cornutum*, entrapped in overlapping sheets of hardware cloth used in a fence in Cochise County, Arizona.

On 15 April 2007, a young adult female *P. cornutum* (ca. 77 mm SVL) was found dead in a hardware cloth fence in the San Simon Valley, Cochise County, Arizona (31.93°N, 109.08°W; elev. 1330 m). The lizard was found entrapped in the area of overlap where two sections of 6.4-mm mesh wire hardware cloth had been joined to form a single fence (Fig. 1). The 60-cm tall fence was riveted at the top through a 15-cm tall strip of sheet metal, and at the ground the fence was buried in the soil. The lizard was aligned nearly vertically about midway up the 45 cm of exposed fence, ca. 10 cm from the edge of the overlap, where a small gap between the sheets of hardware cloth likely allowed entry. The lizard's occipital horns were protruding from and enmeshed in the hardware cloth. The body was completely desiccated but showed no other decay and was not noticeably faded. The only damage visible was two holes ca. 2 mm in diameter bored dorsally though the midway of tail and ventrally just caudal to the cloaca, likely the result of scavenging insects.

We suspect that the lizard was unable to escape entrapment of its horns in the mesh and later died. Entrapment or entanglement in anthropogenic materials has been recorded for many animal species, in some cases in surprising numbers (e.g., Barton and Kincead 2005. J. Soil Water Cons. 60:33A–35A; Loegering 1997. Wilson Bull. 109:353–355), though the cause of mortality we report is probably rare. Because of the potential for entrapment, however, we suggest caution in the use of hardware cloth in areas where horned lizards of conservation concern occur, such as in the range of the Flat-tailed Horned Lizard, *P. mcallii*. Thoroughly securing the overlapping ends of fencing would likely alleviate the potential for such mortality.

Submitted by **MEGAN E. LAHTI**, Department of Biology, 5305 Old Main Hill, Utah State University, Logan, Utah 84321-5305, USA (e-mail: phrynosoma@biology.usu.edu); and **RYAN P. O'DONNELL**, Department of Biology and the Ecology Center, 5305 Old Main Hill, Utah State University, Logan, Utah 84321-5305, USA (e-mail: Ryan@biology.usu.edu).

PHRYNOSOMA CORNUTUM (Texas Horned Lizard). **PREDATION.** *Phrynosoma cornutum* has a broad distribution in North America (Price 1990. Cat. Amer. Amphib. Rept. 469.1–7; Sherbrooke 2003. Introduction to Horned Lizards. University of California Press, Berkeley 178 pp.). Knowlton (1942. Copeia 1942:186), Miller (1948. Copeia 1948:67), and Sherbrooke (2003, *op. cit.*) indicate that the vertebrate predators of *P. cornutum* include the Greater Roadrunner (*Geococcx californianus)*, Red-shouldered Hawk (*Buteo lineatus*), Chihuahuan Raven (*Corvus cryptoleucus),* and the Western Diamond-backed Rattlesnake (*Crotalus atrox*). Prey decapitation, a important feeding tactic used by some raptorial birds to minimize risk from dangerous prey, has not been reported to be used on horned lizards. Hence, we report an observation of prey decapitation by Swainson's Hawk (*Buteo swainsoni*) on *P. cornutum.*

Our observations were made between 15 June and 15 August 2006 during research on the reproductive biology of *B. swainsoni* in the Reserva de la Biosfera de Janos (31.1854528°N, 108.9469917°W and 31.1909583°N, 108.9394694°W, datum: NAD27; elev. 1350–1400 m), in Janos, Chihuahua, México. Our study was conducted in open desert scrubs dominated by Honey Mesquite (*Prosopis glandulosa*), Viscid Acacia (*Acacia neovernicosa or A.constricta*), and Catclaw Mimosa (*Mimosa biunicifera*). We analyzed the regurgitated pellets and prey remains of 19 occupied *B. swainsoni* nests with young, all of which were in *P. glandulosa* trees. We found the remains of *P. cornutum* in 8 nests, all of which were heads. To estimate body size, we recorded the mean distance between the distal ends of temporal (squamosal bone) horns. For the 14 *P. cornutum* heads collected, temporal horn distance (D_{th}) ranged from 1.72 mm to 2.14 mm. Based on a preliminary linear SVL–temporal horn distance regression (SVL $= 1.978 + 3.244[D_{th}]$) constructed from 34 *P. cornutum* in the herpetological collection of the Universidad Autónoma de Nuevo León, the heads all belonged to adults estimated to range in size from 75 mm to 89 mm. No other remains from these lizards were found in the nests, suggesting that the lizards were either dismembered and fed to juveniles or consumed by adult hawks.

Buteo swainsoni, with a broad distribution throughout much of North and South America (Clements 2000. Birds of the World: A Checklist. Ibis Publishing Co., Vista, California. 867 pp.), has a diet that includes horned lizards (Bednarz 1988. The Condor 90:311–323). Bednarz (*op. cit.*) notes that based on biomass, *B. swainsoni* diet included 6.2% *P. texanus (= cornutum*). Our observations imply that *B. swainsoni* may decapitate *P. cornutum* prior to eating them.

Submitted by **DAVID LAZCANO, BLANCA IDALIA GONZALEZ.-.GARZA NERYEDA CRUZ.-.MALDONADO,** and **JOSE IGNACIO GONZALEZ.-.ROJAS,** Universidad Autónoma de Nuevo León, Facultad de Ciencias Biológicas, Laboratorio de Herpetología, Apartado, Postal - 513, San Nicolás de los Garza, Nuevo León, C.P. 66450, México (e-mail [DL]: dvlazcano@hotmail.com).

PHRYNOSOMA MCALLII (Flat-tailed Horned Lizard). **RAIN HARVESTING**. Three species of *Phrynosoma* (*P. cornutum, P. modestum*, and *P. platyrhinos*) exhibit a behavior termed rain harvesting (Sherbrooke 2003. Introduction to Horned Lizards of North America. University of California Press, Berkeley, California. 178 pp.). In this stylized behavior, occurring during infrequent desert rains, a lizard elevates its venter from the substrate and positions its dorsal surface so that it intercepts rain which then moves by capillary action and bulk flow to the head and mouth where it is imbibed. We tested for this behavior in a fourth species of horned lizard, *P. mcallii,* because it lives in the most arid habitat of any horned lizard. Moreover, *P. mcallii* has never been observed to drink standing water in captivity and is assumed to procure moisture from its food (Mayhew 1968. *In* G. W. Brown, Jr. [ed.], Desert Biology: Special Topics on the Physical and Biological Aspects of Arid Regions, Volume 1, pp. 195–356. Academic Press, New York). Here, we provide evidence that *P. mcallii* engages in rain harvesting.

We equipped 64 lizards with radio-telemeters in the East Mesa area of the Imperial Valley (32.8222222°N, 115.2527778°W, datum: NAD83; elev. 10–15 m), California during the summer and fall of 2004. This area is in the Colorado Desert subsection of the Sonoran Desert. Large Creosote (*Larrea tridentata*) and sandy substrate characterizes the habitat. To test whether *P. mcallii* exhibits the stylized rain-harvesting posture, we opportunistically sprayed 5 relocated lizards with a heavy mist from a spray bottle. Two of these lizards exhibited behaviors consistent with rain harvesting. One lizard, from which we had just removed the transmitter harness, moved under a small, leafless bush where we sprayed it with water. This lizard promptly elevated its venter from the substrate, angled its dorsum to slope downward toward its head, and expanded its dorsum. Water sprayed on the lizard flowed toward the head and rhythmic swallowing motions followed. The second responsive lizard exhibited a less prompt and stylized response. This lizard walked under a small, leafless shrub that did little to obstruct our view or the sprayed water. When the lizard was sprayed, it remained off the substrate with its venter elevated and expanded the dorsum slightly. This lizard was standing on a slope which effectively angled its dorsum downward. Water collected on its dorsum and collected near the mouth, where it was swallowed. The transmitter was left on this individual. These observations parallel those described for the three species of *Phrynosoma* known to harvest rain; namely, venter elevated from the substrate, dorsum spread, and dorsum sloped toward head.

The Bureau of Land Management (particularly Daniel Steward, Lynette Elser, and Chris Knauf at the El Centro, California, Field Office), Department of the Navy, Bureau of Reclamation, and U.S. Fish and Wildlife Service provided support for this project. The California Department of Fish and Game approved a scientific collecting permit No. SC-007201. Erin Dreyfuss, Christi Duncan, Brent Eastty, Angela Hyder, Darren Kelly, Justin Nobel, and Amanda Trahan assisted with field work.

Submitted by **TYLER J. GRANT** (e-mail: tgrant7@cnr.colostate.edu) and **PAUL F. DOHERTY**, Department of Fishery and Wildlife Biology, Colorado State University, Fort Collins, Colorado 80523, USA.

PHRYNOSOMA SOLARE (Regal Horned Lizard). ***CROTALUS* ENVENOMATION**. On 14 September 2006, we studied the behavioral responses of an adult male *Phrynosoma solare* (93 mm SVL, 50.5 g) to a venomous ophidian predator, the Western Diamondback Rattlesnake (*Crotalus atrox*; Vorhies 1948. Copeia 1948:302–303). The capture and experimental trial site of the *P. solare* was near a small volcanic hill in Altar Valley, Pima Co., Arizona, USA (32.0365278°N, 111.3962778°W, datum: WGS84; elev. 822 m). To conduct these trials, we released the lizard at its capture site after 5 days in captivity. We had collected the *C. atrox* (82 cm SVL, 8 cm tail, 385 g) in Avra Valley west of the Tucson Mountains, Pima Co. Tucson, Arizona (32.1859722°N, 111.0996944°W; elev. 792 m). For the trials, we placed the snake in a 46-cm long, 3.5-cm diameter clear plastic tube. The snake's head and forebody extended 15-cm from one end of the tube, while the tail extended from the opposite end. The experimenter held the posterior end of the tubed snake during presentations of its foreparts to the lizard. During trial encounters with this lizard, the lizard was unexpectedly struck by the snake on the third presentation of the snake. Before envenomation, the lizard had failed to exhibit antipredator escape-running or body-flip behaviors, but did tilt its body laterally toward the snake, raise its horns, and was contacted by the snake's head in all three encounters (Sherbrooke and May, unpubl. data). Though episodes of rattlesnake predation on horned lizards, including full and partial ingestions (successful or unsuccessful), have been reported (Beavers 1976. Southwest. Nat. 20:503–515; Funk 1965. Herpetologica 21:15–17; Sherbrooke 2003. Introduction to Horned Lizards of North America, University of California Press, Berkeley. 178 pp.), no accounts exist of the effects of *Crotalus* envenomation on horned lizards.

At 0928 h MST, a bite strike resulted in one fang penetrating the lizard's dorsal skin 11 mm anterior to the left hind leg and 8 mm above the lateral fringe scales. A small drop of blood immediately oozed from the wound, but did not spread. By 0934 h, a clear fluid (ca. 0.5 ml) seeped from the wound onto the experimenter's hand, and the lizard seemed to be experiencing difficulty moving its limbs. By 0950 h, the lizard was lethargic, with continuing fluid loss and a lightening of its body color; and by 1004 h, a pinkish fluid (presumably blood and lymph) was exuding from both nostrils. The lizard now appeared dead and was unresponsive to handling. Its head remained in a strong horns-raised posture (90° angle; Sherbrooke 1987. Southwest. Nat. 32:512–515).

The lizard was frozen and later formalin/ethanol preserved and is deposited in the Herpetology Collection of the University of Arizona, Tucson (UAZ 56621). Scientific collecting permit (SP651312) to WCS was provided by the Arizona Department of Game and Fish.

Submitted by **WADE C. SHERBROOKE**, Southwestern Research Station, American Museum of Natural History, Portal, Arizona 85632, USA (e-mail: wcs@amnh.org); and **CLAYTON J. MAY,** 5814 S. Hopdown Lane, Tucson, Arizona 85746, USA.

PHYMATURUS ANTOFAGASTENSIS (NCN). **DIET**. *Phymaturus antofagastensis* is a viviparous lizard inhabiting the high plains (altiplano) of the Andean Mountains in central and northern Argentina (Cabrera and Willink 1980. Biogeografía de América Latina. Washington, DC. 109 pp.). This species is sexually dimorphic and females exhibit biennial reproduction (Boretto and Ibargüengoytía 2006. Amphibia-Reptilia 27:25–36). Its conservation status is ambiguous as it is defined as a species for which "insufficient knowledge" exists (Lavilla et al. 2000. Categorización de los Anfibios y Reptiles de la República Argentina. Asoc. Herpetol. Arg., San Miguel de Tucumán, Argentina, 97 pp.). The genus *Phymaturus* is considered herbivorous (Espinoza et al. 2004. Proc. Nat. Acad. Sci. 101:16819–16824), but we provide preliminary data indicating that *P. antofagastensis* can also be frugivorous.

Our observations were based on data from 10 *P. antofagastensis* collected during January 2005 in Valle de Chaschuil, Catamarca Province, Argentina (27.72°S, 68.15°W, datum: WGS84; elev. 4200 m). Soil is sandy and rocky; the vegetation is shrub-steppe. The climate is cold and semiarid with broad diel thermal range and intense solar radiation (Boretto and Ibargüengoytía 2006, *op. cit*.. The lizards were taken to the laboratory, where they defecated; we examined their feces with a magnifying lens. We analyze a total of 10 feces, counting the number of seeds of fruits present. We estimated the percent volume of non-seed vegetable matter as a function of total fecal volume.

We found no arthropod remains. Only plant material representing the seeds of *Lycium chanar* and *Ephedra multiflora*, and non-seed material representing leaves or fruit tissue of *L. chanar* and *E. multiflora* were observed. The fruits of *L. chanar* and *E. multiflora* in the feces were in various stages of digestion. We counted a total of 488 seeds. Mean number of *L. chanar* seeds in each fecal pellet was 45.7 (SD = 30.9, range: 0–103); mean number of *E. multiflora* seed was 3.1 (SD = 5.3, range: 0–16). Mean percent volume of non-seed vegetative material was 56.4 (SD = 7.1, range: 45–65).

Our results indicate that the fruits of *L. chanar* and *E. multiflora* represent an important source of nutrition for *P. antofagastensis*. Other Andean lizards (e.g., *Pristidactylus scapulatus* [Acosta et al. 2004. Herpetol. Rev. 35:171–172], *Liolaemus ruibali* [Villavicencio et al. 2005. Multequina: Latin American J. Nat. Res. 14:47–52], and *Homonota andicola* [Blanco and Acosta 2005. VI Congreso Argentino de Herpetología. 70 pp.]) also consume *Lycium* and *Ephedra* fruits. Fruits represent food with easily usable energy wealth for many squamates (Pough et al. 2001. Herpetology. Prentice Hall, New Jersey. 612 pp.). *Phymaturus antofagastensis* also has the potential to behave as a seed disperser. Given the observed frugivory, plant-lizard interactions involving *P. antofagastensis* and fruit-producing shrubs deserve investigation. We thank M. Hayes for comments and suggestions.

Submitted by **JUAN CARLOS ACOSTA** (e-mail: jcacosta@sinectis.com.ar), **HÉCTOR JOSÉ VILLAVICENCIO** (e-mail: hjvillavicenciomartin@yahoo.com.ar),***** and **GRACIELA BLANCO,** Departamento de Biología e Instituto y Museo de Ciencias Naturales, Facultad de Ciencias Exactas, Físicas y Naturales, Universidad Nacional de San Juan, Avenida España 400 (N), Caixa Postal 5400, San Juan, Argentina. *Becario CONICET.

PLESTIODON FASCIATUS (Common Five-lined Skink)**. ESCAPE BEHAVIOR.** *Plestiodon fasciatus* is a small, diurnal skink that occurs throughout most of the eastern half of the United States (Mount 1996. The Reptiles and Amphibians of Alabama. University of Alabama Press. Tuscaloosa, Alabama). Between 1400 and 1500 h on 27 June 2006, during a visit to Glen Burnie, Maryland (39.16°N, 76.61°W), I witnessed what appeared to be a distinctive escape maneuver by *P. fasciatus*. Skinks (N = 7) basking on a wooden railing along a path through a woodland fled for cover under a log when I approached. When I pursued the first skink by overturning the log under which it had taken cover, a large number of ants were found swarming beneath it. The same situation arose with another four *P. fasciatus*, each found along the same path. These lizards were also beneath cover objects (debris and leaf litter) housing ants. All five lizards observed appeared to be sub-adults,between ca. 50 and 65 mm SVL. When 25 nearby cover objects—similar to those used by skinks—were randomly sampled, only four housed ants beneath them, and (excepting the five mentioned above) only one had a high density of ants similar to that observed under cover objects utilized for escape by *P. fasciatus*.

Once established, *P. fasciatus* appear to have stable home ranges (Fitch 1954. Univ. Kansas Publ. Mus. Nat. Hist. 8:1–156). Consequently, individuals should be familiar with most possible escape routes within their home range. My observations imply that *P. fasciatus* preferentially flee to ant-associated cover objects during escape attempts. To my knowledge, this behavior is undocumented among lizards. Predators that pursue skinks under cover objects with ants may be deterred by the agitated ants. The smooth scales and associated osteoderms of *Plestiodon* would protect them from ants. Further observations are needed to determine how widespread this behavior is and to test the hypothesis of whether *P. fasciatus* actually exploit ant colonies within their home range for defense.

I thank Laurie J. Vitt, Mark O'Shea, Graeme Ruxton and Maggie Reilly for their comments on the text.

Submitted by **KEVIN ARBUCKLE**, 10 Crinan Place, Bellshill, North Lanarkshire, Scotland, United Kingdom, ML4 2LZ; e-mail: snakeboy349@msn.com.

PLESTIODON MULTIVIRGATUS EPIPLEUROTUS (Variable Skink). **MATING.** Little is known about reproduction in *Plestiodon multivirgatus epipleurotus* (Degenhardt et al. 1996. Amphibians and Reptiles of New Mexico. University of New Mexico Press, Albuquerque. 431 pp.). Everett (1971. J. Herpetol. 5:189–190) provided details of courtship and mating among captive specimens (two males and one female) from Lamb County, Texas, but mating has not been observed in the field. Hence, we report a field observation of mating in a high altitude population of *P. m. epipleurotus* near the southwestern limit of this taxon's range (Brennan and Holycross 2006. A Field Guide to Amphibians and Reptiles in Arizona. Arizona Game and Fish Department, Phoenix. 150 pp.).

At ca. 1300 h on 2 May 2007, CBG discovered a mating pair of *P. m. epipleurotus* in a small depression under a piece of particleboard in Fry Canyon, just west of US Hwy 89A, ca. 20 km SSW Flagstaff, Coconino Co., Arizona (35.0637833°N, 111.73315ºW, datum: WGS84; 2003 m elev.). Ambient temperature was 18.3°C and the sky was mostly overcast. A considerably smaller male (ca. 50 mm SVL) was in copula with a larger female (ca. 70 mm SVL), biting the left side of her neck and oriented on her left side. The lizards were apparently not disturbed, as they remained in copula after they were uncovered until the board was quickly (<10 sec) returned to its original position.

Timing of this mating event appears consistent with the seasonal appearance of eggs and hatchlings among other high-elevation populations of this taxon. Gehlbach (1956. Trans. Kansas Acad. Sci. 59:364–373) found hatchlings on 29 July at 2060 m elevation in McKinley Co., New Mexico. He also discovered a clutch of eggs on 28 June and hatchlings on 29 July and 17 August in the Zuni Mountains (2215 m), Cibola Co., New Mexico (Gehlbach 1965. Proc. U.S. Nat. Mus. 116:243–332). Similarly, Maslin (1957. Herpetologica 13:87–90) reported 4 egg clutches found 7 June on the Upper Colorado Plateau (2555 m) in San Juan Co., Utah. Our observation of an early May mating implies that egg development likely requires 5–7 weeks before the onset of oviposition.

We thank Andrew Holycross for suggestions and Kenneth Krysko for key literature.

Submitted by **CHARLES B. GROGAN**, Department of Music, University of Arizona, Tucson, Arizona 85285, USA (e-mail: cgrog@earthlink.net); and **WILLIAM L. GROGAN, JR**., Department of Biological Sciences, Salisbury University, Salisbury, Maryland 21801, USA (e-mail: wlgrogan@salisbury.edu).

PLICA PLICA (NCN). **REPRODUCTION.** The scansorial arboreal tropidurid *Plica plica* is widespread in Amazonia (Vitt 1990. Can. J. Zool. 69:504–511). Reproduction in *P. plica* has been studied (Vitt, *op. cit*.), but data are lacking on the incubation interval. Here, we provide preliminary data on the incubation interval for *P. plica*.

On 4 March 2005, the day after the capture of a gravid female (145 mm SVL, 80 g) at Floresta Nacional de Carajás, Estado do Pará, Brazil (06.30°S, 50.20°W; elev. 500 m), she laid a clutch of five eggs in laboratory. The clutch was maintained in a plastic tub with moist sand and litter, at a constant 24°C. Placed near a glass window, the tub received indirect ambient light throughout incubation, but light regime was not rigorously controlled. One egg spoiled the week after oviposition and was discarded. The remaining four eggs hatched in 122 days (on 4 July 2005). Hatching from the first three eggs was almost simultaneous (at 1340, 1345 and 1346 h), but the fourth hatchling emerged almost two hours later (1538 h). Table 1 provides measurements of hatchlings. Simultaneous hatching of the first 3 eggs agrees with Vitt (*op. cit*.), who described synchronous hatching of *P. plica* eggs after disturbance.

Compahia Vale do Rio Doce supported this work.

TABLE 1. Measurements of four *Plica plica* hatchlings from Estado do Pará, Brazil.

Individual	Snout–vent length (mm)	Tail length (mm)	Mass (g)
1	40.8	77.0	3.0
2	41.5	77.0	3.0
3	41.0	74.0	3.0
4	39.6	72.0	2.9

Submitted by **ELILDO A.R. CARVALHO-JR, JOSAPHAT A.D. CHAVES,** and **CÉSAR DE SÁ CARVALHO NETO,** Instituto Ambiental Vale do Rio Doce, Estrada Raymundo Mascarenhas, km 26, s/n, 68516-000 Serra dos Carajás, Parauapebas, Pará, Brazil; e-mail: elildojr@gmail.com.

POLYCHRUS MARMORATUS (NCN). **MATING.** The neotropical genus *Polychrus* contains five medium-sized (120–150 mm) diurnal arboreal species (Peters and Orejas-Miranda 1986. Catalogue of Neotropical Squamata. Part II, Lizards and Amphisbaenians. Revised ed. Smithsonian Institution, Washington, DC. 293 pp.). Vitt and Lacher (1981. Herpetologica 37:53–63) described mating in *P. acutirostris,* and Beebe recorded but did not describe mating in *P. marmoratus* (Beebe 1944. Zoologica 29:195–216). Here we describe mating by *P. marmoratus* in eastern Brazilian Amazônia.

Our observations were made at Floresta Nacional de Carajás (06.0569444°S, 50.145°W, datum: SAD69, elev. 500 m). Lizards were captured on the morning of 23 December 2004 in low vegetation along a road in continuous forest. They were placed in a transparent plastic tub (25 × 35 × 15 cm) in a room under artificial (fluorescent) light where the temperature was a constant 24°C (slightly below field temperature at the time of capture). Observations began at 1800 h the same day, when we found the male riding the female's back while biting her neck and trying to penetrate her cloaca. Copulation began at 1820 h and lasted more than an hour. The male made rhythmic movements with his pelvis until 1930 h, and during all this time, a white secretion was visible on his vent. After copulation, the male disengaged from the female, gaped and made atypically fast respiratory movements. The male remained on female's back until 2315 h. The female remained passive througout all the process and no interactions were observed after separation.

Mating in *P. marmoratus* is similar to mating in *P. acutirostris*. However, prominent differences exist, including the longer copulatory period and the absence of aggressive behaviour on the part of the female post-copulation; however, these differences may be artifacts of the captivity that only field observations can elucidate. Vitt ánd Lacher (*op. cit.*) observed female aggressive behavior toward the male before copulation and concluded that females select their mates. Our observations began after mating had begun, preventing us assessing pre-copulatory behavior.

Compahia Vale do Rko Doce supported this work.

Submitted by **ELILDO A. R. CARVALHO-JR** and **MARIA LÚCIA C. B. CAMPELLO**, Instituto Ambiental Vale do Rio Doce, Estrada Raymundo Mascarenhas, km 26, s/n, 68516-000 Serra dos Carajás, Parauapebas, Pará, Brazil; e-mail: elildojr@gmail.com.

RHACODACTYLUS AURICULATUS (Gargoyle Gecko). **SAP FEEDING.** *Rhacodactylus auriculatus* is a large (125 mm maximum SVL) diplodactylid gecko endemic to mainland New Caledonia (Bauer and Sadlier 2000. The Herpetofauna of New Caledonia. SSAR, Ithaca, New York. 322 pp.). Between 28 July and 4 August 2004, an adult female (122.8 mm SVL, 36.0 g) that was being tracked by radiotelemetry in Parc Provincial de la Rivière Bleue, Province Sud (22.1000°S, 166.6444444°E, datum: WGS84; elev. 225 m), was seen licking sap from a *Cunonia macrophylla* tree on three separate nights. During daylight hours, she perched on vegetation within a radius of < 3 m from the tree, returning within 2 h of sunset to feed on a wound on the tree trunk. During video-documentation lasting 69 min 56 sec, this female licked the sap 662 times (Fig. 1). Licking was intermittent; several consecutive licks were followed by pauses of up to 1 min 41 sec.

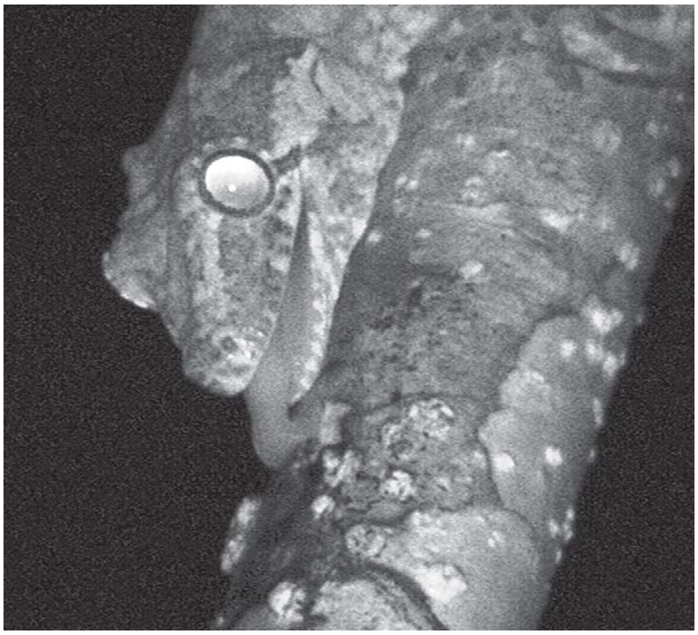


FIG. 1. Infrared video still of *Rhacodactylus auriculatus* feeding on the sap of *Cunonia macrophylla*.

Rhacodactylus auriculatus has been reported to feed on flowers and flower parts since the species was first described. Bavay (1869. Mém. Soc. Linn. Normandie 15:1–37) observed *R. auriculatus* feeding on flowers of *Geissois* spp., another member of the Cunoniaceae. Flower consumption by *R. auriculatus* has further been confirmed by the recovery of anthers and stamens referable to either the family Cunoniaceae or Myrtaceae from the stomach of a preserved specimen (Bauer and Sadlier 1994. Russ. J. Herpetol. 1:108–113) and Bauer and Sadlier (2001. Amphib. Rept. Conserv. 2:24–29) reported *R. auriculatus* activity on a flowering *Geissois* sp., though ingestion of plant material was not observed. Our observation constitutes the first field-based report of *R. auriculatus* feeding on sap and further supports the regular dietary use of plants of the Cunoniaceae by *R. auriculatus*.

Many gecko species (chiefly insular forms) have been implicated in frugivory or nectivory (Whitaker 1987. New Zeal. J. Bot. 25:315–328; Cooper and Vitt 2002. J. Zool. London 257:487–517), however field observations of feeding on sap has previously been recorded only for three species of the genus *Gehyra* (Couper et al. 1995. Mem. Qld. Mus. 38:396; Dell 1985. W. Aust. Nat. 16:69–70; Letnic and Madden 1998. W. Aust. Nat. 21:207–208). Because they are rapidly absorbed by the digestive tract, nectar and sap are difficult to identify in gut contents retrieved from preserved specimens. Flower parts consumed incidentally to high-energy nectar can indicate nectivory (Bauer and Sadlier 1994, *op. cit.*), but sap feeding is virtually undetectable unless a feeding event is actually observed.

Submitted by **JOSHUA P. SNYDER** (e-mail: joshua.p.snyder@villanova.edu), **LESLIE A. SNYDER**, and **AARON M. BAUER**, Department of Biology, Villanova University, 800 Lancaster Avenue, Villanova, Pennsylvania 19085, USA.

SCELOPORUS OLIVACEUS (Texas Spiny Lizard). **PREDATION.** *Sceloporus olivaceus* occurs from the Texas-Oklahoma border into México, extending to southern Tamaulipas, all through Nuevo León, southeastern Coahuila, northeast San Luis Potosí, and a tiny portion of northern Veracruz (Kennedy 1973. Cat. Amer. Amphib. Rept 143.1–143.4; Smith 1979. Handbook of Lizards of the United States and Canada. Comstock Publishing Associates, Ithaca, New York. 557 pp; Köhler and Heimes 2002. Stachelleguane. Herpeton, Offenbach, Bundesrepublik, Deutschland. 174 pp.). Several snake predators of *S. olivaceus* are reported, including *Masticophis flagellum*, *Hypsiglena ochrorhyncha texana*, *Crotalus lepidus lepidus,* and *Sistrurus catenatus* (Strecker 1927. Contr. Baylor Univ. Mus. [10]:1–14; Werler 1951. Zoologica 36:37–48; Wright and Wright 1957. Handbook of the Snakes of the United of the United States and Canada. Cornell University Press, Ithaca, New York. 564 pp.; Greene and Oliver 1965. Herpetologica 21:225–228), but *Imantodes cenchoa* is not among them. Hence, here we report an observation of *I. cenchoa* predation on *S. olivaceus*.

At 2343 h on 16 July 2006 during a collecting trip through the municipality of Aldama (state of Tamaulipas), we found a DOR adult female *I. cenchoa* (480 mm SVL, 223 mm TL, 19.81 g) as we were road-collecting at 8 km E of Aldama-Barra de Tordo (22.9414194°N, 99.9954°W, datum: NAD27; elev. 141 m). As we were preserving the specimen, we extracted a juvenile *Sceloporus olivaceus* (37 mm SVL, 65 mm TL, 1.94 g) from an expanded loop in the body mid-section. This snake species had been repeatedly found on this road in previous years. The area of Barra del Tordo is undergoing intensive human development, which has greatly increased the number of DOR animals encountered.

Imantodes cenchoa, which occurs at low to moderate elevations in Mexico from Chiapas on the Pacific slope to Tamaulipas on the Atlantic slope (Lee 1996. The Amphibians and Reptiles of the Yucatan Peninsula. Comstock Publishing Associates, Cornell University Press, Ithaca, New York. 500 pp.), is documented to consume lizards and frogs. Myers (1982. Amer. Mus. Novit. 2738:1–50) reported that anoles make up most of its diet, and Landy et al. (1966. J. Ohio Herpetol. Soc. 5:93–101) found an unidentifed *Anolis* sp. and reptile eggs in the stomach of one individual. Specific reports include *Anolis capito* and *A. uniformis* (Stuart 1948. Misc. Publ. Zool. Univ. Michigan [91]:1–31); *A. marianrum* (Gutierrez and Arredondo-S. 2005. Herpetol. Review.36:324); and juvenile *Basiliscus vittatus* (Lee, *op. cit.*). However, ours is the first report of *S. olivaceus* as prey.

The specimens of *S. olivaceus* (UANL 6831) and *I. cenchoa* (UANL 6830) were deposited in the herpetological collection of the Universidad Autónoma de Nuevo Leon. Research and collecting were conducted under the authority of SEMARNAT scientific research permits OFICIO NÚM/SGPA/DGVS/00800 issued to DL.

Submitted by **DAVID LAZCANO,** Universidad Autónoma de Nuevo León, Facultad de Ciencias Biológicas, Laboratorio de Herpetología, Apartado, Postal - 513, San Nicolás de los Garza, Nuevo León, C.P. 66450, México (e-mail: dvlazcano@hotmail.com); **ALAN KARDON,** Reptile/Amphibian/Aquarium Department, San Antonio Zoo, 3903 N. St. Mary's Street, San Antonio, Texas 78212, USA (e-mail: reptiles@sazoo-aq.org); **IAN RECCHIO**, 5333 Zoo Drive, Las Angeles, California 90027, USA (e-mail: Pardoo2@aol.com); and **CHRIS RODRIGUEZ**, Department of Biology, California State University, Northridge,18111 Nordhoff Street Northridge, California 91330-8303, USA (e-mail: sleepingbird@aol.com).

STENOCERCUS CADUCUS (NCN). **REPRODUCTION.** Data on reproduction in *Stenocercus caducus* are scarce, and little is known about its biology in Paraguay. Cei (1993. Mus. Reg. Sci. Nat. Torino Monogr. 14:1–949) pointed out that few data exist on its reproductive activity. Clutch size has been reported in related species, such as *S. azureus* (Carreira and Baletta 2004. Herpetol. Rev. 35:270; Torres-Carvajal 2004. Herpetol. Rev. 35:172), but the nesting habits of *S. caducus* are unreported. Here, we provide preliminary observations on nesting in *S. caducus*.

At 1640–1700 h on 15 November 2006, we found a female *S. caducus* laying eggs along a forest path at Kangüery Biological Station (27.5126944°S, 55.7852222°W, datum: WGS84; elev. 158 m), inside San Rafael National Park. The female was laying the eggs in a small burrow (3–4 cm depth and 4–5 cm width) made in the earth underground, covered with leaf litter. The shape of the nesting burrow was a simple round hole, and the egg chamber was equal to or slightly small than the opening. Two eggs were deposited with an interval of ca. 10 min between them. The just-laid eggs were pale grey with white longitudinal stripes; after less than 10 sec, the eggs turned completely white, the stripes disappearing. Egg shape also changed, because just-laid eggs were bilaterally symmetrical, becoming ovoid as they dried in contact with air. We could not obtain precise egg measurements because eggs were not removed from the nest, but we estimated that they averaged 23 mm in major axis diameter. The female was 67.2 mm of SVL and 142.0 mm total length. Egg major axis diameter was 34% of the female's SVL. Measurements were taken once she finished laying eggs, and she was subsequently released at the same place.

While the female laid eggs, she was vulnerable to predator attack; however, she appeared highly cryptic on the leaf litter background. Only her hind limbs and posterior body were inside the small burrow; the rest of the body, including a large portion of the long tail, was exposed. The next day, we re-examined the nest location, and found it covered with soil and leaf litter; we could see no obvious evidence of the nest made the day before.

We thank A. A. Yanosky, J. L. Cartes, P. Smith, and M. Hayes for suggestions on the manuscript. This work was done within the framework of the Biological Monitoring of San Rafael project, supported by the Forestry Bureau, Council of Agriculture (Taiwan Government) through BirdLife International.

Submitted by **PIER CACCIALI**, Asociación Guyra Paraguay, Cnel. Rafale Franco 381, Asunción, Paraguay (e-mail: piercantropus@gmail.com); and **MÓNICA RUMBO**, Sección Etología, Facultad de Ciencias, Iguá 4225, Montevideo, Uruguay (e-mail: mrumboca@yahoo.com).

TROPIDURUS HISPIDUS (NCN). **HATCHLING SIZE.** *Tropidurus hispidus* has a broad distribution from central-eastern and northeastern Brazil to Venezuela (Rodrigues 1987. Arq. Zool. 31:105–230; Rodrigues 1988. *In* Heyer and Vanzolini [eds.], Proceedings of a Workshop on Neotropical Distribution Patterns, pp.

305–315. Academia Brasileira de Ciências, Rio de Janeiro, Brazil). Data are available on clutch size (Vitt 1983. Copeia 1983:131–141), but information on hatchling size is lacking. Here, we provide preliminary data on hatchling size for *T. hispidus*.

At 0820 h on 8 June 2005, CMCAL and TBGC collected 8 eggs of *T. hispidus* at the Parque Estadual das Dunas do Natal (05.8135278°S, 35.1920278°W, datum: WGS84; elev. 72 m), Natal City, Estado do Rio Grande do Norte, Brazil. Located within Atlantic Forest Domain, the nest site, a small terrestrial cavity (5 cm diameter × 3 cm deep) covered with herbaceous and shrubby vegetation (notably *Anthurium affine*, *Aechmea aquilega*, and *Krameria tomentosa*), was encountered during a transect survey. On 4 April 2007 at 0910 h, EMXF collected one egg (1109 mm^3, 0.68 g) of this species, next to DBEZ - Departamento de Botânica, Ecologia e Zoologia (Department of Botany, Ecology and Zoology) at the Campus of Universidade Federal do Rio Grande do Norte – UFRN (05.8426667°S, 35.2018611°W; elev. 69 m), Natal City. The collection location of this egg was a garden area surrounded by a forest patch. The single egg was found in soil beneath sparse leaf litter (< 1 cm deep). Eggs from each collection date were placed in a terrarium (20 × 12 × 20 cm) in a sand substrate, and maintained at the Laboratório de Herpetologia (Departamento de Botânica, Ecologia e Zoologia/UFRN). We placed the terrarium next to a window protected from direct solar radiation, but we made no efforts to otherwise control light or temperature; incubation occurred under ambient conditions. In Natal City, ambient temperatures during the June–July incubation interval for the first clutch varied from 22.0°C to 30.0°C, whereas ambient temperatures during the brief April incubation of the second single egg varied from 24.0°C to 34.0°C.

On 12 July 2005, about five weeks after the first clutch was found, juveniles began to emerge; on 8 April 2007, four days later the single egg was found, the juvenile emerged. Body measurements were taken immediately upon hatching, and each individual was sexed following euthanization (Table 1). Coefficients of variation (CV) for data among all hatchlings were quite low (< 0.05) except for mass (CV = 0.25). Tail length/body length ratio differed significantly between males (mean = 1.70 ± 0.04 mm; N = 4) and females (mean = 1.59 ± 0.04 mm; N = 5; Mann-Whitney U test: $P = 0.0143$).

TABLE 1. Data on nine *Tropidurus hispidus* hatched in the Laboratory (Estado do Rio Grande do Norte, Natal City, Brazil). Individuals 1–8 were from one clutch; individual 9 was from a second clutch. Snout–vent length (SVL), tail length (Tail), head length (HL), head width (HW) measurements are in millimeters; mass is in grams.

	Morphological Variables				
Individual	SVL	Tail	HL	HW	Mass
1 (Female)	28.2	45.7	9.4	6.5	0.58
2 (Female)	27.7	43.5	8.8	6.1	0.52
3 (Male)	27.6	48.9	9.4	6.4	0.59
4 (Male)	28.7	48.6	9.6	6.6	0.74
5 (Male)	28.6	48.3	9.2	6.1	0.55
6 (Male)	28.4	47.5	9.2	6.3	0.63
7 (Female)	27.2	43.3	9.0	5.7	0.37
8 (Female)	27.6	45.7	8.8	6.0	0.35
9 (Female)	27.6	42.7	9.3	6.6	0.40
Mean	28.0	46.0	9.2	6.3	0.53
SD	0.53	2.4	0.28	0.30	0.13

Mean body size of nine *T. hispidus* hatchlings is similar to that observed by Vitt (*op. cit.*) for individuals hatched in the laboratory (mean = 27.8 ± 0.45 mm SVL; N = 5), but their average mass is somewhat less than that recorded by Vitt (*op. cit.*; mean = 0.74 ± 0.09 g).

The *T. hispidus* (CHBEZ 1167–1174; 1715) were deposited in the herpetological collection of Universidade Federal do Rio Grande do Norte, Natal City. We thank two anonymous reviewers for helpful comments. The Conselho Nacional de Desenvolvimento Científico e Tecnológico (CNPq) supported LBR with a research grant (Process 141993/2006-5).

Submitted by **LEONARDO B. RIBEIRO,** Programa de Pós-Graduação em Psicobiologia, Universidade Federal do Rio Grande do Norte, Centro de Biociências, Departamento de Fisiologia, Caixa Postal 1511, 59078-970, Natal, RN, Brazil (e-mail: ribeiro.lb@gmail.com); **CAROLINA M. C. A. LISBOA** (e-mail: carolisboabio@yahoo.com.br) and **THAÍS B. GUEDES** (e-mail: thaisbguedes@yahoo.com.br), Departamento de Botânica, Ecologia e Zoologia, Centro de Biociências, Universidade Federal do Rio Grande do Norte, 59072-970, Natal, RN, Brazil; **MIGUEL F. KOLODIUK** (e-mail: miguelkolodiuk@yahoo.com.br), and **ELIZA M. X. FREIRE** (e-mail: elizajuju@ufrnet.br), Programa de Pós-Graduação em Psicobiologia, Universidade Federal do Rio Grande do Norte, Centro de Biociências, Departamento de Fisiologia, Caixa Postal 1511, 59078-970, Natal, RN, Brazil.

VARANUS KOMODOENSIS (Komodo Dragon). **OCCURRENCE.** *Varanus komodoensis* is a large monitor endemic to the Lesser Sunda region of southeastern Indonesia. Over the last 30 years, extant populations of *V. komodoensis* have been recorded from six islands (Auffenberg 1981. The Behavioral Ecology of the Komodo Monitor. Univ. Press of Florida, Gainesville, Florida. 406 pp.), five of which occur within Komodo National Park (KNP). However, a 1998 survey failed to detect any sign of *V. komodoensis* on Padar Island, a small mountainous island (20 km^2) within KNP, suggesting that this population was extirpated (Ciofi and deBoer 2004. Herpetol. J. 14:99–107). In 1969–1970, Auffenberg (*op. cit.*) had estimated that ca. 60 dragons inhabited this hilly and largely savannah-covered island. Anecdotal evidence suggests that this population of *V. komodoensis* had been extirpated by the early 1980s, possibly because of illegal harvesting of the Timor Deer (*Cervus timorensis*), their preferred prey.

Recently, reports from fishermen plying local waters have suggested the renewed presence of *V. komodoensis* on Padar Island. To confirm these reports, we conducted trapping and visual surveys on Padar Island over five days in December 2004. To assess for *V. komodoensis* presence, direct and indirect methods were used. Direct methods involved the placement of two aluminium box traps (300 cm × 50 cm × 50 cm) baited with ca. 0.5 kg of goat meat. These traps were positioned in the largest forested valley on

FIG. 1. Gastric pellet believed to be that of a Komodo Dragon collected from Padar Island that contains the flight feathers from a sea bird (top of pellet) and the hoof of a juvenile Timor Deer (mid center of pellet) surrounded by a matrix of grass. For scale, the ruler is 30 cm.

the south eastern corner of the island. Indirect methods involved focal searches along 30 × 150 m long transects randomly positioned across the island from which any signs of *V. komodoensis* were recorded. Transects were concurrently used for assessing the density of Timor Deer, based on fecal pellet counts. Visual sampling was also conducted between transects when walking to the start point of the next transect. The purpose of indirect sampling was to identify potential signs specific to Komodo Dragons, including: 1) gastric pellets or large boluses of indigestible food matter (keratin and often bone-based material) encased in grass, that *V. komodoensis* regurgitate after ingestion of prey; 2) foot or tail tracks or burrows; and 3) feces.

Despite laying out two baited traps, no *V. komodoensis* were captured or directly sighted on Padar over the trapping period. However, fresh tracks with the characteristic accompanying tail drag and a body pit that was likely to have been made on the day of sampling were recorded on a beach (8.656417°S, 119.600722°E, datum: WGS, elev. 4 m) ca. 1.3 km WSW from our trap locations. In addition, we found several feces (8.655175ºS, 119.5814722ºE; and 8.6554917ºS; 119.5814361ºE) that contained the remains of sea turtle eggs (either *Eretmochelys imbricata* or *Chelonia mydas*), and *C. timorensis*, prey items consistent with *V. komodoensis*. However, the sign most suggestive of *V. komodoensis* presence was a 40 cm × 10 cm gastric pellet (Fig. 1). This pellet contained the remains of deer bones and fur, and the remains of a large raptor (probably a White-bellied Sea Eagle, *Haliaeetus leucogaster*).

Our results suggest that either *V. komodoensis* engaged in inter-island movement or that the species was not completely extirpated on Padar Island. Based on the low density of tracks and scats, only one dragon may now occupy Padar Island. If inter-island movement was the source of this animal, the closest occupied islands would be Rinca (1.95 km) and Komodo Island (4.71 km). Anecdotal reports indicate that *V. komodoensis* has some capacity to swim and thus potentially move among islands. However, the frequency of such events in both ecological (TJ, unpubl. data) and evolutionary time (Ciofi and Bruford 1999. Mol. Ecol. 8:17–30) seems infrequent, suggesting that fidelity of individuals to their respective islands is high. Even within islands, dragons are very rarely recorded to move among valleys (TJ, unpubl. data).

Submitted by **TIM JESSOP**, Conservation and Research of Endangered Species, Zoological Society of San Diego, Escondido, California 92027, USA & Department of Wildlife Conservation and Research, Zoos Victoria, PO Box 74 Parkville VIC 3052, Australia (e-mail: tjessop@zoo.org.au); **ALO SAHU** and **DEVI OPAT**, Komodo National Park, Labuan Bajo, Flores, NTT, Indonesia.

VARANUS KOMODOENSIS (Komodo Dragon). **PARASITES.** Ticks that parasitize varanid lizards are primarily from the genera *Amblyomma* and *Aponomma* (King and Green 1999. Goannas: The Biology of Varanid Lizards. Univ. New South Wales Press, Sydney. 116 pp.). Ticks as ectoparasites consume host blood, cause disease, and can function as important vectors for microparasites, which can negatively influence fitness (Main and Bull 2000. Oecologia 122:574–581). *Varanus komodoensis*, a large monitor lizard from southeastern Indonesia, has been documented to carry three species of tick, including *Amblyomma helvolum,* a generalist, and two host-specific species: *A. robinsoni* and *Aponomma komodoense* (Auffenberg 1981. The Behavioral Ecology of the Komodo Monitor. University Press of Florida, Gainesville, Florida. 406 pp.). However, little is known about either the general ecology or fitness implications of this host-parasite relationship. One aspect of this relationship that remains unknown is the possibility that ticks have evolved strategies to parasitize hatchling dragons upon emergence from their nests. From a parasite's perspective, the host environment of juvenile animals provide the least resistance with respect to immunological (i.e., lack of acquired immunity) or physical (e.g., skin thickness) barriers. Hence, here we report on the ability of ticks to parasitize hatchling Komodo Dragons recently emerged from three nests on Komodo Island.

In January of each observation year (2003–2006), a sample of known active nests of *V. komodoensis* were caged to enable capture of hatchlings as part of routine annual monitoring of the ecology and life-history of this species in Komodo National Park (Jessop et al. 2004. Biol. Conserv. 117:463–470). Prior to emergence in March/April (late wet season), nest cages were monitored twice daily (morning and afternoon coinciding with emergence of hatchlings from the ground) to enable measurement and marking of individual hatchlings prior to release. In 2004, we also examined for the presence of ectoparasites of hatchlings on Komodo Island, one of four extant populations within Komodo National Park.

From the three nests monitored on Komodo Island in 2004 (referred to as A, B, and C), 47 Komodo hatchlings were captured in March (estimated within 4 h post-emergence). These hatchlings averaged 95.5 ± 2.2 g in mass and 18.8 ± 0.1 cm SVL. Hatchlings from two of the three nests were found to be carrying nymphal ticks of one species, *Aponomma komodoense*. Two of 12 hatchlings (16.6%) from Nest A had ticks; one had 1 tick, the other had 3 (Table 1). Clutch B had 16 individuals, 9 (56.3%) of which carried ticks. The number of ticks ranged from 1 to 20, with an average number of ticks per hatchling of 7.4 ± 2.5. Hatchlings (N = 18) from Nest C lacked ticks.

Our results indicate that alongside larger Komodo Dragons, hatchlings can act as tick hosts. By parasitizing hatchlings on emergence, ticks could greatly increase their capacity to find new and, potentially of greater importance, immunologically naïve hosts (i.e., no acquired resistance). Female dragons, which act as hosts for ticks, spend considerable periods of time around the nest both prior to and after oviposition and thus could enable ticks to deposit eggs (that remain quiescent) around the nest site until hatchlings emerge. Seasonal cues, including the conspicuous transition from an extended dry season through to the short summer wet season coinciding with the monsoon represent an important cue for many organisms breeding in the wet-dry tropics of Australasia (Madsen et al. 2006. Aust. J. Ecol. 31:30–37). This seasonal cue could provide necessary environmental information enabling nymphal ticks to synchronise their own hatching and questing activity with the emergence of hatchling dragons. At present, we do not know whether ticks impart specific fitness consequences to their hatchling hosts.

Submitted by **TIM JESSOP**, Conservation and Research of Endangered Species, Zoological Society of San Diego, Escondido, California 92027, USA & Department of Wildlife Conservation and Research, Zoos Victoria, PO Box 74 Parkville VIC 3052, Australia (e-mail: tjessop@zoo.org.au); and **M. JERI IMANSYAH**, Conservation and Research of Endangered Species, Zoological Society of San Diego, Escondido, California 92027, USA.

SQUAMATA – SNAKES

AGKISTRODON CONTORTRIX (Copperhead). **DIET.** *Agkistrodon contortrix* is known to take a variety of prey items, including the snakes *Carphophis amoenus*, *C. vermis*, *Coluber constrictor*, *Crotalus lepidus*, *Diadophis punctatus*, *Rhinocheilus lecontei*, *Storeria dekayi*, and *Tantilla coronata* (McCrystal and Green 1986. Herpetol. Rev. 17:61; Gloyd and Conant 1990. Snakes of the *Agkistrodon* Complex: A Monographic Review. Society for the Study of Amphibians and Reptiles, Oxford, Ohio. 614 pp.; Palmer and Braswell 1995. Reptiles of North Carolina. University of North Carolina Press, Chapel Hill. 412 pp.; Fitch 1999. A Kansas Snake Community: Composition and Changes Over 50 Years. Krieger Publishing Co., Malabar, Florida. 165 pp.; and references therein). Here we report a novel prey item for *A. contortrix*.

On 23 May 2007 at 1415 h, one of us (JRL) captured a female *A. contortrix* (260 mm SVL, 44 mm tail length, 15 g) on the Camp Shelby Joint Forces Training Center, Perry County, Mississippi, USA. The snake appeared to have recently fed, as indicated by a large bolus in its stomach. Upon palpating, the *A. contortrix* regurgitated a female *Storeria occipitomaculata*, a gravid *Anolis carolinensis*, and a *Scincella lateralis* of undetermined sex. To the best of our knowledge this observation represents the first instance of a *S. occipitomaculata* having been consumed by an *A. contortrix*.

Submitted by **JAMES R. LEE** (e-mail: jlee@tnc.org), **AARON L. HOLBROOK**, and **JENNIFER W. FREY**, The Nature Conservancy, Camp Shelby Joint Forces Training Center, CSJFTC-ENV Building 6678, Camp Shelby, Mississippi 39407, USA.

CROTALUS MOLOSSUS (Blacktail Rattlesnake). **MORPHOLOGY**. Congenital absence of the rattle in rattlesnakes (Crotalidae) has been previously reported for *Crotalus cerastes*, *C. horridus*, and *C. pricei* by Klauber (1972. Rattlesnakes. Their Habits, Life Histories, and Influence on Mankind. Univ. California Press, Berkeley, California. 740 pp.), for *C. atrox* by Painter et al. (1999. Herpetol. Rev. 30:44) and Holycross (2000. Herpetol. Rev. 31:177–178), for *C. viridis* by Holycross (2000. Herpetol. Rev. 31:177), and for *C. lepidus* by Christman et al. (2004. Herpetol. Rev. 35:62). We report here this irregular condition of a missing rattle from another rattlesnake species discovered on a site in far west Texas.

On 6 July 2007, at 2326 h, an adult male *Crotalus molossus* (701 mm SVL, 64 mm tail length, 238.9 g) was found by two of us (LM and VMS) 0.6 km SE of the headquarters complex on Indio Mountains Research Station (IMRS) located ca. 40 km SW of Van Horn, Hudspeth County, Texas, USA (30.773056°N, 105.011944°W, WGS84; 1233 m elev.). IMRS is administered by The University of Texas at El Paso and includes 39,000 acres of rugged Chihuahuan Desert scrub landscape.

Close examination of the posterior end of the tail revealed a blunt tip with overlapping scales devoid of scar tissue, which indicates that the absence of the rattle was congenital and not the result of prior trauma. After processing for body measurements, the rattlesnake was pit-tagged (AVID #: 025-258-063) for permanent identification and then released at original capture site. Because of the snake's size and healthy condition and because the rattleless feature has been reported for adults of other rattlesnake species, we infer that lacking a rattle has negligible effects on survivability. On the other hand, the condition appears to be rare because of the 42 individuals of *C. molossus* captured and marked on IMRS since 1997, only the individual reported herein exhibited the rattleless condition. A digital image is deposited in the photographic collection, Laboratory for Environmental Biology, Centennial Museum, The University of Texas at El Paso (UTEP G2007.1.1).

Submitted by **LUIS MIRANDA JR.** (e-mail: lmiranda1@miners.utep.edu), **VICENTE MATA-SILVA**, **STEVEN DILKS**, **HECTOR RIVEROLL JR.**, and **JERRY D. JOHNSON**, Department of Biological Sciences, The University of Texas at El Paso, El Paso, Texas 79968, USA.

LATICAUDA SAINTGIRONSI (Sea Krait). **PREDATION.** Field observations and laboratory experiments indicate that sea snakes quickly and efficiently kill their prey (fishes) by injecting potent venom (Ineich and Laboute 2002. Sea Snakes of New Caledonia. IRD et Muséum national d'Histoire naturelle Editions, Collection Faune et flore tropicales, Paris. 302 pp.). Rapid death is probably essential to foraging success in sea snakes. If death were delayed (as is the case in most species of terrestrial venomous snakes) the fish might escape and be difficult to locate in the marine environment. Here, we present the first record of any species of sea snake regurgitating a living prey item (Moray Eel, *Gymnothorax undulatus*).

In the course of conducting mark-recapture studies of large populations of *Laticauda saintgironsi* and *Laticauda laticaudata* on Signal Islet, New Caledonia (22.29°S, 166.29°E), we captured an adult male (695 mm SVL) *L. saintgironsi* on 8 December 2005 at 1740 h while it was returning from the sea. Immediately after capture, it regurgitated a *G. undulatus*. Spontaneous regurgitation is uncommon in Sea Kraits. We have captured over 3300 Sea Kraits with prey items in their stomachs and have observed less than 10 spontaneous regurgitations. Upon regurgitation, the eel dropped into shallow (5 cm) water and was not easy to catch. We rapidly placed the eel in a container filled with seawater (which we replaced every 15 min.). Initially, the eel moved slowly but was still able to swim. Multiple bite marks visible on the body and the eel's behavior suggest it had been envenomated. The eel became increasingly lethargic over time. After 78 min. it lay on its back, but righted itself after tactile stimulation with a finger. After 95 min., gill (respiratory) movements became almost undetectable. It died 110 min after the regurgitation, very likely due to the effect of the venom. Overall, the eel survived ca. two hours after being bitten, swallowed, and then regurgitated. The eel was deposited in a collection of Sea Krait prey (# 630) at the Centre d'Etudes Biologiques de Chizé (CEBC-CNRS UPR 1934). Our observations indicate that although the venom of *L. saintgironsi* certainly causes a paralysis (perhaps transitory or partial) it does not necessarily kill prey instantaneously. Our observation also indicates that *L. saintgironsi* sometimes forages very near their home islands.

We thank the Direction des Ressources Naturelles de la Province Sud, New Caledonia for issuing the necessary permits.

Submitted by **MARGOT DE CRIGNIS**, **FRANCOIS BRISCHOUX**, **XAVIER BONNET**, and **SOPHIE LORIOUX**, CEBC-CNRS UPR 1934, Centre d'Etudes Biologiques de Chizé, F-79360 Villiers en Bois, France.

MICRURUS CORALLINUS (Painted Coral Snake). **DIET.** The diet of *Micrurus corallinus* includes amphisbaenians, snakes, lizards, and also caecilians (Marques and Sazima 1997. Herptol. Nat. Hist. 5:88–91; Roze 1996. Coral Snakes of the Americas: Biology, Identification and Venoms. Krieger Publ. Co., Malabar, Florida. 328 pp.). The natural predators of *Luetkenotyphlus brasiliensis* (São Paulo Caecilian) are largely unknown (Taylor 1968. The Caecilians of the World. Kansas Univ. Press, Lawrence. 848 pp.). Here, we report a previously unpublished predator-prey relationship.

The record is based on museum specimens from the herpetological collection of the Zoological Museum Hamburg (ZMH). A *L. brasiliensis* (249 mm total length) was removed from the stomach of a *M. corallinus* (ZMH R08751; 899 mm total length) collected by Wilhelm Ehrhardt in March 1910 at "Rio Humboldt-Gebiet (Flussgebiet des Itapocú), Brasilien" (in the vicinity of the Rio Humboldt River basin of the Itapocú, Corupá, Santa Catarina, Brazil). Orientation of the prey in the gut indicates that the snake ingested the caecilian tail-first. The caudal end of the caecilian is partially digested and the last 39 vertebrae are exposed. The anterior part of the caecilian up to the 89th primary annulus is completely undamaged, which is about two-thirds of the animal compared to the 125–131 primary annuli reported for *L. brasiliensis* (Nussbaum 1986. J. Herpetol. 30:441–444).

Submitted by **THOMAS KLEINTEICH,** Universität Hamburg, Biozentrum Grindel und Zoologisches Museum, Martin-Luther-King Platz 3, 20146 Hamburg, Germany (e-mail: Thomas.Kleinteich@uni-hamburg.de); **ALEXANDER GUTSCHE,** Humboldt-Universität zu Berlin, Department of Biology, Sensory Biology, Invalidenstrasse 43, 10115 Berlin, Germany (e-mail: Alexander-Gutsche@web.de); and **JAKOB HALLERMANN,** Universität Hamburg, Biozentrum Grindel und Zoologisches Museum, Martin-Luther-King Platz 3, 20146 Hamburg, Germany (e-mail: Hallermann@zoologie.uni-hamburg.de).

PANTHEROPHIS VULPINA VULPINA (Western Fox Snake). **MORTALITY.** Sources of mortality in *Pantherophis vulpina vulpina* are rarely reported. Ernst and Ernst (2003. Snakes of the United States and Canada. Smithsonian Books, Washington D.C.) indicate that predatory birds take adult *P. v. vulpina*, and suggest that carnivorous mammals, automobiles, and habitat destruction might also be significant sources of mortality.

Snake mortality due to prescribed fire has been reported in other regions of North America (e.g., Durbian 2006. Am. Midl. Nat. 155:329–334; Erwin and Stasiak 1979. Am. Midl. Nat. 101:247–249; Smith et al. 2001. Southwest. Nat. 46:54–61), but has not been reported from the upper Midwest. On 16 April 2007 a prescribed fire was ignited on a 10-acre prairie preserve on the McKenzie Environmental Center near Poynette (Columbia County, Wisconsin, USA). Sunny skies, an air temperature of 17°C, and 20% humidity characterize weather conditions on the day of the burn. A post-burn survey of the property revealed a dead *P. v. vulpina* (ca. 90–105 cm TL), which had not escaped the fire. The *P. v. vulpina* was located in a corner of the burn unit where head fire, flanking fire, and backing fire converged. Flame lengths averaged ca. 3 m, giving off an estimated 980 BTUs/foot/second. Vogt (1981. Natural History of Amphibians and Reptiles of Wisconsin. Milwaukee Public Museum, Milwaukee. 205 pp.) suggests that *P. v. vulpina* typically emerge from winter dormancy during the last two weeks of April. If the individual caught in the fire had emerged from dormancy immediately prior to the prescribed fire, it may have been too sluggish to avoid the fire. Because prescribed fire is an effective management tool for short grass habitats, we recommend conducting burns prior to April 1 to avoid causing direct mortality to reptiles emerging from dormancy.

Snakes have been listed as a major predator of *Meleagris gallapavo* (Wild Turkey) eggs and poults (Reagan and Morgan 1980. Proc. Natl. Wild Turkey Symp. 4:136–144), and numerous bird species are reported to actively attack snakes in response to a perceived threat (e.g., Gottfried 1979. Condor 81:251–257). However, defensive attacks upon potential nest raiders are rarely reported (Hewitt [ed.]. 1967. The Wild Turkey and its Management. The Wildlife Society, Washington, D.C. 589 pp.), and those few reports involve attacks against predatory birds or mammals (Johnson 1961. Auk 78:646; Spear et al. 2003. Canad. Field Nat. 117:645–647). The potential effect of these mortality sources on snake populations, particularly in regards to *M. gallapavo* (i.e., predation of snakes and actively attacking snakes to deter a potential nest-predator) is unknown as accounts are mostly anecdotal and difficult to substantiate (but see Beasom and Pattee 1975. Wilson Bull. 87:281–282).

On 16 May 2007 at 1215 h in Polk County (Wisconsin, USA) we (MMC, KAM, and DW) observed an adult hen *M. gallapavo* repeatedly striking the head of an adult *P. v. vulpina* (ca. 1 m TL, 323 g). The bird struck the animal with both its beak and feet for approximately 20 min. MMC and KAM left for approximately 45 min to retrieve a camera and returned to find the hen still striking the snake. The hen was disturbed by their approach and flew off, carrying the snake for a distance before dropping it. An examination revealed that, prior to the hen's attack, the snake was not underweight and bore no obvious deformities. The snake appeared to be alive throughout the majority of the attack, and attempted to defend itself against the hen before succumbing to its injuries. Although no poults or eggs were noticed, the fact that the turkey did not attempt to consume the snake, suggests that the hen's behavior was defensive rather than predatory.

Although *M. gallapavo* historically occurred in Wisconsin, they had been nearly or completely extirpated from the state by the late 1800s (Kubisiak et al. 2001. Wild Turkey Ecology and Management in Wisconsin. Wisconsin Dept. Nat. Res. PUB-SS-955 2001). Since the 1950s, significant effort has been made to re-establish populations of this species in several upper Midwestern states, including Wisconsin, with great success (Kubisiak et al. 2001, *op. cit.*). Whether turkey populations are a significant source of mortality for syntopic snake populations remains unclear.

Submitted by **MICHELLE CARLISLE**, Wisconsin Department of Natural Resources, 941 Mallard Lane #104, Balsam Lake, Wisconsin 54810, USA; **RALPH SHEFFER**, Wisconsin Department of Natural Resources, N3344 Stebbins Road, Poynette, Wisconsin 53955, USA; **KEVIN A. MORGAN**, Wisconsin Department of Natural Resources, 127 S. 4th Street, Barron, Wisconsin 54812, USA; **DOUG WAHLSTROM**, 1118 River Road, Dresser, Wisconsin 54009, USA; and **JOSHUA M. KAPFER**, Natural Resources Consulting, Inc., 119 Main Street, PO Box 128, Cottage Grove, Wisconsin 53527, USA (e-mail: jnjkapfer@hotmail.com).

PORTHIDIUM HESPERE (Western Hognose Viper). **ENDOPARASITES.** *Porthidium hespere* is limited in distribution to western Mexico where it is known only from a few specimens (Campbell and Lamar 2004. The Venomous Reptiles of the Western Hemisphere, Vol. 1. Comstock Publ. Assoc., Ithaca, New York. 475 pp.). There are no reports of endoparasites from *P. hespere*. The purpose of this report is to establish the initial helminth list for *P. hespere*. One road-kill female *P. hespere* (total length = 30 cm) was found on Hwy 200, 2.7 km N of the turn off to El Faro de Bucerias, (18.379272°N, 103.515758°W) Michoacán, Mexico on 10 August 2005. It was deposited in the Museo de Zoologia, Facultad de Ciencias, Universidad Nacional Aútonoma de Mexico, Mexico as MZFC 19742. Three nematodes were recovered from the carcass. These were placed in a drop of glycerol on a glass slide. A cover glass was placed on top and the preparation was examined under a compound microscope. The nematodes were identified as one adult male, one adult female, and a juvenile of *Ophidascaris arndti*. These were deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland as USNPC (100204).

Sphren (1929. Zool. Anz. 83:280–282) described *O. arndti* from specimens recovered from a *Lachesis lanceolatus* (currently *Bothrops lanceolatus*), a Brazilian snake in the Berlin Aquarium. Sprent (1988. Sys. Parasit. 11:165–213) divided the genus into five groups of species, Group 5 (arndti group) containing two species found in South American crotalines and colubrids, *O. arndti* and *O. sicki*, respectively. Hosts for *O. arndti* also include *Bothrops atrox* (Panama), and *B. cotiara* and *Crotalus durissus* (Brazil) (Sprent, *op. cit.*). Preliminary observations suggest that *O. arndti* is transmitted to crotalines by feeding on infected rodents (Sprent, *op. cit.*). *Porthidium hespere* represents a new host record for *O. arndti*. Mexico is a new locality record.

We thank Christopher I. Grünwald for collection of the *P. hespere*.

Submitted by **STEPHEN R. GOLDBERG,** Whittier College, Biology Department, Whittier, California 90608, USA (e-mail: SGoldberg@whittier.edu); **JACOBO REYES-VELASCO,** Calle del Parque 45, Colonia Chapalita, Zapopan, Jalisco 45042, México; **CHARLES R. BURSEY,** Pennsylvania State University, Shenango Campus, Department of Biology, Sharon, Pennsylvania 16146, USA; and **ERIC DUGAN,** Loma Linda University, Department of Earth and Biological Sciences, Loma Linda, California 92350, USA.

PSAMMOPHIS BISERIATUS (Link-marked Sand Snake). **DIET**. As part of a larger study of psammophiine diets, we dissected and measured specimens of the East African species *Psammophis biseriatus* (N = 25). The sex ratio of the sample was 48% female and 52% male. None of the 20 adults examined contained prey. However, three juvenile females from Kenya had prey remains in their stomachs. CAS 131093 (516 mm SVL, 311 mm TL, 35.1 g) from Kakuma (3.70°S, 34.85°E; map datum WGS84), CAS 134502 (533 mm SVL, tail broken, 42.2 g) from near Mandera (3.93°S, 41.85°E), and CAS 165565 (474 mm SVL, 263 mm TL, 24.9 g) from near Kibwezi (2.70°S, 38.62°E) contained a *Trachylepis striata* (Striped Skink), *Trachylepis brevicollis* (Short-necked Skink), and *Hemidactylus mabouia* (Tropical House Gecko), respectively. None of the prey items extracted was intact enough for accurate measuring and weighing and all were ingested head first.

Psammophis biseriatus is a diurnal, predominately arboreal snake described as an ambush forager (Spawls et al. 2002. A Field Guide to the Reptiles of East Africa. Academic Press, San Diego. 543 pp.). Its known distribution is in eastern Africa, from western Somalia, Eritrea, and eastern Ethiopia south through Kenya to Tanzania, where its distribution becomes discontinuous (Brandstätter 1996. Die Sandrennattern. Westarp-Wissenschaften, Magdeburg, 142 pp.; Spawls et al. 2002, *op. cit.*). This species has been reported to take mostly lizard prey; however, few specific dietary records exist. Previous prey records for *P. biseriatus* are *Latastia longicaudata revoili*, *Heliobolus spekii*, *Trachylepis quinquetaentia*, *T. planifrons*, and *Chamaeleo dilepis roperi* (Loveridge 1940. Bull. Mus. Comp. Zool. 87:1–61; Spawls et al. 2002, *op. cit.*). These new prey records emphasize that scincids are important food sources for *P. biseriatus*, as they are for African *Psammophis* in general (Akani et al. 2003. J. Herpetol. 37:208–211; Shine et al. 2006. Copeia 2006:650–664), and also provide the first evidence of gekkonid prey for the species.

Submitted by **AMANDA M. COTTONE** (e-mail: amanda.cottone@villanova.edu) and **AARON M. BAUER** (e-mail: aaron.bauer@villanova.edu), Department of Biology, Villanova University, 800 Lancaster Avenue, Villanova, Pennsylvania 19085, USA.

PSAMMOPHIS CRUCIFER (Cross-marked Grass Snake). **OPHIOPHAGY**. As part of a larger study of psammophine diets, we dissected specimens of the temperate southern African species *Psammophis crucifer.* The stomach of an adult female (TM 69360, 425 mm SVL, 125 mm TL, 19.0 g) from Malolotja Nature Reserve, Swaziland (26.07°S, 31.12°E; map datum WGS84) contained a largely intact *Duberria lutrix* (Slug Eater) (186 mm SVL, 17 mm TL, 5.2 g [relative prey mass 27.2%]) that had been ingested head first. A portion of an unidentified lizard tail was found posterior to the *Duberria*. In addition, the hind gut of an adult male *P. crucifer* (PEM 974, 392 mm SVL, 168 mm TL, 20.2 g) from Port Elizabeth, Eastern Cape, South Africa (33.92°S, 25.57°E) contained the highly digested remains of a small snake that had been ingested head first. The color pattern of the remaining intact scales suggested that the prey was a *Psammophis* species, but the remains were too fragmentary to make a positive identification.

Psammophis crucifer is a habitat generalist that can be found in lowland and montane forest, moist savanna, grassland, and fynbos of southern Africa from the southwestern Cape, to the escarpment of Mpumalanga (Branch 1998. A Field Guide to Snakes and Other Reptiles of Southern Africa. Struik Publishing, Capetown, South Africa. 368 pp.). In Swaziland it co-occurs with *Duberria lutrix* in the highveld and western middleveld (Boycott 1992. A Herpetofaunal Survey of Swaziland. M.Sc. thesis, University of Natal, Durban, South Africa. v + [7] +175 pp., 43 figs.; Boycott and Culverwell 1992. J. Herpetol. Assoc. Afr. 40:38–41). It has been reported to feed chiefly on lizards, especially geckos, and frogs (Broadley 1983. FitzSimon's Snakes of Southern Africa, 3rd ed. Jonathan Ball and Ad. Donker Publishers, Parklands, South Africa. 387 pp.). The diet of most *Psammophis* species comprises predominantly lizards, frogs, or small mammals (Shine et al. 2006. Copeia 2006:650–664.) but ophiophagy has been reported in *P. phillipsi* (Haagner 1990. J. Herpetol. Assoc. Afr. 37:47; Luiselli et al. 2004. Amphibia-Reptilia 25:415–423), and the closely related *P. mossambicus* (Bruton and Haacke 1980. *In* Bruton and Cooper [eds.], Studies on the Ecology of Maputaland, pp. 251–287. Rhodes University, Grahamstown, South Africa; Broadley et al. 2003. Snakes of Zambia. An Atlas and Field Guide. Edition Chimaira, Frankfurt, Germany. 280 pp.; Shine et al., *op. cit.*), both mainly tropical in their distribution and considerably larger in size than *P. crucifer*. This record represents the first record of ophiophagy in *P. crucifer*, or any temperate southern African psammophiine.

Submitted by **AMANDA M. COTTONE** (e-mail: amanda.cottone@villanova.edu) and **AARON M. BAUER** (e-mail: aaron.bauer@villanova.edu), Department of Biology, Villanova University, 800 Lancaster Avenue, Villanova, Pennsylvania 19085, USA.

PTYAS MUCOSA (Oriental Rat Snake). **FORAGING BEHAVIOR.** *Bufo bankorensis* (Bankor Toad) is the most abundant anuran species in Da-Jiou-Shi Experimental Forest (Mao, unpubl.) and appears in the diet of *Ptyas mucosa* in Taiwan (Lin et al. 1996. NOW 3:19–21 [in Chinese]).

On 20 June 2006 at 1105 h a *P. mucosa* (ca. 1800 mm TL) was observed subduing a *B. bankorensis* on the northwestern side of a bridge over Orchid Stream (24.45722ºN, 121.67888ºE, 286 m elev.). When we approached the snake it released the toad and escaped. The toad had inflated its body and was covered in blood. Although still alive, the toad appeared to be paralyzed. Forty minutes later, when we returned to the site, we found the same snake with the same toad in its jaws. Our presence disturbed the snake and it again immediately released the toad and escaped into the nearby forest. At 1300 h we returned to the site again, and found the snake once again had the toad in its jaws. This time the snake carried the toad to the southeastern side of the stream, where it then released the toad and fled. At 1400 h when we checked for the last time, we found that the snake had returned and had almost completely ingested the toad.

Prey retrieval behavior is common in predatory birds, especially some raptors (Collopy 1977. Condor 79:63–68). In ophidians, some crotaline species will retrieve their prey after it has been envenomated (Diller 1990. J. Herpetol. 24:95–97). But in our review of the literature, we found no reports describing colubrid snakes retrieving prey they had previously abandoned. The persistence of the snake is especially interesting considering that the predation process was thrice interrupted by potential predators.

Submitted by **JEAN-JAY MAO**, Department of Natural Resources, National Ilan University. No. 1, Sec. 1, Shen-Lung Rd., Ilan, Taiwan 260 (e-mail: jjmao@niu.edu.tw); **GERRUT NORVAL**, Applied Behavioural Ecology & Ecosystem Research Unit, Department of Nature Conservation, UNISA, Private Bag X6, Florida, 1710, Republic of South Africa; and **LAN-WEI YEH**, Department of Natural Resources, National Ilan University.

RHABDOPHIS SWINHONIS (Taiwanese Keel-back). **DIET**. *Rhabdophis swinhonis* is a diurnal, small to medium sized snake, endemic to Taiwan. Because of its cryptic behavior, little is known of its ecology, behavior, or diet (Lue et al. 1989. Taiwan Snakes. Taiwan Provincial Dept. of Education, Taiwan, Republic of China, 94 pp. [in Chinese]). On 22 April 2006 at 1422 h in Huban District (Yunlin County) we found a *R. swinhonis* (ca. 600 mm TL) ingesting a large earthworm by the side of a trail (23.72194ºN, 120.62444ºE; 191 m elev.). To the best of our knowledge, this is the first reported prey for *R. swinhonis*.

Submitted by **JEAN-JAY MAO**, Department of Natural Resources, National Ilan University. No. 1, Sec. 1, Shen-Lung Rd., Ilan, Taiwan 260 (e-mail: jjmao@niu.edu.tw); **GERRUT NORVAL**, Applied Behavioural Ecology and Ecosystem Research Unit, Department of Nature Conservation, UNISA, Private Bag X6, Florida, 1710, Republic of South Africa; and **HSU-LIANG TSENG** and **I-SHUO TSENG**, Minshiang Environmental & Ecological Research CO., Ltd.

RHINOBOTHRYUM BOVALLII (NCN). **REPRODUCTION.** *Rhinobothryum bovallii* is a large, slender colubrid restricted to Atlantic lowlands from southern Honduras to western Venezuela and to the Pacific lowlands from central western Panama to western Ecuador (4–550 m elev.) (Savage 2002. The Amphibians and Reptiles of Costa Rica: A Herpetofauna between Two Continents, between Two Seas. Univ. Chicago Press, Chicago, Illinois. 934 pp.). There are no published accounts of reproduction in either of the two species of *Rhinobothryum*.

Here we report on two clutches of eggs laid at the Fort Worth Zoo, Fort Worth, Texas. All of the parental snakes were captured prior to September 2000 from Guayacán de Siquirres, Limon Province, Costa Rica. Three eggs were laid by a female (1320 mm SVL, 282.0 g) on 10 February 2006. The eggs were set up in a 1.5:1 ratio of vermiculite to water at a constant 29.4ºC. Two infertile eggs were discarded on 15 and 23 February 2006. The single fertile egg (52 mm long, 18.1 mm wide, 11.7 g) was opened on 25 April 2006 revealing a seemingly fully developed dead snake (260 mm SVL, 80 mm tail, 11.4 g; University of Texas at Arlington Amphibian and Reptile Diversity Research Center, UTA R 54423) with kinked vertebrae.

A second clutch of 2 eggs (53.4 and 52.1 mm long, 17 and 17.4 mm wide, and 9.5 and 9.8 g respectively) was laid by another female (1142 mm SVL, 182.0 g) on 1 June 2006. Because high incubation temperatures often result in kinked vertebrae, this clutch was incubated on a shelf in a cooler section of the herpetarium in a substrate of 2:1 vermiculite to water by weight. Temperatures varied from a daytime high of ca. 26.6ºC to a nighttime low of ca. 21.1ºC. The morning of 26 September 2006 one egg was manually slit revealing a fully developed live neonate (307 mm SVL, 78 mm tail, 9.8 g). The second neonate (330 mm SVL, 89 mm tail, 10.0 g) hatched without assistance later the same day. All neonates had the same pattern and coloration as the adults, and both surviving neonates fed on juvenile *Anolis carolinensis* after their first ecdysis.

Submitted by **STEPHEN HAMMACK** and **ANDREW BRINKER**, Department of Herpetology, Fort Worth Zoo, 1989 Colonial Parkway, Fort Worth, Texas 76110, USA (e-mail: herp@fortworthzoo.org).

STORERIA OCCIPITOMACULATA OCCIPITOMACULATA (Northern Red-bellied Snake). **MAXIMUM SIZE**. On 2 June 2007, a gravid female *Storeria o. occipitomaculata* measuring 344 mm SVL and 422 mm TL, and weighing 18 g, was found beneath a flat rock on a wooded slope along the east side of US Hwy 62 (41.8148234°N, 79.2723545°W, WGS 84), Warren County, Pennsylvania, USA. This individual exceeds the maximum length record of 406 mm TL reported by Wright and Wright (1957. Handbook of Snakes of the United States and Canada. Comstock Publishing Associates, Ithaca, New York.). On 21 July 2007, this specimen produced a litter of 12 young, three of which were unable to escape from their egg membranes and perished. The litter size is well below the maximum of 21 reported for Pennsylvania *S. o. occipitomaculata* by Hulse et al. (2001. Amphibians and Reptiles of Pennsylvania and the Northeast. Cornell University Press, Ithaca, New York. 419 pp.). Size of the neonates (mean SVL = 71.0, SD = 7.8, range 52–76; mean TL = 91.0, SD = 9.0, range 69–97; N = 12) was slightly below the mean of 73.8 mm for SVL and 96.7 mm for TL reported by Hulse et al. (2001, *op. cit.*). The litter weighed 4 g; postpartum weight of the female was 14 g. The relative clutch mass (RCM) was 0.28, which is lower than the reported mean of 0.60 for this species (Ernst and Ernst. 2003. Snakes of the United States and Canada. Smithsonian Institution Press, Washington, D.C. 668 pp.).

Specimens deposited in Tom Ridge Environmental Center Natural History Collections at Presque Isle State Park (TREC R-031, TREC R-032). Material that was not mounted has been deposited in the Sternberg Museum of Natural History, Fort Hays University, Hays, Kansas. We thank J. T. Collins and J. Beane for encouragement and assistance.

Submitted by **BRIAN S. GRAY,** 1217 Clifton Drive, Erie, Pennsylvania 16505, USA (e-mail: brachystoma@hotmail.com); and **MARK LETHABY,** 535 East 29th Street, Erie, Pennsylvania 16504, USA (e-mail: mlethaby@verizon.net).

THAMNOPHIS BRACHYSTOMA (Short-headed Gartersnake). **PREDATION**. *Thamnophis brachystoma* has a very limited distribution that includes the Allegheny drainage of the Allegheny High Plateau from extreme southwestern New York to northwestern Pennsylvania (Asplund 1963. Herpetologica 19:128–132) and is listed as a species of conservation concern in Pennsylvania. Herein we describe predation on *T. brachystoma* by small mammals, including *Blarina brevicauda* (Northern Short-tailed Shrew) and/or *Microtus pennsylvanicus* (Meadow Vole).

During spring and summer 2005 we recorded vertebrates encountered under an array of 24 sheets of plywood and ca. 34 m^2 of corrugated asphalt (Ondura brand) boards. The coverboard array was located in DuBois, Clearfield County, Pennsylvania (USA) in a seasonally flooded wet meadow dominated by grass and forbs. The coverboard array was checked a total of 38 times during the period 13 April – 19 September at least once a week. On 4 July, 9 September, and 19 September boards were checked twice each day. The three most common vertebrates under the boards in order of occurrence were *T. brachystoma* (N = 183), *M. pennsylvanicus* (N = 81), and *Thamnophis sirtalis* (Eastern Garter Snake; N = 22). *B. brevicauda* (N = 9), *Condylura cristata* (Star-nosed Mole; N = 9), and *Diadophis punctatus* (Ring-necked Snake; N = 1) were also observed. Numerous grass nests were found under the plywood boards and five litters of young *M. pennsylvanicus* were recorded.

On five occasions we found dead *T. brachystoma* under coverboards. In two cases the snakes' head and neck were missing, with the most anterior part of the spine exposed and soft tissue removed, as if stripped off by a predator. These snakes had 38 cm and 33 cm of body and tail length remaining, indicating they were adults. A third dead snake was a 15 cm long juvenile with head and body intact but signs of mutilation on the head and body. The largest dead snake (38 cm remaining) was found with the anterior spine dangling down into a soil crack. Soil cracks were common under the plywood coverboards and snakes and small mammals escaped into them when the boards were lifted. On 23 Sept 2004 (prior to the start of our formal surveys) we observed a dead *T. brachystoma* under the same board as a large food cache of seven earthworms (dead, dying, partially paralyzed or eaten) and snail shells. Although the head was missing, 21 cm of the snake's body remained. A female *B. brevicauda* was captured under the same coverboard in close proximity to the cache two days prior. On 28 Sept 2004 the posterior portion (6 cm) of another freshly killed *T. brachystoma* was found under the same coverboard. A *B. brevicauda* was also under the coverboard in an extensive grass nest (ca. 30 cm in diameter). On four occasions *T. brachystoma* and *M. pennsylvanicus* were found under a plywood board at the same time. The snakes and voles were found from 25 cm to 46 cm apart, and lifting the board caused one or both of the animals to flee for cover.

Small mammals, birds of prey (Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Books, London. 668 pp.), and other reptiles (Rossi and Rossi 2003. Snakes of the United States and Canada: Natural History and Care in Captivity. Krieger Publishing Co., Malabar, Florida. 520 pp.) are reported as potential predators of *T. brachystoma*. However, we were unable to find other reports of predation by specific small mammals on this species. The close association of the garter snakes and *M. pennsylvanicus* and *B. brevicauda* under the coverboards and repeated observations of mutilated *T. brachystoma* under the boards suggests that one or both of these mammals is killing these snakes.

Submitted by **JOSEPH W. HUMMER** and **KEELY TOLLEY ROEN**, Pennsylvania State University, DuBois Campus, 1 College Place, DuBois, Pennsylvania, USA (e-mail: kat175@psu.edu).

THAMNOPHIS EQUES (Mexican Gartersnake). **ENDOPARASITISM.** *Thamnophis eques* ranges from the vicinity of the Pico de Orizaba northwest through Sonora and Chihuahua, Mexico to Arizona and New Mexico in the United States (Conant 2003. Amer. Mus. Novit. 3406:1–64). On 15 October 2006 we found an adult male *T. eques* (620 mm SVL, 820 mm TL; Colección Nacional de Anfibios y Reptiles, Instituto de Biología, Universidad Nacional Autónoma de México, CNAR 12229) dead on the bank of Laguna de Yuriria, Guanajuato, Mexico (20.2777°N, 101.1111°W). The snake's mid-body was swollen and upon dissection we found two fourth-stage larvae of *Eustrongylides* sp. in the mesentery and some lesions in the surrounding tissues. Both nematodes were alive, red in color, and measured 97 mm and 124 mm in length (Colección Nacional de Helmintos, Instituto de Biología, Universidad Nacional Autónoma de México, CNHE 5599).

Larval forms of *Eustrongylides* sp. have been reported from *Nerodia sipedon* in Accomack County, Virginia, USA (Bursey 1986. J. Wildl. Dis. 22:527–532), from *T. eques* in Lago de Pátzcuaro, Mexico, and *T. melanogaster* in Pátzcuaro and Cuitzeo Michoacán lagoons, Mexico (Jiménez et al. 2002. J. Parasitol. 88:454–460). Death of captive *T. sirtalis,* with swellings in the body due to infection by *Eustrongylides* sp., has been reported (Lichtenfels and Lavies 1976. Lab. Anim. Sci. 26:465–467). *Eustrongylides* spp. adults have been reported from birds throughout much of the world and are associated with disease and even death in many avian species (Franson and Custer 1994. Colon. Waterbirds 17:168–172; Wiese et al. 1977. J. Wildl. Dis. 13:376–382). The life cycle of this genus of nematodes includes freshwater oligochaetes as first intermediate hosts, fish as second intermediate hosts, and piscivorous birds as definitive hosts. Amphibians and reptiles have been recorded as paratenic hosts (Cooper et al. 1978. J. Parasitol. 64:102–107; Yamaguti 1961. Systema Helminthum. Vol III. The Nematodes of Vertebrates. Interscience Publishers, Inc. New York. 669 pp.). In this context, our observation raises the possibility that infection of *T. eques* by *Eustrongylides* sp. larvae might be a cause of mortality in some wild populations.

Submitted by **ELISA CABRERA GUZMÁN,** Colección Nacional de Anfibios y Reptiles, Instituto de Biología, Departamento de Zoología, Universidad Nacional Autónoma de México, Circuito exterior, Ciudad Universitaria, México D. F. C.P. 04510; e-mail: anfisbenido@yahoo.com.

TRIMORPHODON VILKINSONII (Chihuahuan Lyre Snake). **ARBOREAL BEHAVIOR**. *Trimorphodon vilkinsonii* is a secretive snake that ranges throughout the central and western regions of the Chihuahuan Desert. This species has primarily been encountered in rock piles, rock outcroppings, and talus slopes (Werler and Dixon 2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press). *Trimorphodon biscutatus* is known to be arboreal (Scott and McDiarmid 1984. Cat. Amer. Amph. Rep. 353.1–353.4), as is *T. lambda* (Lowe 1964. The Vertebrates of Arizona. University of Arizona Press, Tucson); however, arboreality has yet to be noted in *T. vilkinsonii*. Here we report on two separate instances of aboreality in this species.

We found a female *T. vilkinsonii* (TNHC 66487; 591 mm SVL, 115 mm TL, 37.4 g) on 5 May 2007 at 2143 h in Cottonwood Canyon of the Sierra Vieja, Presidio County, Texas (30.54739°N, 104.67831°W, 1483 m elev.). This snake was climbing one of the main trunks of a large Gray Oak (*Quercus grisea*) and was 3.35 m above the ground. The cluster of oaks was located where the canyon bottom and rocky talus slope meet. A second female *T. vilkinsoni* (TNHC 66513; 715 mm SVL, 131 mm TL, 88 g) was found on 29 June 2007 at 2230 h in Box Canyon, a drainage on the eastern side of the Sierra Vieja, Presidio County, Texas (30.55059°N, 104.65919°W, 1378 m elev.). The female was found crawling among the outer branches of a Catclaw Acacia (*Acacia gregii*) approximately 1 m above the ground and 2 m from the dry wash.

Both specimens were collected under Texas Parks and Wildlife Scientific Collecting Permit SPR-1097-912 to Travis J. LaDuc. We thank the Miller family for support and assistance.

Submitted by **DREW R. DAVIS**, Texas Natural Science Center, The University of Texas at Austin, 10100 N. Burnet Road, PRC 176/R4000, Austin, Texas 78758, USA; **BRYAN BOX,** 40 Edgewater Drive, Lakeside City, Texas 76308; USA; and **TOBY J. HIBBITTS**, Texas Cooperative Wildlife Collection, Department of Wildlife and Fisheries Sciences, Texas A&M University, College Station, Texas 77843-2258; USA.

GEOGRAPHIC DISTRIBUTION

Herpetological Review publishes brief notices of new geographic distribution records in order to make them available to the herpetological community in published form. Geographic distribution records are important to biologists in that they allow for a more precise determination of a species' range, and thereby permit a more significant interpretation of its biology.

These geographic distribution records will be accepted in a **standard format** only, and all authors *must* adhere to that format, as follows: SCIENTIFIC NAME, COMMON NAME (for the United States and Canada as it appears in Crother [ed.] 2008. *Scientific and Standard English Names of Amphibians and Reptiles of North America North of Mexico*. SSAR Herpetol. Circ. 37:1–84, available from SSAR Publications Secretary, ssar@herplit.com; for Mexico as it appears in Liner 1994, *Scientific and Common Names for the Amphibians and Reptiles of Mexico in English and Spanish.* Herpetol. Circ. 23:1–113), LOCALITY (use metric for distances and give precise locality data, including lat/long coordinates in **decimal degrees** and cite the map datum used), DATE (day-month-year), COLLECTOR, VERIFIED BY (*cannot* be verified by an author—curator at an institutional collection is preferred), PLACE OF DEPOSITION (where applicable, use standardized collection designations as they appear in Leviton et al. 1985, *Standard Symbolic Codes for Institutional Resource Collections in Herpetology and Ichthyology,* Copeia 1985[3]:802–832) and CATALOG NUMBER (required), COMMENTS (brief), CITATIONS (brief), SUBMITTED BY (give name and address in full—spell out state or province names—no abbreviations). If publishing specific locality information for a rare or endangered species has the potential to jeopardize that population, please consult with the Section Editor at time of record submission. If field work and/or specimen collection occurred where permits were required, please include permit number(s) and authorizing agency.

Some further comments. This geographic distribution section does not publish "observation" records. Records submitted should be based on preserved specimens which have been placed in a university or museum collection (private collection depository records are discouraged; institutional collection records will receive precedence in case of conflict). A good quality color slide or photograph may substitute for a preserved specimen *only* when the live specimen could not be collected for the following reasons: it was a protected species, it was found in a protected area, or the logistics of preservation were prohibitive (such as large turtles or crocodilians). Color slides and photographs *must* be deposited in a university or museum collection along with complete locality data, and the photographic catalog number(s) must be included in the same manner as a preserved record. Before you submit a manuscript to us, check Censky (1988, *Index to Geographic Distribution Records in Herpetological Review: 1967–1986*; available from the SSAR Publications Secretary) and other sources to make sure you are not duplicating a previously published record. The responsibility for checking literature for previously documented range extensions lies with authors. **Do not submit range extensions unless a thorough literature review has been completed.**

Please submit any geographic distribution records in the **standard format only** to one of the Section Co-editors: **Alan M. Richmond** (USA & Canadian records only); **Jerry D. Johnson** (Mexico and Central America, including the Caribbean Basin); **Indraneil Das** (all Old World records); or **Gustavo J. Scrocchi** (South American records). Short manuscripts are discouraged, and are only acceptable when data cannot be presented adequately in the standard format. **Electronic submission of manuscripts is required** (as Microsoft Word or Rich Text format [rtf] files, as e-mail attachments)**.** Refer to inside front cover for e-mail addresses of section editors.

Recommended citation for new distribution records appearing in this section is: Schmitz, A., and T. Ziegler. 2003. Geographic distribution. *Sphenomorphus rufocaudatus*. Herpetol. Rev. 34:385.

CAUDATA – SALAMANDERS

AMBYSTOMA MACULATUM (Spotted Salamander). USA: ARKANSAS: Desha Co.: 4.8 km SE Dumas off US 65 (Sec. 11, T10S, R4W). 12 April 2006. Nick Covington. Verified by S. E. Trauth. Arkansas State University Herpetological Museum (ASUMZ 30819). New county record filling a distributional gap between previous records in Lincoln (Robison 2006. Herpetol. Rev. 37:481) and Arkansas counties (Trauth et al. 2004. Amphibians and Reptiles of Arkansas. Univ. Arkansas Press, Fayetteville. 421 pp.).

Submitted by **HENRY W. ROBISON**, Department of Biology, Southern Arkansas University, Magnolia, Arkansas 71754, USA (e-mail: hwrobison@saumag.edu); and **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA (e-mail: cmcallister@csc.edu).

AMBYSTOMA MACULATUM (Spotted Salamander). USA: TENNESSEE: Weakley Co: Greenfield (36.2031667°N, 88.8884333°W). 14 November 2007. Joshua M. Hall. Verified by A. Floyd Scott. Austin Peay State University's David H. Snyder Museum of Zoology (APSU 18822 [color slide]). One adult specimen found beneath damp log. New county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Austin Peay State Univ. Misc. Publ. 12:1–94; Redmond and Scott 1996. Atlas of Amphibians in Tennessee. The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. http//www.apsu.edu/amatlas/).

Submitted by **JOSHUA M. HALL**, Union University, 4105 Caldwell Drive, Milan, Tennessee 38358, USA.

AMPHIUMA TRIDACTYLUM (Three-toed Amphiuma). USA: ARKANSAS: Praire Co.: Hazen (34.7776444°N, 91.5866444°W). 18 October 2007. Jeffery Hunter Sebern. Verified by Stanley E. Trauth. Arkansas State University Museum of Zoology Herpetological Collection (ASUMZ 30821). New county record (Trauth et al. 2004. The Amphibians and Reptiles of Arkansas. Univ. of Arkansas Press, Fayetteville. 421 pp.).

Submitted by **MICHAEL V. PLUMMER**, Department of Biology, Box 12251, Harding University, Searcy, Arkansas 72149, USA (e-mail: plummer@harding.edu), **SARAH G. JOHNSON**, and **JOHN R. JOHNSON,** 2401 Audley Bolton, Searcy, Arkansas 72143 USA (e-mail: scientificker@hotmail.com).

ANEIDES AENEUS (Green Salamander). USA: INDIANA: Perry Co.: Hoosier National Forest: 38.3 m W of Crawford County Line (38.12133°N, 86.46278°W). 14 November 2007. Angela Garcia. Verified by Chris Phillips, Illinois Natural History Survey (INHS 2007e). New county record (Minton 2001. Amphibians and Reptiles of Indiana. 2nd ed., revised. Indiana Academy of Science. vii–404 pp.) This observation is outside of Indiana's proposed range, suggesting an undiscovered westward projection of a population along the bluffs of the Ohio River. The salamander was captured by hand alongside a small crevice within a rock outcrop. *Aneides aeneus* is a state endangered species, previously known to occur in only two isolated populations in Crawford County.

Submitted by, **ANGELA GARCIA**, **NATHAN ENGBRECHT**, **JAMIE FALLER**, and**, ZACK WALKER**, Wildlife Diversity Section, Indiana Department of Natural Resources Division of Fish and Wildlife, 553 E. Miller Drive, Bloomington, Indiana 47401, USA (e-mail: zwalker@dnr.IN.gov).

HEMIDACTYLIUM SCUTATUM (Four-toed Salamander). USA: ARKANSAS: Perry Co.: 5.6 km NE Hollis, T3N, R19W, Sec18. 19 October 2007. Tobin Fulmer and Destiny Fulmer. Verified by Renn Tumlison. Henderson State University Museum of Zoology (HSUMZ 1412, 1413). Two adults were found under a log in a wooded area. New county record extends known range 24 km N of closest record (Saugey and Trauth 1991. Proc. Arkansas. Acad. of Sci. 45:88–91). A closer specimen, HSU 1086, 16.3 km S, is cataloged in the HSU collection.

Submitted by **TOBIN FULMER,** Arkansas Natural Heritage Commission, 1500 Tower Building, 323 Center Street, Little Rock, Arkansas 72201, USA; and **DESTINY FULMER**, 2305 Laverne Street, Bryant, Arkansas 72022 USA.

HEMIDACTYLIUM SCUTATUM (Four-toed Salamander). USA: INDIANA: Tippecanoe Co.: Brookston 7.5 minute USGS quad map; SW 1/4 of Sec. 21, T24N, R3W. 04 May 2005. Kyle W. Arvin. Verified by Christopher Phillips. Illinois Natural History Survey (INHS photo voucher 2007c). First record for county (Minton 2001. Amphibians and Reptiles of Indiana, rev. 2nd ed., Indiana Academy of Science, Indianapolis, Indiana).

First specimen found in upland woods under a log. On 06 May, 07 May and 08 May, 2005, additional specimens were found under logs in upland woods habitat by Kyle Arvin, Delano Arvin, and Rick Howard. On 14 May 2005, LAC found a female attending well-developed eggs in a clump of sedge and mosses. The nesting location was an open, spring-fed wetland dominated by clumps of sedges (*Carex* spp.), skunk cabbage (*Symplocarpus foetidus*), and marsh marigold (*Caltha palustris*). The wetland used for nesting was adjacent to the upland woods where the other salamanders were found.

Submitted by **LEE A. CASEBERE**, Indiana Department of Natural Resources, Division of Nature Preserves, 402 W. Washington Street, Rm. W-267, Indianapolis, Indiana 46204, USA (e-mail: Lcasebere@dnr.in.gov); and **KYLE W. ARVIN**, New York State Seed Testing Laboratory, New York State Agricultural Experiment Station, Cornell University, Sturtevant Hall, 630 West North Street, Geneva, New York 14456, USA (e-mail: kwa3@nysaes.cornell.edu).

NOTOPTHALMUS VIRIDESCENS (Eastern Newt). USA: ARKANSAS: Lonoke Co.: 2.2 km E Ward (T5N, R9W, S25). 3 April 2005. Donald F. McKenzie. Verified by Stanley E. Trauth. Arkansas State University Museum of Zoology Herpetological Collection (ASUMZ 30822). New county record (Trauth et al. 2004. The Amphibians and Reptiles of Arkansas. Univ. of Arkansas Press, Fayetteville. 421 pp.).

Submitted by **MICHAEL V. PLUMMER**, Department of Biology, Box 12251, Harding University, Searcy, Arkansas 72149, USA (e-mail: plummer@harding.edu); and **DONALD F. McKENZIE,** Wildlife Management Institute, 2396 Cocklebur Road, Ward, Arkansas 72176, USA (e-mail: wmidm@ipa.net).

NOTOPHTHALMUS VIRIDESCENS LOUISIANENSIS (Central Newt). USA: ARKANSAS: Lincoln Co.: county road 21 at Ten Mile Creek S of Cornerville (Sec. 27, T10S, R8W). 16 March 2006. Henry W. Robison. Verified by S. E. Trauth. Arkansas State University Herpetological Museum (ASUMZ 30820). New county record partially filling a distributional gap between Cleveland and Desha counties (Trauth et al. 2004. Amphibians and Reptiles of Arkansas. Univ. Arkansas Press, Fayetteville. 421 pp.).

Submitted by **HENRY W. ROBISON**, Department of Biology, Southern Arkansas University, Magnolia, Arkansas 71754, USA (e-mail: hwrobison@saumag.edu); and **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA (e-mail: cmcallister@csc.edu).

ANURA – FROGS

ACRIS CREPITANS (Northern Cricket Frog). USA: TENNESSEE: Haywood Co.: Hatchie National Wildlife Refuge, Hillville 35.48599°N, 89.19597°W. 20 May 2004. Jonathan W. Stanley. Verified by A. Floyd Scott. Austin Peay State University Museum of Zoology (APSU 18222, audio voucher). First county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12. The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 94 pp.).

Submitted by **JONATHAN W. STANLEY,** Department of Biological Sciences, Arkansas State University, State University, Arkansas 72467, USA; and **BRIAN P. BUTTERFIELD**, Department of Biology, Freed-Hardeman University, Henderson, Tennessee 38340, USA (e-mail: bbutterfield@fhu.edu).

ACRIS CREPITANS (Northern Cricket Frog). USA: TENNESSEE: Tipton Co.: Bear Creek at the junction of Bluff, Coon Valley, Herring, and Pryor roads in the Drummonds 7.5 Minute Quad (35.44682°N, 89.96216°W; NAD83). 17 July 2007. Joshua R. Ennen and Brian R. Kreiser. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 18806). One adult captured upstream from bridge in an isolated pool in the flood plain. First record for Tipton County (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12, The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 16 pp. Hard copy and Internet (http// www.apsu.edu/amatlas/) versions, the latter of which includes links to information on Tennessee amphibians that has appeared since 1996; accessed 1 November 2007).

The voucher specimen was collected by authors under authority of Tennessee Wildlife Resources Scientific Collecting Permit 1662 and deposited in Austin Peay State University's Museum of Zoology, APSU 18806.

Submitted by **JOSHUA R. ENNEN** (e-mail: Joshua.Ennen@usm.edu) and **BRIAN R. KREISER,** Department of Biological Science, University of Southern Mississippi, Hattiesburg, Mississippi 39406, USA (e-mail: brian.kreiser@usm.edu).

ANAXYRUS AMERICANUS (American Toad). USA: TENNESSEE: Knox Co.: Hardin Valley Area (35.94468°N, 084.20568°W). 06 August 2007. Ted M. Faust. Verified by A. Floyd Scott. Austin Peay State University Museum of Zoology (APSU 18815, color photo). One individual was found on the front porch of a house shortly after sunset at 2105 h. Air temperature 25°C. Two more individuals were found in a nearby flower garden within an hour and a half of the first individual. New county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12, The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 94 pp. Hard copy and Internet versions, the latter [http://www.apsu.edu/amatlas/ accessed 07 August 2007] including links to data on amphibians in Tennessee that have appeared since 1996).

Submitted by **TED M. FAUST**, 11828 Couch Mill Road, Knoxville, Tennessee 37932, USA; e-mail: tmfaust21@gmail.com.

ANAXYRUS FOWLERI (Fowler's Toad). USA: TENNESSEE: Morgan Co.: 5.4 km SW of Lancing (36.0941194°N, 84.703147°W). 11 May 2007. Ted M. Faust. Verified by A. Floyd Scott. Austin Peay State University Museum of Zoology (APSU 18814, color photo). One individual was found at the base of a bluff at 0940 h. A second individual was found under an overhanging bluff at 1045 h. New county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12, The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 94 pp. Hard copy and Internet versions, the latter [http://www.apsu.edu/amatlas/ accessed 07 August 2007] including links to data on amphibians in Tennessee that have appeared since 1996).

Submitted by **TED M. FAUST**, 11828 Couch Mill Road, Knoxville, Tennessee 37932, USA; e-mail: tmfaust21@gmail.com.

ELEUTHERODACTYLUS CYSTIGNATHOIDES (Rio Grand Chirping Frog). USA: LOUISIANA: Lafayette Parish: Zoo of Acadiana grounds (30.1171333°N, 91.93995°W). 19 April 2007. James W. Beck and Robert C. Dobbs. Color photograph verified by David Cannatella. TNHC 66880. This individual was singing from exotic grass ca. 1 m above the ground within a natural area among zoo buildings, at ca. 2100 h under humid conditions. This represents the first documented record for Lafayette Parish (Dundee and Rossman 1989. Amphibians and Reptiles of Louisiana, Louisiana State Univ. Press, Baton Rouge. 300 pp.), and only the second documented state record (Hardy 2004. Southwest. Nat. 49:263–268). This species is expanding its range to the north and east, presumably as a result of human activity, and is now established throughout much of southeastern Texas, north to Smith and Nacogdoches counties (Dixon 2000. Amphibians and Reptiles of Texas, 2nd ed. Texas A&M Univ. Press, College Station. 421 pp.; Hibbitts and Saenz 2007. Herpetol. Rev. 38:350), and in Caddo Parish in northwestern Louisiana (Hardy 2004, *op. cit*.).

Submitted by **JAMES W. BECK**, 100 Aurelien Loop #101, Broussard, Louisiana 70518, USA (e-mail: agkistrodon@worldnet.att.net); and **ROBERT C. DOBBS**, Department of Biology, Queen's University, Kingston, Ontario K7L 3N6, Canada (e-mail: 6rcd1@queensu.ca).

GASTROPHRYNE CAROLINENSIS (Eastern Narrow-mouthed Toad). USA: TENNESSEE: Gibson Co.: Milan (35.970314647°N, 88.714316025°W). 29 April 2007. Joshua M. Hall. Verified by A. Floyd Scott. Austin Peay State University's David H. Snyder Museum of Zoology (APSU 18801 [color slide]). One adult specimen found underneath damp log. New county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Austin Peay State Univ. Misc. Publ. 12:1–94; Redmond and Scott 1996. Atlas of Amphibians in Tennessee The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. http://www.apsu.edu/amatlas/).

Submitted by **JOSHUA M. HALL**, Union University, 4105 Caldwell Drive, Milan, Tennessee 38358, USA.

GASTROPHRYNE CAROLINENSIS (Eastern Narrow-mouthed Toad). USA: TENNESSEE: Haywood Co.: Hatchie National Wildlife Refuge, Hillville (35.48599°N, 89.19597°W) 20 May 2004. Jonathan W. Stanley. Verified by A. Floyd Scott. Austin Peay State University Museum of Zoology (APSU 18221, audio voucher). First county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12. The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 94 pp.).

Submitted by **JONATHAN W. STANLEY,** Department of Biological Sciences, Arkansas State University, State University, Arkansas 72467, USA; and **BRIAN P. BUTTERFIELD**, Department of Biology, Freed-Hardeman University, Henderson, Tennessee 38340, USA (e-mail: bbutterfield@fhu.edu).

HYLA AVIVOCA (Bird-voiced Treefrog). USA: TENNESSEE: Fayette Co.: Wolf River Wildlife Management Area, Mineral Slough (35.0292998°N, 89.2734998°W). 05 May 2007. J. Ream and E. Johansen. Verified by A. Floyd Scott. Austin Peay State University's David H. Snyder Museum of Zoology (APSU 18816). Voucher specimen collected by hand at 2200 h along the westernmost shore of a water tupelo/cypress slough adjacent to the Wolf River; many other individuals calling in vicinity. New county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12, The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 94 pp. Hard copy and Internet versions, the latter [http://www.apsu.edu/amatlas/ accessed 20 November 2007] including links to data on amphibians in Tennessee that have appeared since 1996).

Submitted by **JOSHUA TAYLOR REAM**, Center of Excellence for Field Biology, Austin Peay State University, Clarksville, Tennessee 37044, USA; e-mail: jream14@apsu.edu.

HYLA CINEREA (Green Treefrog). USA: TENNESSEE: Tipton Co.: Bear Creek at the junction of Bluff, Coon Valley, Herring, and Pryor roads in the Drummonds 7.5-minute quad (35.44682°N, 89.96216°W; NAD83). 17 July 2007. Joshua R. Ennen and Brian R. Kreiser. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 18806). One adult captured upstream from bridge in an isolated pool in the flood plain. First record for Tipton County (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Misc. Publ. No. 12, The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 19 pp. Hard copy and Internet (http//www.apsu.edu/amatlas/) versions, the latter of which includes links to information on Tennessee amphibians that has appeared since 1996; accessed 1 November 2007).

The voucher specimen was collected by authors under authority of Tennessee Wildlife Resources Scientific Collecting Permit 1662 and deposited in Austin Peay State University's Museum of Zoology, APSU 18805.

Submitted by **JOSHUA R. ENNEN** (e-mail: Joshua.Ennen@usm.edu) and **BRIAN R. KREISER,** Department of Biological Science, University of Southern Mississippi, Hattiesburg, Mississippi 39406, USA (e-mail: brian.kreiser@usm.edu).

HYLA GRATIOSA (Barking Treefrog). USA: GEORGIA: Athens-Clarke Co.: Whitehall Forest, off of Phoenix Rd. (33.8906°N, 83.3578333°W; WGS 84). 21 August 2007. Collected by Andrew Richard Ferreira. Verified by John Jensen. UGAMNH (GMNH 50083). New county record (Williamson and Moulis 1994. Distribution of Amphibians and Reptiles in Georgia. Savannah Sci. Mus. Spec. Publ. No. 3, Savannah, Georgia. 712 pp.; Mitchell. 2005. *In* M. Lannoo [ed.], Amphibian Declines: The Conservation Status of United States Species, pp. 455–456. University of California Press, Berkeley, California). An adult *H. gratiosa* was found in a plastic cattle tank which held ca. 10 cm of water. On 18 and 29 September 2007, three metamorphic individuals (Gosner Stage 42; Gosner 1960. Herpetologica 16:183–190) were dip-netted from the edge of a pond within Whitehall Forest, located 0.5 km from above specimen location (33.8886139°N, 83.3628°W; WGS 84). The adult and metamorphic individuals suggest a breeding population in Whitehall Forest, Athens-Clarke County, Georgia.

Submitted by **ANDREW R. FERREIRA**, D. B. Warnell School of Forestry and Natural Resources, University of Georgia, Athens, Georgia 30602, USA; e-mail: ferreiraa@warnell.uga.edu.

HYLA VERSICOLOR (Gray Treefrog). CANADA: ONTARIO: TIMISKAMING DISTRICT: South Lorrain Twp., southern terminus of Hwy 567 at Ontario Power Generation's Lower Notch Generating Station (47.1386°N, 79.4542°W; NAD27). 21 August 2007. Richard Burns. Royal Ontario Museum (ROM) 43033, digital photograph. Verified by Ross D. MacCulloch. One individual perched on a steel platform at the top of an 18 m metal tower at approximately 1300–1500 h (sunny, air temperature 20°C). First record for Timiskaming District, extending its documented range ca. 80 km ENE of the Sheppard Twp. location, ca. 98 km NNW of the location E of North Bay, and ca. 119 km NW of the location W of Deux-Rivières. Reduces the hiatus in NE Ontario 165 km to the NW between this record and an undocumented observation near Parting Lake (47.9703°N, 81.2825°W; NAD27), and another undocumented observation 225 km to the NNW at Nahma Bog, (48.9214°N, 80.8839°W; NAD27).

The documented northern range limit in northeastern Ontario is represented by specimens from the following three localities (west to east): Sudbury Dist., Sheppard Twp., near Sturgeon River (46.9315°N, 80.4555°W; NAD27), uncatalogued larvae identified by F. R. Cook, but material housed at Canadian Museum of Nature (formerly National Museum of Canada, NMC) – 1994; Nipissing Dist., 17 km E of North Bay on Hwy 17 (46.2671°N, 79.2481°W; NAD27), NMC 7067 – May 1963); and, Nipissing Dist., 21 km W of Deux-Rivières on Hwy 17 (46.2850°N, 78.5141°W; NAD27), NMC 28778 – September 1986).

Submitted by **WAYNE F. WELLER**, Ontario Power Generation – Hydro Business, 14000 Niagara Parkway, Niagara-on-the-Lake, Ontario L0S 1J0, Canada.

HYLOMANTIS GRANULOSA (Granular Leaf Frog). BRAZIL: BAHIA: Municipality of Amargosa, Timbó farm (13.1225°S, 40.7752778°W) 750 m elev. 15 April 2007. P. Fonseca. Museu de Zoologia da Universidade Federal da Bahia, Salvador, Brazil (MZUFBA 7.300, 7.301, 7.302). Collected at night on the ground of a cacao plantation. Verified by M. Napoli. Previously known from Recife, Pernambuco state, Brazil (Carnaval et al. 2003. Herpetol. Rev. 34:381; Cruz 1989 [1988] Arquivos da Universidade Federal Rural do Rio de Janeiro 11:41; Cruz 1991. Rev. Brasil. Biol. 50:720). First state record, extends distribution ca. 750 km S from Recife, Pernambuco state (Carnaval et al. 2003, *op. cit*).

Submitted by **MARCO ANTÔNIO DE FREITAS,** Programa de pós-graduação em zoologi.a, UESC (Universidade Estadual de Santa Cruz) CEP 46.500-000 Rodovia Ilhéus/Itabuna, Ilhéus, Bahia, Brazil (e-mail: philodryas@hotmail.com); **THAÍS FIGUEIREDO SANTOS SILVA,** Biogeographia publicações e consultoria ambiental, Rua E quadra D lote 11, Jardim Aeroporto, CEP 42700-000, Lauro de Freitas, Bahia, Brazil; **PATRÍCIA MENDES FONSECA** (e-mail: patriciamfonseca@yahoo.com.br) and **RAFAEL OLIVEIRA DE ABREU,** Instituto de Biologia, Museu de Zoologia da Universidade Federal da Bahia (UFBA) CEP 40000-000, Salvador, Bahia, Brazil (e-mail: rafaoabreu@gmail.com).

HYPSIBOAS ALBOPUNCTATUS (Spotted Treefrog). BOLIVIA: BENI: Los Lagos (12.78275°S, 65.8233833°W; 168 m elev.). 08 January 2007. M. Jansen, O. Helmig, and J. Q. Vidoz. Museo Noel Kempff Mercado, Santa Cruz, Bolivia (MNKR 8357), and Museum Senckenberg Frankfurt, Germany (SMF 86655) Specimens found calling at 2140 h in the vegetation 120 cm above ground at the border of an inundated forest. Verified by G. Köhler. Previously known from central, southern, and southeastern Brazil, northeastern Argentina (Provincia Corrientes), northern Uruguay, eastern Paraguay, and eastern Bolivia, Department of Santa Cruz (Frost 2007. Amphibian Species of the World: an Online Reference. Version 5.1 (10 October, 2007) at http://research.amnh.org/herpetology/amphibia/index.php). First departamental record, extending the range ca. 450 km NW from a previous record in the Noel Kempff National Park, Department of Santa Cruz, and ca. 290 km S from a previous record in the Department of Pando, both at the border to Brazil (Reichle 2007. Distribution, Diversity and Conservation Status of Bolivian Amphibians. Diss. Univ. Bonn, Germany. 182 pp.).

Submitted by **MARTIN JANSEN**, Forschungsinstitut und Naturmuseum Senckenberg, Senckenberganlage 25, 60325 Frankfurt am Main, Germany (e-mail: martin.jansen@gmx.net); **JULIAN Q. VIDOZ**, Estancia y Centro de Investigación Ecológica Caparú, Bolivia; and **OSWALDO HELMIG**, Museo de Historia Natural Noel Kempff Mercado, Santa Cruz, Bolivia.

PHYLLODYTES MACULOSUS (NCN). BRAZIL: BAHIA: Municipality of Porto Seguro, Estação Veracel (16.3881667°S, 40.1134444°W), 77 m elev. 22 July 2007. M. A. de Freitas and T. Figueiredo Santos Silva. Museu de Zoologia da Universidade Federal da Bahia, Salvador, Brazil (MZUFBA 7479). Collected at night on bromeliads. Verified by C. A. Gonçalves Cruz. Previously known from Bandeira Municipality, Minas Gerais, and Mascote Municipality, Bahia, Brazil (Cruz et al. 2006. Arquivos do Museu Nacional, Rio de Janeiro 64[4]:321–324). First record from Porto Seguro Municipality southern Bahia, Brazil, and extends distribution ca. 110 km S from Mascote municipality, Bahia (Cruz et al. 2006, *op. cit.*).

Submitted by **MARCO ANTÔNIO DE FREITAS,** Programa de pós-graduação em zoologia, UESC (Universidade Estadual de Santa Cruz) CEP 46.500-000 Rodovia Ilhéus/Itabuna, Ilhéus, Bahia, Brazil (e-mail: philodryas@hotmail.com); and **THAÍS FIGUEIREDO SANTOS SILVA,** Biogeographia publicações e consultoria ambiental, Rua E quadra D lote 11, Jardim Aeroporto, CEP 42700-000, Lauro de Freitas, Bahia, Brazil.

PHYSALAEMUS CUVIERI (Barker Frog). BOLIVIA: BENI: Los Lagos (12.7691167°S, 65.8153333°W; 153 m elev.). 10 January 2007 at 2230 h. M. Jansen, O. Helmig, and J. Q. Vidoz. Museo Noel Kempff Mercado, Santa Cruz, Bolivia (MNKA 8358), and Museum Senckenberg Frankfurt, Germany (SMF 86656). Verified by G. Köhler. Previously known from northeastern, central, and southern Brazil, Misiones and Entre Ríos in Argentina, eastern Paraguay, Department of Santa Cruz in Bolivia, and possibly lowlands of southern Venezuela (Frost 2007. Amphibian Species of the World: an Online Reference. Version 5.1, 10 October 2007 at http://research.amnh.org/herpetology/amphibia/index.php). First departmental record, and the most northwestern locality known in

Bolivia. Nearest known locality is ca. 450 km SSE in the Department Santa Cruz (Reichle 2007. Distribution, Diversity and Conservation Status of Bolivian Amphibians. Diss. Univ. Bonn, Germany. 182 pp.). The specimens were found calling from small puddles in the Cerrado savanna after heavy rain during the day and calls were recorded.

Submitted by **MARTIN JANSEN**, Forschungsinstitut und Naturmuseum Senckenberg, Senckenberganlage 25, 60325 Frankfurt am Main, Germany (e-mail: martin.jansen@gmx.net); **JULIÁN Q. VIDOZ** (e-mail: qvidoz@yahoo.com.ar) and **OSWALDO HELMIG** (e-mail: oswaldito_ha@hotmail.com) Museo de Historia Natural Noel Kempff Mercado, Avenida Irala 565 Casilla postal 2489 Santa Cruz, Bolivia.

PSEUDACRIS FERIARUM (Upland Chorus Frog). USA: TENNESSEE: Gibson Co.: Milan (35.9058°N, 88.7653°W). 24 February 2007. Joshua M. Hall. Verified by A. Floyd Scott. Austin Peay State University's David H. Snyder Museum of Zoology (APSU 18131 [color slide]). One adult male found calling and collected along with one clutch of eggs in roadside ditch. New county record (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Austin Peay State Univ. Misc. Publ. 12:1–94; Redmond and Scott 1996. Atlas of Amphibians in Tennessee. The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. http//www.apsu.edu/amatlas/).

Submitted by **JOSHUA M. HALL**, Union University, 4105 Caldwell Drive, Milan, Tennessee 38358, USA.

SCAPHIOPUS COUCHII (Couch's Spadefoot). USA: TEXAS: Kaufman Co.: Terrell (33.228°N, 96.4561667°W). August 1978. D. Schwab. Verified by A. Braswell. North Carolina Museum of Natural Sciences (NCSM 67561). New county record partially filling distribution gap just outside of eastern range limit among Ellis and Hunt Counties (Dixon 2000. Amphibians and Reptiles of Texas, 2nd ed. Texas A&M University Press, College Station, Texas, 421 pp.).

Submitted by **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA; e-mail: cmcallister@csc.edu.

TESTUDINES – TURTLES

CHELYDRA SERPENTINA (Snapping Turtle). USA: ALABAMA: Bibb Co.: Alabama Hwy 5, 5 km SW of Brent (32.8769444°N, 87.2238889°W). 23 April 2005. Peter V. Lindeman, Kathleen M. Ryan, and Denise I. Stetson. Juvenile specimen of ca. 100 mm midline carapace length observed and photographed crossing the road. Florida Museum of Natural History Herpetology Dept. photographic archive (UF 152443; two images). New county record (Mount 1975. The Reptiles and Amphibians of Alabama. Auburn Univ. Agric. Exp. Sta., Auburn, Alabama. 347 pp.).

Submitted by **PETER V. LINDEMAN**, Department of Biology and Health Services, 150 Cooper Hall, Edinboro University of Pennsylvania, Edinboro, Pennsylvania 16444, USA; e-mail plindeman@edinboro.edu.

CHELYDRA SERPENTINA SERPENTINA (Eastern Snapping Turtle). USA: ARKANSAS: Lincoln Co.: 3.2 km S Star City (Sec. 29, T9S, R7W). 10 November 1985. M. Evans. Verified by S. E. Trauth. Arkansas State University Herpetological Museum (ASUMZ 30825 [hatchling]). New county record completely filling all the eastern counties of the state (Trauth et al. 2004. Amphibians and Reptiles of Arkansas. Univ. Arkansas Press, Fayetteville. 421 pp.). This turtle has now been documented from 69 of 75 (92%) Arkansas counties.

Submitted by **HENRY W. ROBISON**, Department of Biology, Southern Arkansas University, Magnolia, Arkansas 71754, USA (e-mail: hwrobison@saumag.edu); and **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA (e-mail: cmcallister@csc.edu).

GRAPTEMYS NIGRINODA (Black-knobbed Map Turtle). USA: ALABAMA: Autauga Co.: Alabama River, Steele's Landing at southern end of Autauga County Rd. 1. (32.4304333°N, 86.8327389°W). 23 May 2007. Peter V. Lindeman. Verified by James Godwin. Three basking adult males photographed from close range at 30× with a Kowa spotting scope with built-in digital camera. Florida Museum of Natural History Herpetology Dept. photographic archive (UF 152444). New county record (Mount 1975. The Reptiles and Amphibians of Alabama. Auburn Univ. Agric. Exp. Sta., Auburn, Alabama. 347 pp.).

Submitted by **PETER V. LINDEMAN**, Department of Biology and Health Services, 150 Cooper Hall, Edinboro University of Pennsylvania, Edinboro, Pennsylvania 16444, USA; e-mail plindeman@edinboro.edu.

GRAPTEMYS PULCHRA (Alabama Map Turtle). USA: ALABAMA: Autauga Co.: Alabama River, 0.5 km downstream of Steele's Landing at southern end of Autauga County Rd. 1 (32.431811°N, 86.8381472°W). 23 May 2007. Peter V. Lindeman. Verified by James Godwin. Two basking adult females photographed from close range at 30× with a Kowa spotting scope with built-in digital camera. Florida Museum of Natural History Herpetology Dept. photographic archive (UF 152445). New county record (Mount 1975. The Reptiles and Amphibians of Alabama. Auburn Univ. Agric. Exp. Sta., Auburn, Alabama. 347 pp.). Perry Co.: Cahaba River 200 m S of crossing of County Road 49 near Heiberger (32.7741667°N, 87.2737389°W). 19 April 2007. Peter V. Lindeman. Verified by James Godwin. Basking juvenile female photographed from close range at 30× with a Kowa spotting scope with built-in digital camera. Florida Museum of Natural History Herpetology Dept. photographic archive (UF 152446). New county record (Mount 1975, *op. cit.)*

Submitted by **PETER V. LINDEMAN**, Department of Biology and Health Services, 150 Cooper Hall, Edinboro University of Pennsylvania, Edinboro, Pennsylvania 16444, USA; e-mail plindeman@edinboro.edu.

MESOCLEMMYS TUBERCULATA**.** BRAZIL: MARANHÃO: Municipalidade de Paulino Neves (2.6646389°S, 42.618388°W). 11 July 2005. A. Batistella and C. Pötter. Amphibian and Reptiles Collection, Instituto Nacional de Pesquisas da Amazônia - INPA, Manaus, Brazil (INPA-H 14842–4 collected in rainwater ponds between sand dunes and the sea). Verified by R. Bernhard. Species previously known from Brazilian States of Ceará, Piauí, and Bahia (Luederwaldt 1926. Rev. Mus. Paulista 14:404–468.; Bour and Pauler 1987. Mesogee 47:3–23; Bour and Zaher 2005. Pap. Avuls Zool. 45:295–311; Loebmann et al. 2006. Check List 2:32–33). First state record, extends the distribution 113 km SW from Luís Correia, Piauí, the previous closest record (Loebmann et al., *op. cit.*).

Submitted by **ALEXANDRE BATISTELLA** (e-mail: alexbatistella@yahoo. com.br), **CAROLINA PÖTTER** (e-mail: pottercarolina@yahoo.com.br), **LARISSA BARRETO** (e-mail: lara@ele.com.br), and **RICHARD C. VOGT,** Instituto Nacional de Pesquisas da Amazônia - INPA, Coleção de Anfíbios e Répteis, Av. André Araújo, nº. 2936, C.P. 478, Manaus, Amazonas, Brazil, CEP 69011-970 (e-mail: vogt@inpa.gov.br).

PSEUDEMYS CONCINNA (River Cooter). USA: ALABAMA: AUTAUGA CO.: Alabama River, 0.5 km downstream of Steele's Landing at southern end of Autauga Co. Rd. 1 (32.4318111°N, 86.8381472°W). 23 May 2007. Peter V. Lindeman. Verified by James Godwin. Three adults photographed from close range at 30× with a Kowa spotting scope with built-in digital camera. Florida Museum of Natural History Herpetology Dept. photographic archive (UF 152447). New county record (Mount 1975. The Reptiles and Amphibians of Alabama. Auburn Univ. Agric. Exp. Sta., Auburn, Alabama. 347 pp.). DALLAS CO.: Alabama River, 1 km upstream of Edmund Pettus Bridge in Selma (32.4062°N, 87.0149722°W). 23 May 2007. Peter V. Lindeman. Verified by James Godwin. Adult male photographed from close range at 30× with a Kowa spotting scope with built-in digital camera. Florida Museum of Natural History Herpetology Dept. photographic archive (UF 152448). New county record (Mount 1975, *op. cit.*)

Submitted by **PETER V. LINDEMAN**, Department of Biology and Health Services, 150 Cooper Hall, Edinboro University of Pennsylvania, Edinboro, Pennsylvania 16444, USA; e-mail plindeman@edinboro.edu.

STERNOTHERUS ODORATUS (Eastern Musk Turtle). USA: WISCONSIN: MANITOWOC CO.: Pigeon Lake (43.9877778°N, 87.8763889°W). 25 July 2007. Stephen Surendonk. Photographs verified by Robert Hay (Wisconsin Department of Natural Resources-Bureau of Endangered Resources) and Gary Casper. Illinois Natural History Survey catalogue number: INHS 2007d. New county record. Extends range of species in Wisconsin (Casper 1996. Geographic Distributions of the Amphibians and Reptiles of Wisconsin. Milwaukee Public Museum, 87 pp.). One individual captured in a fyke net.

Submitted by **STEPHEN SURENDONK**, Wisconsin Department of Natural Resources, 2220 E. County Highway V, Mishicot, Wisconsin 54228, USA; and **JOSHUA M. KAPFER**, Natural Resources Consulting, Inc., 118 Main St, PO Box 128, Cottage Grove, Wisconsin 53527, USA.

TERRAPENE ORNATA LUTEOLA (Desert Box Turtle). USA: ARIZONA: PINAL CO.: Gila River floodplain, ca 4.2 km (airline) W and 0.2 km (airline) N Gila Co. line at Hwy 177 (W of Hayden), 33.0058°N, 110.8402°W; NAD27; 571 m elev. 23 July 2004. N. Olsker, H. Messing. UAZ 56315-PSV. Within 60 m of San Pedro River, Old PZ Ranch site, S of Dudleyville, (32.8743°N, 110.7224°W; NAD27), 625 m elev. 29 September 2004. R. Llewellyn. UAZ 56302-PSV. Alongside road, ca 0.6 km (airline) SSE Cook's Lake, 32.8546°N, 110.7160°W; NAD27; 657 m elev. 10 August 2003. D. Hall. UAZ 56325-PSV. E side San Pedro River, 9.7 km (by River Rd) SSE Copper Creek Rd (32.6642°N, 110.5930°W; NAD2), 738 m elev. 21 July 2005. R. Bezy, K. Bezy, K. Bolles. UAZ 56301-PSV. ARIZONA: COCHISE CO.: San Pedro River Valley, ca 3.3 mi (by Cascabel Rd) SE Cascabel (32.2617°N, 110.3348°W; NAD27), 976 m elev. 22 July 2002. D. Hall. UAZ 56330-PSV. All verified by George Bradley.

These include the first records for Pinal County and extend the range 120 km N from the Whetstone Mts., Cochise County (Turner et al. 2003. Southwest. Nat. 48:347–355) and 40 km NW from Aravaipa Creek and the Bonita grasslands, Graham Co. (Nickerson and Mays 1970. Trans. Kansas Acad. Sci. 72:492–505). The scattering of these records along a 90 km stretch of the lower San Pedro River north to the Gila River floodplain suggests that the populations found in this riparian corridor within the Sonoran Desert are native rather than introduced (released captives).

Submitted by **HENRY MESSING** (e-mail: hmessing@lc.usbr.gov) and **NICHOLE OLSKER** (e-mail: nmolsker@lc.usbr.gov), U.S. Bureau of Reclamation, 6150 West Thunderbird Road, Glendale, Arizona 85306-4014, USA; **ROBIN LLEWELLYN**, 2219 E. Eastland Street, Tucson, Arizona 85719, USA (e-mail: robinia2@msn.com); **DAVID HALL**, 603 N. 3rd Avenue, Tucson, Arizona 85705, USA (e-mail: azkiso@msn.com); **KIT B. BEZY**, Northern Arizona University, Flagstaff, Arizona 86001, USA (e-mail: kbb25@nau.edu); **KATHRYN BOLLES** (e-mail: k_bolles@comcast.net) and **ROBERT L. BEZY** (e-mail: bezy@comcast.net), Natural History Museum of Los Angeles County, Los Angeles, California 90007, USA; and **PHILIP C. ROSEN,** School of Natural Resources, University of Arizona, Tucson, Arizona 85721, USA (e-mail: pcrosen@u.arizona.edu).

TRACHEMYS ADIUTRIX. BRAZIL: PIAUÍ: Lake Sobradinho, Municipalidade de Luis Correia (2.9599444°S, 41.5704444°W). 15 November 2005. A. Rocha Araujo. Amphibian and Reptile Collection, Instituto Nacional de Pesquisas da Amazônia - INPA, Manaus, Brazil (INPA-H 16051– 6). Collected in rainwater-filled ponds between sand dunes and the sea. Verified by R. Bernhard. Previously known only from the type locality: Santo Amaro, Maranhão State, Brazil (Vanzolini 1995. Rev. Brasil. Biol. 55:111–125). First state record, extends the distribution 191 km E from Santo Amaro (Vanzolini, *op. cit.*).

Submitted by **ALEXANDRE BATISTELLA** (e-mail: alexbatistella@yahoo. com.br), **CAROLINA PÖTTER** (e-mail: pottercarolina@yahoo.com.br), and **RICHARD C. VOGT** Instituto Nacional de Pesquisas da Amazônia - INPA, Coleção de Anfíbios e Répteis, Av. André Araújo, no. 2936, C.P. 478, Manaus, Amazonas, Brazil, CEP 69011-970 (e-mail: vogt@inpa.gov.br).

SQUAMATA –LIZARDS

ACANTHOSAURA LEPIDOGASTER (Scale-bellied Spiny Lizard). THAILAND: NAKHON RATCHASIMA PROVINCE: PAK CHONG DISTRICT: Khao Yai National Park, nr. Lamtakong Stream (14.4322222°N, 101.4094444°E) 670 m elev. 09 October 2007.

M. Cota. Thailand Natural History Museum (THNHM-image/071002). Verified by T. Chan-ard. Previously known from northern Thailand and an isolated location in northern peninsular Thailand. Southeastern most record in Thailand, extending known range 230 km S from its nearest previous locality in Phu Khieo National Park, Khon San District, Chaiyaphum Province (Nabhitabhata et al. 2000. Checklist of Amphibians and Reptiles in Thailand. OEPP, Bangkok. 152 pp.; Nabhitabhata and Chan-ard 2005. Thailand Red Data: Mammals, Reptiles and Amphibians. OEPP, Bangkok. 234 pp.). This report represents the sole record in Thailand east of the Chao Phraya River basin and the flood plain region.

Submitted by **MICHAEL COTA**, Thailand Natural History Museum, National Science Museum, Technopolis, Khlong 5, Khlong Luang, Pathum Thani 12120 Thailand; e-mail: Herpetologe@gmail.com.

ELGARIA COERULEA PRINCIPIS (Northwestern Alligator Lizard). USA: IDAHO: Valley Co.: A single individual was observed ca. 4.6 km NE of Edwardsburg along the Big Creek Trail about 200–400 m downstream of the trailhead in the Frank Church River of No Return Wilderness, Payette National Forest (45.1538°N, 115.2935°W; NAD 83). 24 July 2006. Javan Bauder, Ashley Lange, and Brad Tucker. Color voucher photo deposited at the Idaho Museum of Natural History (IMNH 1923). Verified by Charles R. Peterson. Individual was crawling across dirt trail towards a talus slope at ca. 1700 h with sunny skies and 40–50% cloud cover. Had lost all but ca. 3 cm of its tail, the last ca. 1 cm of which appeared to be regrowth. New county record for this species, which extends the range ca. 105 km S of nearest unpublished observation (Idaho Co., Idaho; Bill Bosworth, Idaho Conservation Data Center, Idaho Department of Fish and Game, pers. comm.). Nearest published observations are ca. 195 and 225 km NW in Latah Co., Idaho (Nussbaum et al. 1983. Amphibians and Reptiles of the Pacific Northwest. University of Idaho Press, Moscow, Idaho; Cossel 1997. *Elgaria coerulea*. Digital Atlas of Idaho, http://imnh.isu.edu/digitalatlas/ bio/reptile /main/ repfram.htm, respectively).

Submitted by **JAVAN BAUDER**, Department of Fish and Wildlife Resources, University of Idaho, Moscow, Idaho 83844, USA. Current addres: Department of Biological Sciences, Idaho State University, Pocatello, Idaho 83209, USA; e-mail: baudjava@isu.edu.

HELIOBOLUS NITIDUS NITIDUS (Günther's Desert Lacertid). NIGER: Koure village (13.3433333°N, 02.57515°E; 408 m elev.). L. Chirio and I. Ineich. MNHN 2007.0038–51. Verified by Patrick David. First country record. Extends known range of subspecies 200 km N and into a former country gap, and previously known from Ivory Coast (Rödel et al. 1997. Salamandra 33[4]:225–240 [as *H. n. nitida*]), unreported from Ghana, Togo (Tornier 1901. Arch. Naturgesch., Beiheft Festschrift für Eduard von Martens, pp. 63–88), Benin (Loveridge 1952. Bull. IFAN, Dakar, sér. A, 14:229–242), and Nigeria (Dunger 1967. The Nigerian Field 32[3]:117–132 [as *Eremias nitida*]). A second subspecies, *H. n. garambensis* is known from Cameroon (Monard 1951. Mém. l'IFAN, sér. Sci. Nat. 1:123–170), Central African Republic (Joger 1990. *In* Peters and Hutterer [eds.], Vertebrates in the Tropics. Museum Alexander Koenig, Bonn; Chirio and Ineich 2006. African J. Herpetol. 55[1]:23–59), and Democratic Republic of Congo (formerly Zaire) (Schmidt 1919. Bull. Amer. Mus. Nat. Hist. 39:385–624 [as *Eremias nitida*]). Papenfuss (1969. Wasmann J. Biol. 27[2]:249–325) considered the species as inhabiting west African savannas from Guinea to Cameroon, but distribution clearly extends more to the east.

Submitted by **IVAN INEICH** and **LAURENT CHIRIO**, Muséum national d'Histoire naturelle, Département de Systématique et Evolution, USM 602 (Reptiles – Taxonomie et Collections) CP 30, 25 rue Cuvier, 75231 Paris Cedex 05, France; e-mail: ineich@mnhn.fr.

HEMIDACTYLUS TURCICUS TURCICUS (Mediterranean Gecko). USA: TEXAS: Williamson Co.: Georgetown, Sun City, N of Berry Creek bridge on Del Webb Blvd. 10 May 2007, DOR. TNHC 66523 (JLC 7136). Georgetown, 4304 Miramar. 01 August 2007. TNHC 66524 (JLC 7141*)*. BELL CO. Residence wall on farm road 2484, 3.5 mi. S. of Texas 195. 31 July 2007. TNHC 66525 (JLC 7145*)*. Verified by Travis LaDuc. First county records; fills in records for central Texas. (Dixon 2000. Amphibians and Reptiles of Texas. 2nd Edition. Texas A&M Univ. Press, College Station, 421 pp.).

Submitted by **JAMES L. CHRISTIANSEN**, Texas Natural History Collections, Texas Natural Science Center, The University of Texas at Austin, Austin, Texas 78705, USA; and **RICHARD WOOLHEATER,** 136 Fox Home Lane, Georgetown, Texas 78633, USA.

HEMITHECONYX CAUDICINCTUS (Fat-tailed Gecko). MAURITANIA: GUIDIMAKA REGION: 2 km NW Mbalou village (15.0333333°N, 12.083333°W). 15 November 2003. Jean-François Trape and Georges Diatta. Institut de Recherche pour le Développement at Dakar (IRD TR-166). Verified by Ivan Ineich. First record for Mauritania. A large adult male measuring 205 mm (122 + 83). Found inside a termitarium, ca. 2 m high during a study of borreliosis, when examining the content of a hole by introducing a flexible tube and sucking out contents with a portable petrol-powered aspirator (Vial et al. 2006. Lancet 368:37–43). Most records are from Sudan or Guinea savannah regions, and only two reach the 15th parallel: at Nioro du Sahel (15.233333°N, 09.5833333°W) in Mali, and Yonoféré (15.2666667°N, 14.466666°W) in Senegal (Grandison 1956. Bull. IFAN. 18[A]:224–245; Cisse and Karns 1978. Bull. IFAN. 40:144–211).

Submitted by **JEAN-FRANÇOIS TRAPE** and **GEORGES DIATTA**, Institut de Recherche pour le Développement (IRD, UR 77), B.P. 1386, Dakar, Sénégal; e-mail: trape@ird.sn.

PHYSIGNATHUS COCINCINUS (Chinese Water Dragon). THAILAND: NONG KHAI PROVINCE: Sangkhom District: Sangkhom Forest Reserve/Research Center for Medicinal Plants and Spices, along Nam Prai Stream (18.08°N, 102.2105556°E), 223 m elev.). 17 October 2007. M. Cota. Thailand Natural History Museum (THNHM 12868). Verified by T. Chan-ard. Previously known from eastern Thailand and an isolated locality in Sakon Nakhon Province. Northwestern most record, extending known range 220 km WNW from nearest record in Sakon Nakhon Province and N > 350 km from eastern Thailand at its closest point in Nakhon Ratchasima Province (Nabhitabhata et al. Checklist of

Amphibians and Reptiles in Thailand. OEPP, Bangkok. 152 pp.; Nabhitabhata and Chan-ard 2005. Thailand Red Data: Mammals, Reptiles and Amphibians. OEPP, Bangkok. 234 pp.). This report places the species in the region that transitions from the Khorat Plateau to the mountainous region that forms the topography of northern Thailand. The majority of the Khorat Plateau does not possess suitable habitat and is used primarily for agriculture, but it is possible that there are other isolated populations in areas retaining forest cover and permanent streams, canals, or rivers.

Submitted by **MICHAEL COTA**, Thailand Natural History Museum, National Science Museum, Technopolis, Khlong 5, Khlong Luang, Pathum Thani 12120 Thailand; e-mail: Herpetologe@gmail.com.

SCINCELLA LATERALIS (Little Brown Skink). USA: ARKANSAS: Chicot Co.: 3.2 km N jct. St. Hwy. 8 in Eudora (Sec. 14, T18S, R2W). 18 May 2004. D. J. Monks. Verified by S. E. Trauth. Arkansas State University Herpetological Museum (ASUMZ 30818). New county record. This skink has now been documented for all 75 counties of the state (Trauth et al. 2004. Amphibians and Reptiles of Arkansas. Univ. Arkansas Press, Fayetteville. 421 pp.).

Submitted by **HENRY W. ROBISON**, Department of Biology, Southern Arkansas University, Magnolia, Arkansas 71754, USA (e-mail: hwrobison@saumag.edu); and **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA (e-mail: cmcallister@csc.edu).

SPHAERODACTYLUS ELEGANS ELEGANS (Ashy Gecko). USA: FLORIDA: Broward Co.: Pembroke Pines: 1031 NW 93rd Ave (26.0168833°N, 80.2695694°W; WGS84; elev. <1 m). 27 October 2007. Collected by Lisa A. Charlton. Verified by F. Wayne King, Florida Museum of Natural History, University of Florida. One neonate captured in Jackson sticky trap set to sample insects in an alee tree (*Blighia sapida*). UF 152418. New county record. Originally turned in to Division of Plant Industry (DPI), Florida Department of Agriculture and Consumer Services, Gainesville, Florida, on 31 October 2007; Entomology Log No. 8204. This nonindigenous gecko was first reported from Key West, Monroe County, Florida, more than 85 years ago (Stejneger 1922. Copeia [108]:56). Since that time the species has slowly expanded its range through other islands in the Florida Keys, and onto mainland Florida in Miami, Miami-Dade County (Carr 1940. Univ. Florida Publ. Biol. Sci. Ser. 3[1]:1–118; Krysko and Daniels 2005. Caribb. J. Sci. 41:28–36.). On 11 September 1937, a single *Sphaerodactylus e. elegans* was collected in Port Everglades, Broward County, Florida (UF 183) and on 26 January 1963, another collected from inside a house in Miami, Miami-Dade County, Florida (UF 84234). This latest specimen is the third known mainland record in the continental U.S.

Submitted by **LOUIS A. SOMMA**, Department of Zoology, PO Box 118525, 223 Bartram Hall, University of Florida, Gainesville, Florida 32611-8525, USA (e-mail: las@zoo.ufl.edu); and **KENNETH L. KRYSKO**, Division of Herpetology, Florida Museum of Natural History, Dickinson Hall, University of Florida, Gainesville, Florida 32611, USA (e-mail: kenneyk@flmnh.ufl.edu).

TROPIDOPHORUS ASSAMENSIS (Assamese Water Skink). INDIA: ASSAM: Barail Hill Range, Adakuchi Basti Nullah (24.993°N, 92.7424°E), 163 m elev. 1215 and 1300 h on 29 March 2007. One example (SVL 41.23 mm; BNHM 1783) and another (SVL 68.65 mm; in private collection, AD/BR 05) collected from under bryophyte-covered rocks of evergreen forest streambed. Third specimen photographed from Lakhicherra Nullah, Barail Wildlife Sanctuary (24.9473667°N, 92.7781833°E), 40 m elev., 1022 h on 2 April 2007, under rocks of slow-flowing stream. Verified by Indraneil Das. Previously known from Nengpui Wildlife Sanctuary of Mizoram (Pawar and Birand 2001. A Survey of Amphibians, Reptiles and Birds in Northeast India, CERC Technical Report #6, Centre for Ecological Research and Conservation, Mysore. 118 pp.). First record from Assam state and ca. 300 km N of previous locality.

Submitted by **ABHIJIT DAS**, Division of Herpetology, Aaranyak, Samanwoy Path, Survey, Beltola, Guwahati 28, Assam, India; e-mail: abhijit@aaranyak.org.

SQUAMATA – SNAKES

AGKISTRODON BILINEATUS (Cantil). MÉXICO: SONORA: Municipality of Rosario Tesopaco, 13.2 m N of Tesopaco on Hwy 12. (ca. 27.81°N, 109.35°W). 13 August 1991. Randall D. Babb, Peter J. Mayne, Adam B. Schwalbe, and Amy Haddock. Verified by Charles H. Lowe and Cecil R. Schwalbe. UAZ 56577-PSV (specimen given to the Centro De Ecológico de Sonora). Northernmost record for this species in México and extends the known range ca. 120 km NW from the closest known locality near Alamos, Sonora (Schwalbe and Lowe 2002. *In* Robichaux and Yetman [eds.], The Tropical Deciduous Forest of Alamos, pp. 172–199. Univ. Arizona Press, Tucson, Arizona). The adult male was DOR in an area of tropical dry forest.

Submitted by **RANDALL D. BABB**, Arizona Game and Fish Department, Region VI Office, 7200 E. University, Mesa, Arizona, 85207, USA (e-mail: rbabb@azgfd.gov); and **ERIC A. DUGAN**, Department of Earth and Biological Sciences, Loma Linda University, Loma Linda, California 91330-9303, USA (e-mail: edugan1@hotmail.com).

CARPHOPHIS VERMIS (Western Wormsnake). USA: ARKANSAS: Lafayette Co.: Stamps (Sec. 21, T16S, R23W). 05 October 1989. J. Davis. Verified by S. E. Trauth. Arkansas State University Herpetological Museum (ASUMZ 30824). New county record partially filling distributional gap between Columbia and Hempstead counties (Trauth et al. 2004. Amphibians and Reptiles of Arkansas. Univ. Arkansas Press, Fayetteville. 421 pp.). Most records of this snake are in central and northern Arkansas (Trauth et al. 2004, *op. cit*., fig. 493). Although extensive southern gaps remain, the species has now been reported from five counties in the extreme southern part of the state (see also Robison and McAllister 2007. Herpetol. Rev. 38:245–246).

Submitted by **HENRY W. ROBISON**, Department of Biology, Southern Arkansas University, Magnolia, Arkansas 71754, USA (e-mail: hwrobison@saumag.edu); and **CHRIS T. McALLISTER**, Department of Physical and Life Sciences, Chadron State College, Chadron, Nebraska 69337, USA (e-mail: cmcallister@csc.edu).

CEMOPHORA COCCINEA (Scarletsnake). USA: FLORIDA: Lafayette Co.: 0.48 km N of County Road 360 on State Road 51 (30.01245°N, 83.1937833°W), 2219 h. 4 June 2004. Michael E. Welker and Michael Arnold. Verified by Kenneth L. Krysko. Florida Museum of Natural History, Gainesville, Florida (UF 142711, 142712). New county record and range extension (Ashton and Ashton 1988. Handbook of Reptiles and Amphibians of Florida. Part 1. The Snakes. Second Edition. Windward Publishing, Miami, Florida. 176 pp.). These two specimens were found DOR at the same location. Both were adult males and both have 8-8 upper labials. Further, the first black body blotch is separated from the parietal scales by one scale in both specimens. The Northern Scarletsnake (*C. c. copei*) usually has 6-6 upper labials and the first black body blotch touches the parietal scales. The Florida Scarletsnake (*C. c. coccinea*) usually has 7–7 upper labials and the first black body blotch is separated from the parietal scales by one to four scales. Thus these two specimens appear to more closely resemble the morphological characters for *C. c. coccinea*, the Florida Scarletsnake. However, *C. c. coccinea* is known to occur about 90 miles to the south at the northern extent of its range.

Submitted by **MICHAEL E. WELKER**, 3105 Eads Place, El Paso, Texas 79935, USA (e-mail: herpcon@cox.net); and **MICHAEL ARNOLD**, 1911 NW 55th Terrace, Gainesville, Florida 32605, USA (e-mail: marnold35@cox.net).

COLUBER CONSTRICTOR (North American Racer). USA: MINNESOTA: Fillmore Co.: County Road 27, ca. 3.2 km W of River Road (east of Rushford). 5 October 2007. Stephen J. Schmitt and Philip A. Cochran. James Ford Bell Museum (JFBM 15217). Verified by Kenneth H. Kozak. DOR. First county record (Oldfield and Moriarty 1994. Amphibians & Reptiles Native to Minnesota. University of Minnesota Press, Minneapolis, Minnesota. 237 pp.).

Submitted by **STEPHEN J. SCHMITT** (e-mail: sjschm04@smumn.edu) and **PHILIP A. COCHRAN,** Biology Department, Saint Mary's University of Minnesota, 700 Terrace Heights, Winona, Minnesota 55987, USA (e-mail: pcochran@smumn.edu).

DIADOPHIS PUNCTATUS REGALIS (Regal Ring-necked Snake). USA: IDAHO: Jerome Co.: A single individual was observed ca. 10.4 km S of Jerome within the Snake River Canyon (42.6303°N, 114.5168°W; NAD83/WGS84). 31 May 2001. Javan Bauder and Jared Bauder. Color voucher photo deposited at the Idaho Museum of Natural History (IMNH 1922). Verified by Charles R. Peterson. Individual was lying in cheatgrass at the base of a large boulder ca. 1130 h with sunny skies and <10% cloud cover. Surrounding area consisted of scattered boulders intermixed with cheatgrass. First county record. Extends the published range of this species west ca. 170 km. An unpublished observation of *D. p. regalis* was made from Twin Falls County, Idaho, on the rim of the Snake River Canyon, ca. 6 km SE on 03 April 2006 (Bill Bosworth, Idaho Conservation Data Center, Idaho Department of Fish and Game, pers. comm.), suggesting that these observations may represent a previously undocumented population. Nearest published observations of *D. p. regalis* are from Bannock County, Idaho (Nussbaum et al. 1983. Amphibians and Reptiles of the Pacific Northwest. University of Idaho Press, Moscow, Idaho; Cossel 1997. Digital Atlas of Idaho, http://imnh.isu.edu/digitalatlas/bio/reptile /main/ repfram.htm). *Diadophis punctatus* is also documented from Boise County, Idaho, ca. 190 km NW (Nussbaum et al. 1983, *op. cit.*) and is reported as *D. p. occidentalis* (Wright and Wright 1957. Handbook of Snakes of the United States and Canada. Comstock Publishing Associates, Ithaca, New York).

Submitted by **JAVAN BAUDER**, Department of Biological Sciences, Idaho State University, Pocatello, Idaho 83209, USA; e-mail: baudjava@isu.edu.

ECHINANTHERA CEPHALOSTRIATA (NCN). BRAZIL: BAHIA: Municipality of Jussari, Boa Vista farm. 24 August 1992. M. O. Santos. Coleção Zoológica Gregório Bondar, of the Centro de Pesquisas do Cacau, Ilhéus, Bahia (CZGB 2045); Municipality of Arataca, Alto Rocha farm (15.1533333°S, 39.3025°W, 600–700 m elev.). 10 May–25 June 2001. Collector unknown. Museu de Zoologia da Universidade Estadual de Santa Cruz, Ilhéus, Bahia (MZUESC 1213); Municipality of Barra do Choça, Recanto da Adriana farm (14.9588889°S, 40.5491667°W, 800–900 m elev.) 02 May–29 November 2004. J. O. Ruas. (MZUESC 4337). Municipality of Pau Brasil, Mira Serra farm (15.3722222°S, 39.59°W, 600–700 m elev.) 05 October 2005–01 February 2006. Collector unknown. (MZUESC 4820). Precise data of the last three specimens are unknown, dates indicate periods between visits to the farms. All verified by V. Germano. The species was known from Espirito Santo to northeastern Santa Catarina, Brazil (Di-Bernardo 1996. The Snake 27:120–126). First state records extend known range 550 km N of Santa Teresa, the previous nearest locality known.

Submitted by **ANTÔNIO JORGE SUZART ARGÔLO,** Universidade Estadual de Santa Cruz – UESC, Km 16 Rodovia Ilhéus-Itabuna, CEP 45662-000, Ilhéus, Bahia, Brazil (e-mail: lachesis@uesc.br); and **JULIANA ALVES DE JESUS,** Rua Tertuliano Guedes de Pinho, n. 60, Apt 201, Bairro Jaçanã, CEP 45608-455, Brazil (e-mail: juli_dae@hotmail.com).

LAMPROPELTIS TRIANGULUM ELAPSOIDES (Scarlet Kingsnake). USA: FLORIDA: Hamilton Co.: 21.1 km N of U.S. 41 on County Road 135 (30.4850167°N, 82.7112667°W), 1923 h. 10 May 2004. Michael E. Welker and Michael Arnold. Verified by Kenneth L. Krysko. Florida Museum of Natural History, Gainesville, Florida (UF 142709). New county record and range extension (Ashton and Ashton 1988. Handbook of Reptiles and Amphibians of Florida. Part 1. The Snakes. Second Edition. Windward Publishing, Miami, Florida. 176 pp.). This large adult male was captured alive as it began to enter the roadway. It died in captivity on 29 June 2004.

Submitted by **MICHAEL E. WELKER**, 3105 Eads Place, El Paso, Texas 79935, USA (e-mail: herpcon@cox.net); and **MICHAEL ARNOLD**, 1911 NW 55th Terrace, Gainesville, Florida 32605, USA (e-mail: marnold35@cox.net).

LYCODON JARA (Yellow-speckled Wolf Snake). INDIA: HARYANA: Kalesar National Park (30.8°N, 77.34°E), 314 m elev. 13 March 2007. A. N. Rizvi. Northern Regional Station, Zoological Survey of India, Dehra Dun, Uttarakhand. ZSI NRS R.61. Verified by Indraneil Das. First record from Haryana State. Known from Dehra Dun (Uttarkhand), Ganjam (Orissa), east to the eastern Indian states of West Bengal and Arunachal Pradesh (Whitaker and Captain 2004. Snakes of India. The Field Guide. Draco Books, Chennai. xiv + 481 pp.), and extension of range to the west by ca. 75 km W of Dehra Dun.

Submitted by **ARCHANA BAHUGUNA** and **P. T. BHUTIA**, 218 Kaulagarh Road, Dehra Dun 248195, Uttarakhand, India; e-mail: bahugunaarch_nandini@yahoo.co.in.

MICRURUS FULVIUS (Harlequin Coralsnake). USA: FLORIDA: Lafayette Co.: 5.15 km N of Dixie County line on County Road 357 (29.86575°N, 83.2381167°W), 1700 h. 2 May 2004. 0.97 km N of Dixie County line on County Road 357 (29.8337333°N, 83.237116°W), 1915 h. 29 September 2004. Michael E. Welker and Michael Arnold. Verified by Kenneth L. Krysko. Florida Museum of Natural History, Gainesville, Florida (UF 141545, 142710). New county record and range extension (Ashton and Ashton 1988. Handbook of Reptiles and Amphibians of Florida. Part 1. The Snakes. Second Edition. Windward Publishing, Miami, Florida. 176 pp.). The first specimen (UF 141545) was an adult and the second specimen (UF 142710) was a small juvenile. Both were found DOR.

Submitted by **MICHAEL E. WELKER**, 3105 Eads Place, El Paso, Texas 79935, USA (e-mail: herpcon@cox.net); and **MICHAEL ARNOLD**, 1911 NW 55th Terrace, Gainesville, Florida 32605, USA (e-mail: marnold35@cox.net).

PSAMMODYNASTES PICTUS (Painted Mock Viper). INDONESIA: SULAWESI: Propinsi Sulawesi Tenggara, Kabupaten Kolaka Utara, Kecamatan Lasusua, Desa Tobaku (3.4166667°S, 120.8666667°E), 19–27 m elev. 13 August 2007. J. A. McGuire, R. A. Chong, B. J. Evans, C. W. Linkem, R. Elianur, N. P. Sumanto, and A. Rasit. JAM (Jimmy A. McGuire field series, uncataloged voucher at MZB or MVZ, 7289–91). 4 September 2007. J. A. McGuire, R. A. Chong, C. J. Hayden, N. P. Sumanto, Umilaela, and A. Rachmansyah. (JAM 8352–69, 8422). Verified by D. T. Iskandar. Easternmost record (Stuebing and Inger 1999. A Field Guide to the Snakes of Borneo. Natural History Publications [Borneo] Sdn. Bhd., Kota Kinabalu. vii + 254 pp.). The range currently includes Malaysia, Singapore, and Indonesia (Borneo, Riau Archipelago, and Sumatra), but has not been previously reported from Sulawesi (de Lang and Vogel 2006. The Snakes of Sulawesi. A Field Guide to the Land Snakes of Sulawesi with Identification Keys. Edition Chaimaira, Frankfurt am Main. 312 pp.). A total of 22 individuals were collected: 12 males (SVL 350–435 mm), six females (SVL 360–389 mm), and four juveniles (SVL 230–295 mm). All collected in vegetation alongside or overhanging a small, highly disturbed stream within cacao plantations. Due to the disturbed nature of the habitat and the abundance of specimens collected at this one site, even after thorough sampling of surrounding areas, we suspect it was introduction through human agencies on the island.

Submitted by **REBECCA A. CHONG** (e-mail: rchong@berkeley.edu), and **JIMMY A. McGUIRE** (e-mail: mcguirej@berkeley.edu), Museum of Vertebrate Zoology, University of California, Berkeley, California 94720, USA (e-mail: mcguirej@berkeley.edu).

REGINA RIGIDA RIGIDA (Glossy Crayfish Snake). USA: FLORIDA: Lafayette Co.: 7.6 km E of State Road 51 on County Road 360 (29.9918833°N, 83.1198333°W), 2348 h. 13 May 2005. Michael E. Welker and Michael Arnold. Verified by Kenneth L. Krysko. Florida Museum of Natural History, Gainesville, Florida (UF 144324). New county record and range extension (Ashton and Ashton 1988. Handbook of Reptiles and Amphibians of Florida. Part 1. The Snakes. Second Edition. Windward Publishing, Miami, Florida. 176 pp.). This specimen was found writhing in the road, apparently after just being hit by a motor vehicle. It died a short time later.

Submitted by **MICHAEL E. WELKER**, 3105 Eads Place, El Paso, Texas 79935, USA (e-mail: herpcon@cox.net); and **MICHAEL ARNOLD**, 1911 NW 55th Terrace, Gainesville, Florida 32605, USA (e-mail: marnold35@cox.net).

RAMPHOTYPHLOPS BRAMINUS (Brahminy Blindsnake). USA: FLORIDA: Lake Co.: Astatula, Lake Co. Landfill, 1.1 km SE of junction of State Road 19 and County Road 448 (28.7581667°N, 81.7453333°W; WGS84). 06 December 2007. Ridge Kaiser, Jennifer L. Matusick, and Julie Hanson. Florida Museum of Natural History photographic voucher (UF 152547). Verified by Kenneth L. Krysko. New county record. This exotic species has now been collected from at least 18 counties in Florida (Somma 2007. Herpetol. Rev. 38:355–356), and it has been documented from two adjacent counties, Seminole (Owen et al. 1998. Herpetol. Rev. 29:115) and Orange (Ernst and Brown 2000. Herpetol. Rev. 31:256). The specimen was found while excavating Gopher Tortoise (*Gopherus polyphemus*) burrows at a landfill.

Submitted by **LISA FAIRCHILD,** LPG Environmental and Permitting Services, Inc., 1174 Camp Avenue, Mount Dora, Florida 32757, USA (e-mail: lfairchild@lpgenvironmental.com); and **KEVIN M. ENGE**, Florida Fish and Wildlife Conservation Commission, 4005 South Main Street, Gainesville, Florida 32601, USA (e-mail: kevin.enge@myfwc.com).

SISTRURUS CATENATUS EDWARDSII (Desert Massasauga). USA: NEW MEXICO: Santa Fe Co.: 3.9 km S and 4.2 km E of Stanley (35.1128°N, 105.9337°W; NAD83/WGS84); 1925 m elev. 6 September 2007. J. N. Stuart and A. Q. Roberts. Verified by J. Tomasz Giermakowski. University of New Mexico Museum of Southwestern Biology (MSB 73380). New county record (Degenhardt et al. 1996. Amphibians and Reptiles of New Mexico. Univ. New Mexico Press, Albuquerque. xx + 431 pp.) and second record for the Estancia Basin of New Mexico (Stuart and Brown 1996. Herpetol. Rev. 27:214).

Submitted by **JAMES N. STUART** and **AARON Q. ROBERTS**, New Mexico Department of Game and Fish, Conservation Services Division, Santa Fe, New Mexico 87504-5112, USA (e-mail: james.stuart@state.nm.us).

STORIERIA OCCIPITOMACULATA (Red-bellied Snake). USA: ILLINOIS: McDonough Co.: Spring Lake (40.50309°N, 90.72056°W) On hiking trail. 28 August 2005. Collected by Benjamin R. Wodika. Verified by Michael A. Romano. Western Illinois University Herpetology collection 14161. Adult, 21.0 cm SVL. New county record. This species was believed to be present in the area, but there are few voucher specimens from surrounding counties. (INHS. Illinois Natural History Survey. http://

www.inhs.uiuc.edu/cbd/herpdist/species/st_occipit.html http. Accessed 10 Oct 2007; Phillips et al. 1999. Field Guide to Amphibians and Reptiles of Illinois. Illinois Nat. Hist. Surv. Manual 8. Champaign, Illinois. 282 pp.)

Submitted by **BENJAMIN R. WODIKA**, Department of Biological Sciences, Western Illinois University, 1 University Circle Macomb, Illinois, USA.

VIRGINIA VALERIAE VALERIAE (Eastern Smooth Earthsnake). USA: FLORIDA: WALTON CO.: 3.7 km S of State Road 20 on County Road 83A East (30.46945°N, 86.1536333°W), 0035 h. 08 April 2005. Michael E. Welker. Verified by Kenneth L. Krysko. Florida Museum of Natural History, Gainesville, Florida (UF 144325). New county record and range extension (Ashton and Ashton 1988. Handbook of Reptiles and Amphibians of Florida. Part 1. The Snakes. Second Edition. Windward Publishing, Miami, Florida. 176 pp.). This specimen, along with three Okaloosa County specimens (UF 99675, 115415, 117732), help fill a large gap in this species' range along the gulf coast in the panhandle of Florida. This specimen was found DOR after a heavy rain.

Submitted by **MICHAEL E. WELKER**, 3105 Eads Place, El Paso, Texas 79935, USA**;** e-mail: herpcon@cox.net.

Herpetological Review, 2008, 39(1), 112–121.

Additions to the Herpetofauna of Endau-Rompin, Johor, West Malaysia

PERRY LEE WOOD, JR.*
L. LEE GRISMER**
TIMOTHY M. YOUMANS
Department of Biology, La Sierra University
Riverside, California 92515, USA

NUROLHUDA NASIR
Johor National Park Corporation (Perbadanan Taman Negara Johor)
West Malaysia

NORHAYATI AHMAD
Faculty of Science and Technology, 43600 Bangi
Universiti Kebangsaan, Malaysia, Malaysia

and
JULIANA SENAWI
Institute Biodiversity, Department of Wildlife and National Park,
Bukit Rengit, 28500 Lanchang, Pahang, Malaysia

** Corresponding author; current address:*
Department of Biology, Villanova University
Villanova, Pennsylvania 19085, USA
e-mail: pwoo213@my.lasierra.edu

*** Institute for Environment and Development (LESTARI)*
Universiti Kebangsaan Malaysia 43600 Bangi, Selangor Darul Ehsan Malaysia

The Endau-Rompin National Park is located at the southern end of Peninsular Malaysia. It encompasses 87,000 ha across the southernmost portion of the Banjaran Titi Wangsa mountain range in the states of Pahang and Johor (Fig. 1). The park is comprised of lowland mixed dipterocarp forests, flood plains, riparian environments, and hill top dipterocarp forests. The three highest peaks in the Johor section are Gunung Tiong (1014 m) near the center of the park, Gunung Bekok (953 m) in the west, and Gunung Besar (1036 m) in the northwest, all of which are flanked by many other smaller hills.

To date there have been three published herpetological surveys of the Johor section of Endau-Rompin (Daicus and Hashim 2004; Kiew 1987; Lim 1989), two amphibian surveys on the northern side in Pahang (Norhayati et al. 2004a,b), and an unpublished senior thesis on the amphibians from the Johor portion (Bhaarathyraja 2006). Collectively, these surveys listed one species of caecilian, 48 frogs, 4 turtles, 26 lizards, and 17 snakes. In addition to the herpetological surveys, there have been several other scientific surveys (zoological, botanical, and geological) conducted between 1985 and 2004 (Malayan Nature Society 1988; Shaharuddin et al. 2004).

Presented here are the results of a survey conducted in the Peta region (04°28.318'N, 101°22.635'E) on 24–31 August 2005 and in Selai (02°26.000'N, 103°15.283'E) from 30 August to 5 September 2006 (Fig. 1). Peta comprises 1600 ha of pristine lowland rainforest and Selai, which lies 26 km to the south, comprises 660 ha of similar habitat. These lowland regions are characterized by mixed dipterocarp forests and are crisscrossed by several small streams and one large river, Sungai Endau. New species records presented below are based on voucher specimens deposited in the Zoological Reference Collection (ZRC) at The Raffles Museum of Biodiversity Research, National University of Singapore, the La Sierra University Herpetological Collection (LSUHC), and a collection deposited in the Nature Education Research Center (NERC) in Peta. Voucher photographs of some species are deposited in the La Sierra University Digital Photograph Collection (LSUDPC). An up-to-date checklist of the herpetofauna of Endau-Rompin is presented in Table 1. The taxonomies of Frost et al. (2006) for the *Bufo quadriporcatus* group, Matsui et al. (2005) for *Microhyla,* Gower et al. (2002) for the Ichthyophiidae, Shaffer et al. (1997) for the higher classification of turtles, Carranza and Arnold (2006) for *Hemidactylus,* Mausfeld and Schmitz (2003) for *Eutropis,* Vogel and Van Rooijen (2007) for *Dendrelaphis,* Utiger et al. (2002) for Old World rat snakes, David and Das (2004) for *Ptyas,* David et al. (2006) for the *Trimeresurus puniceus* complex, and Malhotra and Thorpe (2004) for the Viperidae are followed. The following species are newly recorded for the park.

Anura

Ansonia endauensis Grismer, 2006. (Fig. 2). Peta; Sungai Kawal, 31 August 2005, ZRC 1.11555–58.

Ingerophrynus gollum Grismer, 2007. Peta; Sungai Kawal. 28 August 2005. ZRC 1.11569–71.

Microhyla butleri Boulenger, 1900. Peta; Visitor center, 25 August 2005, LSUHC 7619.

Micryletta inornata (Boulenger, 1890). (Fig. 3). Peta, Visitor center, 26 August 2005, LSUHC 7626.

Limnonectes plicatellus (Stoliczka, 1873). (Fig. 4). Selai; Sungai Ketiau Padi. 1 September 2006, LSUHC 8175.

Rana laterimaculata Barbour and Noble, 1916. Peta; Visitor center. 25 August, LSUHC 7634.

Rana miopus Boulenger, 1918. Johor, Endau-Rompin: NERC 50720. No data.

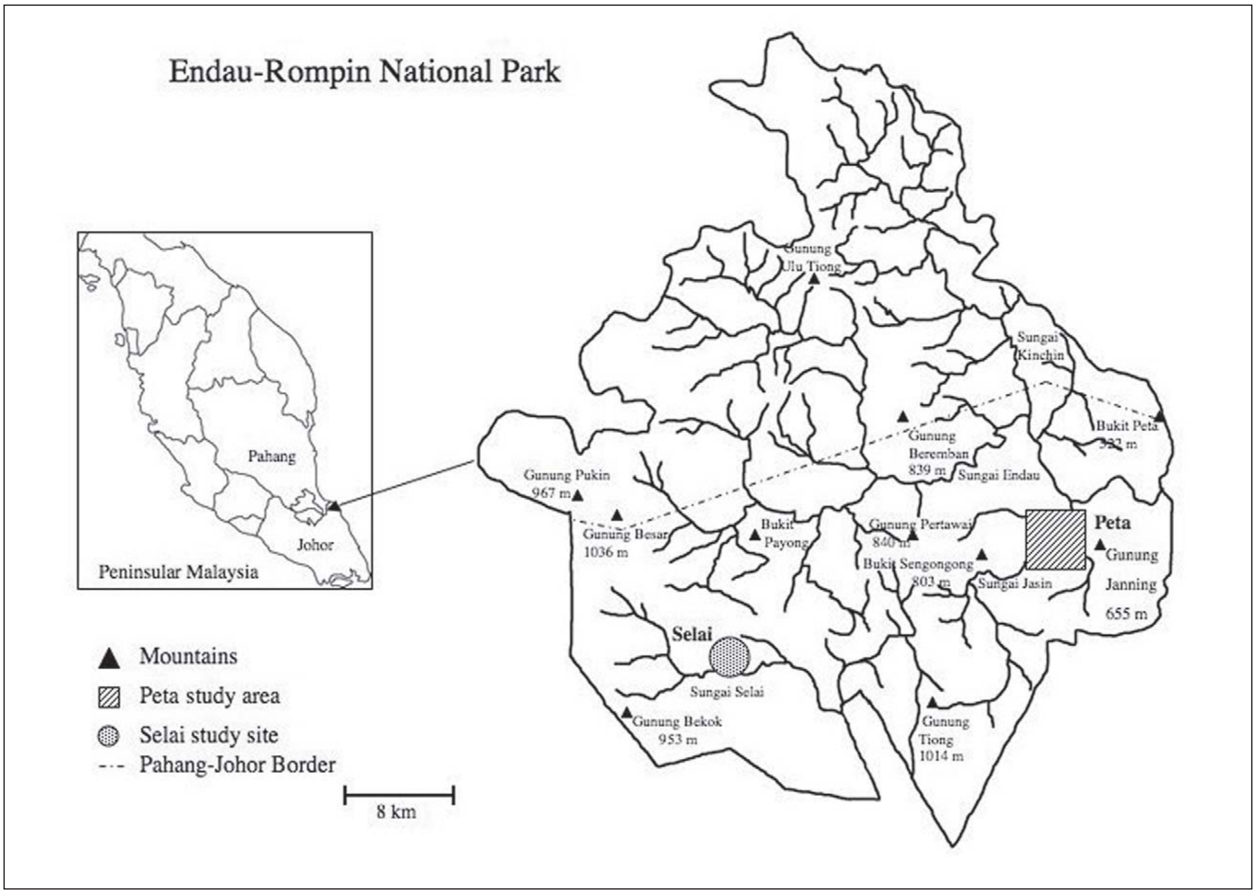


FIG. 1. Location of the Peta and Selai study sites, Johor, West Malaysia.

Rana nicobarensis (Stoliczka, 1870). (Fig. 5). Peta; Visitor center, 25 August, LSUHC 7624.

Testudines

Dogania subplana (Geoffroy Saint-Hillaire, 1809). Selai; Lubuk Merekek, Sungai Selai, 4 September 2006, LSUDPC 2619–29.

Manouria emys Schlegel & Müller, 1844. Selai, LSUDPC 2018. Photograph provided by park staff.

Squamata (Lizards)

Draco maximus Boulenger, 1893. (Fig. 6). Selai; Lubuk Tapah, 1 September 2006, LSUHC 8206.

Gonocephalus abbotti Cochran, 1922. (Fig. 7). Peta; NERC, 26 August 2005, LSUHC 7647. Sungai Semawak, 27 August 2005, LSUHC 7682. Selai; Lubuk Tapah, 31 August 2006, LSUHC 8172. These records represent a southern range extension of approximately 450 km.

Aeluroscalabotes felinus (Günther, 1864). (Fig. 8). Selai; Lubuk Tapah, 2 September 2006, LSUHC 8194.

Cnemaspis kendallii (Gray 1845). Peta; Sungai Semawak, 27 August 2005, LSUHC 7691. Selai; Lubuk Tapah, 30, 31 August and 2 and 4 September 2006, LSUHC 8122, 8210, 8126, 8191.

Gehyra mutilata (Wiegmann, 1834). Peta; Sungai Kawal. 28 August 2005, LSUDPC 1357. Selai; Lubuk Tapah, 2 September 2006, LSUHC 8187.

Hemidactylus craspedotus (Mocquard, 1890). Selai; Lubuk Merekek, 5 September 2006, LSUHC 8230.

Hemidactylus frenatus (Duméril & Bibron, 1836). Peta; Visitor Center. 25 August 2005, LSUHC 7645–46.

Hemidactylus platyurus (Schneider, 1792: 30). Peta; Visitor center, 25 August, LSUHC 7643–7644.

Hemiphyllodactylus typus (Bleeker, 1860). Peta. LSUDPC 1002–04. Photograph provided by park staff.

Ptychozoon kuhli Stejneger, 1902. Peta; Visitor Center, 27 and 30 August 2005, LSUHC 7640, 7716, respectively.

Dasia grisea (Gray, 1845). Selai; LSUPDC 1947–48. Photograph provided by park staff.

FIGS. 2–5, Opposite page, left column, from top to bottom. All photos by PLW. FIG. 2. *Ansonia endauensis* (ZRC 1.11555) from Peta, Sungai Kawal. FIG. 3. *Micryletta inornata* (LSUHC 7626) from the NERC. FIG. 4. *Limnonectes plicatellus* (LSUHC 8175) from Selai, Sungai Ketiau Padi. FIG. 5. *Rana nicobarensis* (LSUHC 7624) from Peta, Visitor Center.

FIGS. 6–8, Opposite page, right column, from top to bottom. All photos by PLW. FIG. 6. *Draco maximus* (LSUHC 8206) from Selai, Lubuk Tapah. FIG. 7. *Gonocephalus abbotti* (LSUHC 7647) near the NERC. FIG. 8. *Aeluroscalabotes felinus* (LSUHC 8194) from Selai, Lubuk Tapah.

Lipinia vittigera (Boulenger, 1894). Selai; Lubuk Merekek, 5 September 2006, LSUHC 8211.

Sphenomorphus scotophilus (Boulenger, 1900). Peta; Sungai Kawal, 28 August 2005, LSUHC 7688. Selai; Lubuk Merekek, 3 September 2006, LSUHC 8199.

Squamata (Snakes)

Ahaetulla facsiolata (Fischer, 1885). Peta: LSUHC 7697, found in collection at NERC with no data.

Ahaetulla mycterizans (Linneaus, 1758). Peta; Sungai Kawal, 26 August 2005, LSUHC 7680. Selai; Lubuk Merekek, 4 September 2006, LSUHC 8224.

Boiga drapiezii (Boie, 1872). (Fig. 9). Selai; Trail to Lubuk Tapah, 31 August and 2 September 2006, LSUHC 8143, 8192, respectively. Lubuk Tapah, 31 August 2006, LSUHC 8157.

Boiga nigriceps (Günther, 1863). (Fig. 10). Selai; Lubuk Merkek, 30 August 2006, LSUHC 8134, Trail to Lubuk Tapah, 31 August 2006, LSUHC 8142, Lubuk Tapah, 3 September 2006, LSUHC 8201.

Boiga jaspidea (Duméril, Duméril & Bibron, 1854). (Fig. 11). Peta; On road to Sungai Kawal, 25 August 2005, LSUHC 7679.

Calamaria lumbricoidea Boie, 1827. Peta; photographed provided by park staff. LSUDPC 1940.

Chrysopelea pelias (Boie, 1827). Peta; found in collection at NERC with no data, LSUHC 7696.

Coelognathus flavolineata (Schlegel, 1873). Selai; LSUDPC 2829. Photograph provided by park staff.

Dendrelaphis caudolineatus (Gray, 1830–1835). Selai; Lubuk Merekek, 5 September 2006, LSUHC 8225–26.

Dryocalamus subannulatus (Duméril, Bibron & (Duméril, 1854). Peta; Sungai Semawak, 27 August 2005, LSUHC 7661. Selai; Lubuk Merekek, 30 August 2006, LSUHC 8136.

Lycodon effraenis (Cantor, 1847). (Fig. 12). Peta; Sungai Semawak, 31 August 2005, LSUHC 7734.

Macrocalamus sp. Peta; LSUDPC 2830–31. No data. Photograph provided by park staff.

Oligodon purpurascens (Schlegel, 1837). (Fig. 13). Selai; on trail to Kuala Marong, August 30, 2005, LSUHC 7715.

Orthriophis taeniurus (Cope, 1861). Peta. LSUDPC 1005. No data. Photograph provided by park staff.

Pareas margaritophorus Jan, 1866. Peta; Bekok, photographed provided by park staff, LSUDPC 1078–79.

Pseudorabdion longiceps (Cantor, 1847). Peta; Visitor Center, 25 August 2005, LSUHC 7602.

Ptyas carinata (Günther, 1858). Peta; Sungai Semawak, 31 August 2005, LSUHC 7731. Selai; Lubuk Tapah, 31 August 200, LSUHC 8173.

Ptyas fusca (Günther, 1858). Peta; Sungai Semawak, 26 August 2005, LSUHC 7723.

Xenodermus javanicus Reinhardt 1836. Peta; LSUDPC 2032. No data. Photograph provided by park staff.

Calliophis bivirgatus (Boie, 1827). (Fig. 14). Peta; NERC 50716, no data. Photograph provided by park staff.

Parias hageni (Lidth De Jeude, 1886). (Fig. 15). Peta; NERC July 2006, LSUHC 8193.

Parias sumatranus (Raffles, 1882). Selai; LSUDPC 1971. Photograph provided by park staff.

Popeia fucata (Vogel, David & Pauwels, 2004). Peta. NERC 50704, 50748, no data. Photograph provided by park staff.

Trimeresurus wiroti (Trutnau, 1981). (Fig. 16). Peta; LSUDPC 952, no data. Photograph provided by park staff.

Discussion.—Daicus and Hashim (2004) reported both *Pelophryne signata* and *Pelophryne brevipes* as being present although only *P. brevipes* occurs in Peninsular Malaysia (Inger 1966). Also reported by Daicus and Hashim (2004) was *Ansonia leptopus*. Unfortunately, the specimen was released and no voucher photograph was taken. Given that *A. leptopus* is restricted to Borneo (Wood et al. 2008) it is likely the specimen was confused with *A. endauensis*. Daicus and Hashim (2004) reported *Rana signata* which we recognize here as *R. picturata* (*fide* Brown and Guttman 2002). *Typhlops diardii* was also reported (Daicus and Hashim 2004) although according to Wallach (2001) *T. diardii* is only found in NE Pakistan, N India, Nepal, Bangladesh, Myammar, Thailand, Laos, Vietnam, and China. This species is easily confused with *Typhlops muelleri* whose presence in the park had previously been reported (Daicus and Hashim 2004).

Kiew (1987) reported *Ansonia malayana* from Endau-Rompin, however there is no voucher specimen. We believe it is unlikely that this montane species, known no further south than Bukit Larut (Grismer 2006; Inger 1960), would not occur in a southern, lowland forest. It is our hypothesis that Kiew (1987) actually collected *A. endauensis,* a species that is superficially very similar to *A. malayana* (Grismer 2006a, b). Kiew (1987) also reported a specimen he referred to as *Microhyla* sp. Unfortunately the whereabouts of the specimen is unknown. It is likely, however, that it belonged to one of the six confirmed species of *Microhyla* known from Endau-Rompin (Table 1) that are often confused with one another.

Lim (1989) reported *Ichthyophis glutinosus* from Endau-Rompin. In light of the current taxonomic problems within the Ichthyophiidae in general and *Ichthyophis* (Gower et al. 2002; Grismer et al. 2006) in particular, it is difficult to accord any individual specimen to a particular species with any degree of confidence. Therefore, we follow Kiew (1987) in recognizing only

---

FIGS. 9–12, Opposite page, left column, from top to bottom. All photos by PLW. FIG. 9. *Boiga drapiezii* from Selai, Lubuk Tapah. FIG. 10. *Boiga nigriceps* from Selai, Lubuk Tapah. FIG. 11. *Boiga jaspidea* (LSUHC 7656) from Peta, at the NERC. FIG. 12. *Lycodon effraenis* (LSUHC 7734) from Peta, Sungai Semawak.

FIGS. 13–16, Opposite page, right column, from top to bottom. FIG. 13. *Oligodon purpurascens* (LSUHC 7715) from Peta, on the trail to Kulau Marong. Photo by PLW. FIG. 14. *Calliophis bivirgatus* from Peta. Photo by LLG. FIG. 15. *Parias hageni* from Peta. Photo by PLW. FIG. 16. *Trimeresurus wiroti* from Peta. Photo by LLG.

Table 1. List of amphibians and reptiles of Endau-Rompin National Park, Johor, West Malaysia, and the most recent reporting author. Species lacking an author designation were reported by earlier workers.

	Kiew 1987	Lim 1989	Norhayati et al. 2004	Daicus & Hashim 2004	Bhaarathyraja 2006	Present survey 2005–06
Species						
Amphibians (Frogs)						
Bufonidae						
Ansonia endauensis Grismer, 2006					X	X
Bufo asper Gravenhorst, 1829	X	X	X	X	X	X
Bufo melanostictus Schneider, 1799				X		
Ingerophrynus parvus (Boulenger, 1892)	X		X	X		X
Ingerophrynus gollum Grismer, 2007						X
Leptophryne borbonica Peters, 1867			X			
Pedostibes hosii (Boulenger, 1892)	X				X	X
Pelophryne brevipes (Peters, 1867)		X		X		
Megophryidae						
Leptobrachium hendricksoni Taylor, 1962	X	X	X			X
Megophrys nasuta (Schelegel, 1858)		X	X	X	X	X
Microhylidae						
Chaperina fusca Mocquard, 1892				X		
Kalophrynus palmatissimus Kiew, 1984				X	X	X
Kalophrynus pleurostigma (Gravenhorst, 1838)	X	X		X		X
Kaloula baleata (Müller, 1836)				X		X
Kaloula pulchra Gray, 1831			X	X		
Metaphrynella pollicaris (Boulenger, 1890)				X		
Microhyla annectens Boulenger, 1900				X		X
Microhyla berdmorei (Blyth, 1856)			X	X	X	
Microhyla butleri Boulenger, 1900						X
Microhyla heymonsi Vogt, 1911			X	X	X	X
Microhyla fissipes Boulenger, 1884				X		X
Microhyla mantheyi Das, Yakkob and Sukumaran, 2007					X	X
Micryletta inornata (Boulenger, 1890)						X
Ranidae						
Amolops larutensis (Boulenger, 1899)			X	X	X	X
Fejervarya cancrivora (Gravenhorst, 1829)				X		
Fejervarya limnocharis (Gravenhorst, 1829)			X	X	X	
Limnonectes blythii (Boulenger, 1920)	X	X	X	X	X	X
Limnonectes kuhlii (Tschudi, 1838)	X	X	X			
Limnonectes laticeps (Boulenger, 1882)	X	X		X		X
Limnonectes malesianus (Kiew, 1984)	X			X		X
Limnonectes plicatellus (Stoliczka, 1873)						X
Occidozyga laevis (Günther, 1858)	X		X	X		
Occidozyga martensii (Peters, 1827)	X					X
Rana erythraea (Schlegel, 1837–1844)		X				X
Rana glandulosa Boulenger, 1882	X		X	X		X
Rana hosii Boulenger, 1891	X	X	X	X	X	
Rana laterimaculata Barbour and Noble, 1916						X
Rana miopus Boulenger, 1918						X
Rana nicobarensis (Stoliczka, 1870)			X			X
Rana picturata Boulenger, 1920	X	X	X	X	X	X
Rana raniceps (Peters, 1871)	X	X	X	X	X	X
Taylorana hascheana (Stoliczka, 1870)	X	X				
Rhacophoridae						
Nyctixalus pictus (Peters, 1871)	X					X
Polypedates colletti Boulenger, 1890		X				
Polypedates leucomystax (Gravenhorst, 1829)	X	X	X	X	X	X
Polypedates macrotis (Boulenger, 1894)	X			X	X	X
Rhacophorus appendiculatus (Günther, 1859 "1858")					X	X
Rhacophorus cyanopunctatus Mathey and Steiof, 1998	X	X				
Rhacophorus nigropalmatus Boulenger, 1895				X	X	X
Rhacophorus pardalis Günther, 1858				X		
Rhacophorus tunkui Kiew, 1987	X					
Amphibians (Caecilians)						
Ichthyophiidae						
Ichthyophis sp.		X				

Table 1. Continued.

	Kiew 1987	Lim 1989	Norhayati et al. 2004	Daicus & Hashim 2004	Bhaarathyraja 2006	Present survey 2005–06
Species						
Reptiles (Turtles)						
Bataguridae						
Heosemys grandis (Gray, 1860)				X		
Heosemys spinosa (Gray, 1831)	X					
Notochyles platynota (Gray, 1834)	X					X
Testudinidae						
Manouria emys Schlegel and Müller, 1844						X
Trionychidae						
Amyda cartilaginea (Boddart, 1770)	X					
Dogania subplana Geffroy Saint-Hillaire, 1809						X
Reptiles (Lizards)						
Agamidae						
Aphaniotis fusca Peters, 1864	X			X		X
Bronchocela cristatella (Kuhl, 1820)				X		X
Calotes versicolor (Daudin, 1802)				X		
Draco blandfordii Boulenger, 1885				X		
Draco fimbriatus Kuhl, 1820				X		X
Draco formosus Boulenger, 1900				X		X
Draco maximus Boulenger, 1893						X
Draco melanopogon Boulenger, 1887				X		X
Draco quinquefasciatus Hardwicke and Gray, 1827	X					X
Draco sumatranus Schlegel, 1844	X			X		X
Gonocephalus abbotti Cochran, 1922						X
Gonocephalus grandis (Gray, 1845)				X		X
Gonocephalus liogaster Günther, 1872				X		X
Eublepharidae						
Aeluroscalabotes felinus (Günther, 1864)						X
Gekkonidae						
Cnemaspis kendallii (Gray, 1845)						X
Cyrtodactylus consobrinus (Peters, 1871)				X		X
Cyrtodactylus pulchellus Gray, 1828				X		
Cyrtodactylus quadrivirgatus Taylor, 1962				X		X
Cyrtodactylus sworderi (Smith, 1925)				X		X
Gehyra mutilata (Wiegmann, 1834)						X
Gekko gecko (Linnaeus, 1758)				X		
Gekko monarchus (Duméril and Bibron, 1836)				X		X
Gekko smithii Gray, 1842				X		X
Hemidactylus craspedotus (Mocquard, 1890)						X
Hemidactylus frenatus (Duméril and Bibron, 1836)						X
Hemidactylus platyurus (Schneider, 1792)						X
Hemiphyllodactylus typus (Bleeker, 1860)						X
Ptychozoon kuhli Stejneger, 1902						X
Scincidae						
Dasia grisea (Gray, 1845)						X
Dasia olivacea Gray, 1839	X					
Eutropis longicaudatus (Hallowell, 1857)				X		
Eutropis macularius (Blyth, 1853)				X		
Eutropis rugiferus (Stoliczka, 1870)				X		
Eutropis multifasciatus (Kuhl, 1820)	X			X		X
Lipinia vittigera (Boulenger, 1894)						X
Sphenomorphus scotophilus (Boulenger, 1900)						X
Varanidae						
Varanus nebulosus (Gray,1831)				X		
Varanus rudicollis (Gray, 1845)	X			X		
Varanus salvator (Laurenti, 1786)	X			X		X
Reptiles (Snakes)						
Typhlopidae						
Typhlops muelleri (Shlegel, 1839)				X		X
Pythonidae						
Python reticulatus (Schneider, 1839)	X					

TABLE 1. Continued.

	Kiew 1987	Lim 1989	Norhayati et al. 2004	Daicus & Hashim 2004	Bhaarathyraja 2006	Present survey 2005–06
Species						
Colubridae						
Ahaetulla facsiolata (Fischer, 1885)						X
Ahaetulla mycterizans (Linneaus, 1758)						X
Ahaetulla prasina (Boie, 1827)	X			X		
Asthenodipsas laevis Boulenger, 1900	X					
Boiga dendrophila (Boie, 1827)	X					
Boiga drapiezii (Boie, 1872)						X
Boiga jaspidea (Duméril,Duméril & Bibron, 1854)						X
Boiga nigriceps (Günther, 1863)						X
Calamaria lumbricoidea Boie, 1827						X
Chrysopelea paradisi Boie, 1827	X					
Chrysopelea pelias (Linnaeus, 1758)						X
Coelognathus flavolineatus (Schlegel, 1873)						
Dendrelaphis caudolineatus (Gray, 1830-1835)						X
Dendrelaphis formosus (Boie, 1827)				X		
Dendrelaphis pictus (Gmelin, 1789)	X					X
Dendrelaphis striatus (Cohn, 1906)						X
Dryocalamus subannulatus (Duméril, Bibron & Duméril, 1854)						X
Gongylosoma baliodeirus Boie, 1827	X					
Lycodon effraenis Cantor, 1847						X
Macrocalamus sp.						X
Macropisthodon rhodomelas (Boie, 1827)	X			X		
Oligodon purpurascens (Schlegel, 1837)						X
Oreophis porphyraceus (Cantor, 1839)	X					
Orthriophis taeniurus (Cope, 1861)						X
Pareas margaritophorus Jan, 1866						X
Pseudorabdion longiceps (Cantor, 1847)						X
Ptyas carinata (Günther, 1858)						
Ptyas fusca (Günther, 1858)						
Rhabdophis chrysargos (Schlegel, 1837)				X		X
Xenodermus javanicus Reinhardt, 1836						X
Elapidae						
Bungarus flaviceps Reinhardt, 1843	X			X		
Calliophis bivirgatus (Boie, 1827)						X
Calliophis intestinalis (Laurenti, 1768)	X					X
Ophiophagus hannah (Cantor, 1836)	X					
Viperidae						
Parias hageni (Lidth De Jeude, 1886)						X
Parias sumatranus (Raffles, 1882)						X
Popeia fucata (Vogel, David and Pauwels, 2004)						X
Trimeresurus wiroti Trutnau, 1981						X
Tropidolaemus wagleri Wagler, 1830	X			X		X

Ichthyophis sp.

Photographs were provided to us by park staff of what we believe is an undescribed species of *Macrocalamus* (LSUDPC 2830–31) based on the head not being distinct from the neck. This genus is currently represented by more northerly montane species and thus, this specimen is not only a southern range extension of approximately 180 km for *Macrocalamus* but the first specimen to be found in a lowland forest which represents a significant ecological departure as well (David and Pauwels 2004).

The new records reported here bring the total number of confirmed species of amphibians and reptiles in the park to 140 (1 caecilian, 51 frogs, 6 turtles, 39 lizards, and 43 snakes), representing an increase in diversity of 33% since Daicus and Hashim (2004) (Table 1). More importantly, these new records clearly underscore the understudied nature of the herpetofauna despite the many surveys that have been conducted (Bhaarathyraja 2006; Diacus and Hashim 2004; Kiew 1987; Lim 1989; Norhayati et al. 2004a,b). Additionally, the discovery of two new species of frogs, *Ansonia endauensis*, Grismer, 2006 and *Ingerophrynus gollum*, Grismer, 2007, the rediscovery of the gecko *Cyrtodactylus sworderi* (Smith, 1925; Grismer et al. 2007), and the discovery of *C. semenanjungensis* Grismer and Leong, 2005 just outside the eastern boundary of the park exemplify the need for continued systematic field work. The same argument could be made for the lowland rainforests of the entire state of Johor. Large portions of this unique section of the southern Malay Peninsula are being converted into oil palm and rubber tree plantations with extensive tracts reaching deep into Endau-Rompin (authors, pers. obs., 2006).

Acknowledgments.—We thank Chan Kin Onn, Daicus M. Belabut, Sumen Govendasamy, Rick Gregory, Thomas Szutz, Bradely Jones, and the staff of the Johor National Park at Peta and Selai for assistance in the field. We would also like to thank Mr. Chew Keng Lin for many valuable photographs of several species. The Economic Planning Unit, Prime Minister's Department issued a research pass 40/2001/1955.1105 to LLG. This research was supported in part by a College of Arts and Sciences Research Grant to

Literature Cited

Bhaarathyraja, T. 2006. Tinjuan Kepelbagaian Spesies Amfibia di Taman Negara Endau-Rompin, Johor. Unpublished senior paper. Universiti Kebangsan Malaysia. 53 pp.

Brown, R. M., and S. I. Guttman. 2002. Phylogenetic systematics of the *Rana signata* complex of the Philippine and Bornean stream frogs: reconsideration of Huxley's modification of Wallace's Line at the Oriental-Australian faunal zone interface. Biol. J. Linn. Soc. 76:393–461.

Carranza, S., and E. N. Arnold. 2006. Systematics, biogeography, and evolution of *Hemidactylus* geckos (Reptilia: Gekkonidae) elucidated using mitochondrial DNA sequences. Mol. Syst. Evol. 38:531–545.

David, P., and I. Das. 2004. On the grammar of the gender of *Ptyas* Fitzinger, 1843 (Serpents: Colubridae). Hamadryad 28:113–116.

———, and O. S. G. Pauwels. 2004. A re-evaluation of the taxonomy of *Macrocalamus lateralis* Günther, 1864 (Serpentes, Colubridae), with the descriptions of two new species. Raffles Bull. Zool. 52:635–645.

———, G. Vogel, S. P. Vijayakumar and N. Vidal. 2006. A revision of the *Trimeresurus punices*-complex (Serpentes: Viperidae: Crotalinae) based on morphological and molecular data. Zootaxa 1293:1–78.

Daicus, B., and R. Hashim. 2004. Herpetofauna of the western region of Endau-Rompin,Johore, Peninsular Malaysia. Malay J. Sci. 23:65–72.

Frost, D. R., T. Grant, J. Favovich, R. H. Bain, A. Haas, C. F. Haddad, R. O. De Sa, A. Channing, M. Wilkinson, S. C. Donnellan, C. J. Raxworthy, J. A. Campbell, B. L. Blotto, P. Moler, R. C. Drewes, R. A. Nussbaum, J. D. Lynch, D. M. Green, and W. C. Wheeler. 2006. The Amphibian Tree of Life. Bull. Amer. Mus. Nat. His. 297:1–370.

Gower, D. J., A. Kupfer, O. V. oommen, W. Himstedt, R. A. Nussbaum, S. P. Loader, B. Presswell, H. Müller, S. B. Krishna, R. Boistel, and M. Wilkinson. 2002. A molecular phylogeny of ichthyophiid caecilians (Amphibia: Gymnophiona: Ichthyophiidae): Out of India or Southeast Asia? Proc. Royal Soc. 269:1563–1569.

Grismer, L. L. 2006a. A new species of *Ansonia* Stoliczka 1872 (Anura: Bufonidae) from a lowland rainforest in Southern Peninsular Malaysia. Herpetologica 62:466–475.

———. 2006b. A new species of *Ansonia* Stoliczka 1872 (Anura: Bufonidae) from central Peninsular Malaysia and a revised taxonomy for Ansonia from the Malay Peninsula. Zootaxa 1327:1–21.

———. 2007. A new species of *Ingerophrynus* (Anura: Bufonidae) from a lowland rain forest in southern peninsular Malaysia. J. Herpetol. 41(2):224–229.

———, and T. M. Leong. 2005. New species of *Cyrtodactylus* (Squamata: Gekkonidae) from southern peninsular Malaysia. J. Herpetol. 39:584–591.

———, P. L. Wood, Jr., and T. M. Youmans. 2007. Redescription of *Crytodactylus sworderi* (Smith 1925) from southern Peninsular Malaysia. Hamadryad 31:250–257.

———, T. M. Youmans, P. L. Wood Jr., A. Ponce, S. B. Wright, B. S. Jones, R. Johnson, K. L. Sanders, D. J. Gower, and N. S. Yaakob. 2006. Checklist of the herpetofauna of Pulau Langkawi, Malaysia, with comments on taxonomy. Hamadryad 30:61–74.

Inger, R. F. 1960. A review of the Oriental toads of the genus *Ansonia* Stoliczka. Fieldiana Zool. 39:473–503.

———. 1966. Systematics and zoogeography of the Amphibia of Borneo. Fieldiana Zool. 52:1–402.

Kiew, B. H. 1987. An annotated checklist of the herpetofauna of Ulu Endau, Johor, Malaysia. Malayan. Nat. J. 41:413–424.

Lim, K. P. K. 1989. The amphibian fauna of Ulu Kinchin, Pahang, Malaysia. Malayan. Nat. J. 43:322–325.

Malayan Nature Society. 1988. Endau-Rompin—A Malaysian Heritage. Star Publications (Malaysia) Berhad, Kuala Lumpur. 221 pp.

Malhotra, A., and R. S. Thorpe. 2004. A phylogeny of four mitochondrial gene regions suggests a revised taxonomy for Asian pitvipers (*Trimeresurus* and *Ovophis*). Mol. Phyl. Evol. 32:83–100.

Matsui, M., H. Ito, T. Shimada, H. Ota, S. K. Saidapur, W. Khonsue, T. Tanaka-Ueno, and G. Wu. 2005. Taxonomic relationships within the pan-Oriental narrow-mouth toad *Microhyla ornata* as revealed by mtDNA analysis (Amphibia, Anura, Microhylidae). Zool. Sci. 22:489–495.

Mausfeld, P., and A. Schmitz. 2003. Molecular phylogeography, intraspecific variation and speciation of the Asian scincid lizard genus *Eutropis* Fitzinger, 1843 (Squamata: Reptilia: Scincidae): taxonomic and biogeographic implications. Organism. Div. Evol. 3:161–171.

Norhayati, A., J. Senawi, and B. L. Lim. 2004. A Photograpic Guide to Amphibians of Endau-Rompin State Park, Pahang. The Forestry Department of Peninsular Malaysia. 59 pp.

———, and S. K. S. Shamada. 2004. Fauna Amfibia di Taman Negri Endau Rompin. Taman Negeri Endau Rompin: Pengurusan, Persekitaran Fizikal dan Biologi. Jabatan Perhutanan Semenanjung Malaysia. pp. 197–206.

Shaffer, H. B., P. Meylan, and M. L. McKnight. 1997. Tests of turtle phylogeny: molecular, morphological, and paleontological approaches. Syst. Biol. 46:235–268.

Shaharuddin, M. I., M. I. Mokhtar, W. Yusoff, W. A. M. Rahim, M. and A. Latiff (Eds.). 2004. Taman Negreri Endau-Rompin: Pengurusan, Persekitaran, Fizikal dan Biologi, Jabatan.

Smith, M. A. 1925. A new ground-gecko (*Gymnodactylus*) from the Malay Peninsula. J. Malay. Br. Roy. As. Soc., Singapore 3(1):87.

Utiger, U., N. Helfenberger, B. Schätti, C. Schmidt, M. Ruf, and V. Ziswiler. 2002. Molecular systematics and phylogeny of Old and New World rat snakes, *Elaphe* auct., and related genera (Reptilia, Squamata, Colubridae). Russian J. Herpetol. 9:105–124.

Vogel, G., and J. Van Rooijen. 2007. A new species of *Dendrelaphis* (Serpentes: Colubridae) from Southeast Asia. Zootaxa 1394:24–45.

Wallach, V. 2001. *Typhlops roxaneae*, a new species of Thai blindsnake from the *T. diardii* species group, with synopsis of the Typhlopidae of Thailand (Serpentes: Scolecophidia). Raffles Bull. Zool. 41:39–49.

Wood, P. L. Jr., L. L. Grismer, A. Norhyati, and S. Juliana. 2008. Two new species of torrent dwelling toads *Ansonia* Stoliczka 1872 (Anura: Bufonidae) from Peninsular Malaysia. Herpetologica *in press*.

BOOK REVIEWS

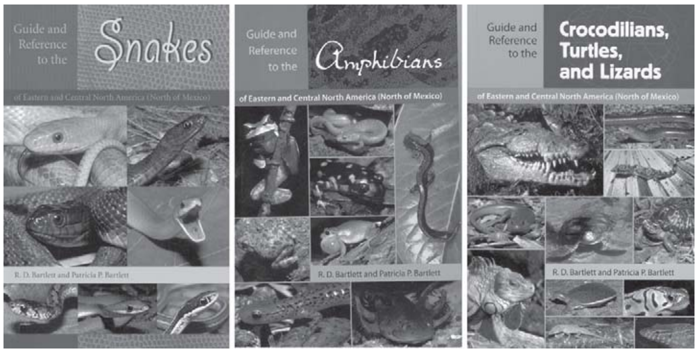
Herpetological Review, 2008, 39(1), 122–123.

Guide and Reference to the Snakes of Eastern and Central North America (North of Mexico), by Richard D. Bartlett and Patricia P. Bartlett. 2005. University Press of Florida (www.upf.com). Softcover. xvii + 342 pp. US $29.95. ISBN 978-0-8130-2935-1.

Guide and Reference to the Crocodilians, Turtles, and Lizards of Eastern and Central North America (North of Mexico), by Richard D. Bartlett and Patricia P. Bartlett. 2006. University Press of Florida. Softcover. xviii + 316 pp. US $29.95. ISBN 978-0-8130-2946-7.

Guide and Reference to the Amphibians of Eastern and Central North America (North of Mexico), by Richard D. Bartlett and Patricia P. Bartlett. 2006. University Press of Florida. Softcover. xv + 283 pp. US $29.95. ISBN 978-0-8130-2950-4.

GAD PERRY
Department of Natural Resource Management, Box 42125
Texas Tech University, Lubbock, Texas 79409-2125, USA
e-mail: Gad.Perry@ttu.edu



Richard and Patricia Bartlett have been busy over the last decade or so. The sheer volume of work is impressive. In addition to the three volumes reviewed here, they coauthored a series of herpetoculture books, each about 50 pages long: three came out in 1999 (all on lizards), four in 2000 (two on frogs and two on snakes), six in 2001 (one on frogs, four on lizards, and one on snakes), one each in 2003 (Poison Dart Frogs) and 2004 (Box Turtles), and three in 2005 (all on snakes). The Bartletts are not newcomers to the field-guide scene either, having published texts about Florida (1998), Texas (1999), the western US (2000), and the Amazon (2003).

The current contributions share a consistent and attractive look, style, and geographic region, but focus on different taxonomic groups. Each offers a brief introduction (about 10 pages); species accounts (about 300 pages); a glossary and a list of additional readings (about four pages each); and an index (about six pages). The greatly similar introductions include information on the use of each book, comments on taxonomy, advice on captive care, some information on typical habitats, and illustrated keys to families. Contrary to what one might expect from authors who have heavily focused on the pet trade, the emphasis here is on conservation. There is discussion of declining species, loss of habitats, and the impacts of invasive species. The authors unambiguously advise against releasing animals in the wild and strongly advocate responsible herpetoculture.

Species accounts tend to be brief, ranging from less than a page to several pages. A nice feature is that the growing number of introduced species is also included, a valuable addition in many places, especially Florida. Each account includes the common and scientific name, both of which mostly follow Crother (2000). Purists may find this annoying, as there have been many proposed changes since then, but I suspect most readers, as the authors intended, will find it a relief to face familiar names. Occasionally, however, they are inconsistent. For example, soft-shelled turtles are all *Apalone* in the species accounts but *Trionyx* in the illustrations of snouts and shells. A range map and at least one photograph are also provided. As the authors point out, "[a]ll species and subspecies and several of the naturally occurring intergrades are pictured. There is no other field guide currently available that provides this important identification tool." Taxa with multiple color forms have multiple pictures, and occasionally this seems to get out of hand, as with the melanistic copperhead illustrated on p 288 (here and below, page numbers refer to the taxon-appropriate book). This appears intended to appeal to the hobbyist, particularly the one interested in the husbandry of unusual color morphs or trying to fill a life-list. Overall, however, this is a useful feature, and my copy of their Texas book is well-thumbed. Pictures are large, typically 1/3 of a page, and – like the distribution maps – are included within the text, rather than in a separate section as in some previous guides. Most pictures are quite good, but the quality varies. Frog pictures tend to be a bit smaller and are sometimes too dark, lacking in contrast, or out-of-focus to be really useful. This is the case with *Bufo americanus charlesithi* (p 17), *Acris c. crepitans* (p 37), *Hyla avivoca ogechiensis* (p 45), and *Rhinophrynus dorsalis* (p 116), for example. Such problems are less common in the other two books but they nonetheless occur, as with the fuzzy picture of *Acrochordus javanicus* (p 18 of the snake book) and *Anolis distichus* (p 243 of the final book), and the poorly contrasted pictures of *Anolis sagrei* and *A. cristatellus* (p 241). In addition to the all-color photographs, black-and-white drawings are occasionally provided (for example, a collared lizard on p 185 or the two salamander species on pages 118-119). These could have been left out, in my opinion, as they contribute little. Serviceable line drawings are used, often in the keys and occasionally elsewhere, to illustrate specific traits or distinguishing characters. Occasionally, as in the drawing of a pitviper head, the level of detail is insufficient: I'm not sure a naïve reader could tell the difference between the black dot representing the nostril and the equal-sized one representing the pit. The distribution maps are crisp and appropriately-sized for the area covered. Unlike some guides they include Canada, and sometimes pieces of Mexico. However, at least the map for the Mediterranean house gecko (p 170) does not include all the current Texas range. The authors may have been using dated sources: the 1987 edition of Dixon's book is referenced, rather than the second edition, published in 2000. In a similar vein, the work of Wilson and Porras (1983) appears in the citations, rather than the more recent Meshaka et al. (2004).

The text which follows offers brief coverage of range and abundance, size, and habitat. Snakes also get an entry on "disposition" and, if venomous, toxicity. Little information on natural history is provided, with most of the text focusing on identifying features. Many of those would require the animal to be in the hand to be discerned, however. Occasionally, a box contains the story of a memorable encounter with the species, printed in different font to indicate its less formal nature. At times, the authors use technical terms that are not given in the glossary (for example, "nuptial" and "distal" in the account of *Agama agama*). At times they use words that do not seem appropriate for the likely audience (e.g., "Lilliputian" [not capitalized for some reason] in the description of the genus *Sphaerodactylus*, "irascible" in describing the Prairie Rattlesnake). Unfortunately, the first paragraph of the preface to the first book I picked up (the amphibian volume) had a problem that is far too common in the age of electronic spellcheckers: the word "know" seems to have been lost, leaving the sentence "But we do that…" rather cryptic. However, I did not find other places with such problems. The index is comprehensive for common and scientific names.

In a review of an Arizona field-guide, Lamb (2007) recently shared a longing for the single-volume "indispensable little green book" such as Conant and Collins (1991) because of its "economy and ergonomic form." Alas, portability and natural history detail were sacrificed here for pictorial comprehensiveness, large font, and well-spaced text. These features, as well as the embedding of the pictures and range maps, and the affordable pricing, make the three books more attractive and easier to read. However, the combined size also makes them less useful as field guides. I can imagine these volumes sitting on the shelf of hobbyists, but I do not foresee well-used copies in the glove-compartments of many professionals.

LITERATURE CITED

Conant, R., and J. T. Collins. 1991. A Field Guide to Reptiles and Amphibians: Eastern and Central North America, 3rd Edition. Houghton Mifflin, Boston, Massachusetts. 450 pp.

Crother, B. I. (ed.) 2001. Scientific and Standard English Names of Amphibians and Reptiles of North America North of Mexico, with Comments Regarding Confidence in Our Understanding. SSAR Herpetological Circular 29. iii + 82 pp.

Dixon, J. R. 2000. Amphibians and Reptiles of Texas: With Keys, Taxonomic Synopses, Bibliography, and Distribution Maps, 2nd edition. Texas A&M University Press, College Station, Texas. 421 pp.

Lamb, T. 2007. [Book review] A Field Guide to amphibians and Reptiles in Arizona. Herpetol. Rev. 38:123–124.

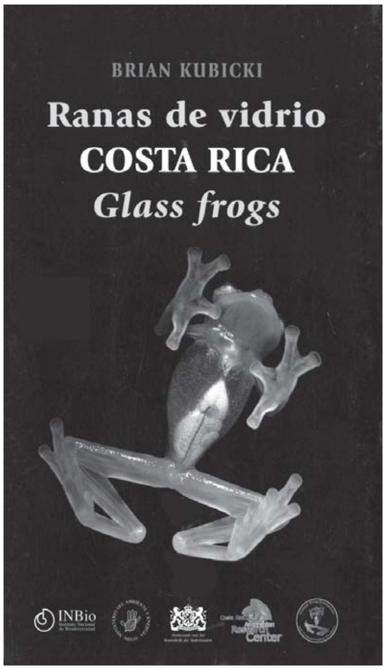
Meshaka, W. E., Jr., B. P. Butterfield, and J. B. Hauge. 2004. The Exotic Amphibians and Reptiles of Florida. Krieger Publ. Co., Malabar, Florida. 155 pp.

Wilson, L. D., and L. Porras. 1983. The Ecological Impact of Man on the South Florida Herpetofauna. Univ. Kansas Mus. Nat. Hist., Spec. Publ. No. 9. 89 pp.

Herpetological Review, 2008, 39(1), 123–124.

Ranas de Vidrio COSTA RICA Glass Frogs, by Brian Kubicki. 2007. Instituto Nacional de Biodiversidad (INBio), San José, Costa Rica (http://www.inbio.ac.cr/en/default.html). 299 pp. Softcover. ISBN 978-9968-927-25-3. US $21.00.

WILLIAM E. DUELLMAN
Natural History Museum and Biodiversity Research Center
University of Kansas, 1345 Jayhawk Blvd
Lawrence, Kansas 66045, USA
e-mail: duellman@ku.edu



With facing pages in English and Spanish and more than 150 color photographs this outstanding book brings to life the Glassfrogs (Centrolenidae) of Costa Rica in a manner that sets an example for the treatment of any group of organisms. The author spent nearly a decade observing, rearing, and photographing Glassfrogs in Costa Rica. In so doing he gathered an enormous amount of data that is the basis for this book that greatly expands the accounts of centrolenid frogs given by Savage (2002).

This paper-bound book of 299 pages (15 × 22.8 cm) is printed on high-quality glossy paper. The first 79 pages contain a brief forward by Jay M. Savage, introductory material, an account of the taxonomic history of the family, and a summary of behavior and ecology of Costa Rican centrolenids. In the introduction, Kubicki thoroughly describes the external morphology of Glassfrogs and illustrates their principal characters. In the section on behavior and ecology, he summarizes information on habitat, diversity, activity, feeding, reproduction, male-male combat, and embryonic and larval development. Throughout these sections he has added his extensive observations to what was known previously (in most cases, very little), thereby providing a comprehensive review of the biology of Costa Rican centrolenids.

The major part of the book (201 pages) is devoted to accounts of each of the 13 species of Costa Rican centrolenid frogs. Each account contains sections on: Holotype, Type Locality, Synonymy and chronological history of the nomenclature, Common name, Etymology, Species group, Description, Similar species, Sex determination, Distribution, Natural history, Call, Calling position, Egg masses and larvae, and Abundance. Although data on morphology used in the descriptions, comparisons, and sex determination are common in recent literature on Glassfrogs, most of the information in the sections on natural history, behavior, and development is new. In the accounts there are 13 distribution maps and 146 excellent color photographs, including 13 photographs of living tadpoles and 22 photographs of clutches of eggs. The account of each species contains no fewer than five photographs of living frogs, including ventral views, pairs in amplexus, and males in combat. Although the calls of each species are described in detail, sonograms and wave forms are not presented.

Kubicki listed 16 species of centrolenid frogs in Central America and noted that all but three occur in Costa Rica. The percentage of Costa Rican Glassfrogs increased with the placement of the Honduran *Hylalinobatrachium cardiacalyptum* and *H. crybetes* in the synonymies of *H. chirripo* and *H. colymbiphyllum,* respectively, by Cisneros-Heredia and McDiarmid (2007). Thus, the only species of Glassfrog extralimital to Costa Rica in Central America is *Hyalinobatrachium aureoguttatum,* a South American species that enters Darién in Panama. There are some minor discrepancies regarding the taxonomy: (1) *ilex* and *prosoblepon* are placed in the genus *Centrolenella,* which is listed as a junior synonym of *Centrolene* by Frost (2007); and (2) *pulveratum* is placed in *Hyalinobatrachium,* whereas the species was placed in *Cochranella* by Cisneros-Heredia and McDiarmid (2006).

Kubicki's "Glass frogs" contains much more information on natural history and has far superior color illustrations than two other recent publications dealing with centrolenid frogs of Ecuador (Cisneros-Heredia and McDiarmid 2007) and Venezuela (Señaris and Ayarzagüena 2005). "Glass frogs" is just one of many outstanding publications produced by the Costa Rican Instituto Nacional de Biodiversidad, an institution that has set high standards for gathering and disseminating data on national biodiversity. Both the author and the institute deserve international accolades.

LITERATURE CITED

CISNEROS-HEREDIA, D. F., AND R. W. MCDIARMID. 2006. A new species of the genus *Centrolene* (Amphibia: Anura: Centrolenidae) from Ecuador with comments of the taxonomy and biogeography of glassfrogs. Zootaxa 1244:1–32.

CISNEROS-HEREDIA, D. F., AND R. W. MCDIARMID. 2007. Revision of the characters of Centroleidae (Amphibia: Anura: Athesphatanura), with comments on its taxonomy and the description of new taxa of glassfrogs. Zootaxa 1572:1–82.

FROST, D. R. 2007. Amphibian Species of the World: an Online Reference. Version 5.1 (10 October, 2007). Electronic Database accessible at http://research.amnh.org/herpetology/amphibia/index.php. American Museum of Natural History, New York (accessed 30 November 2007).

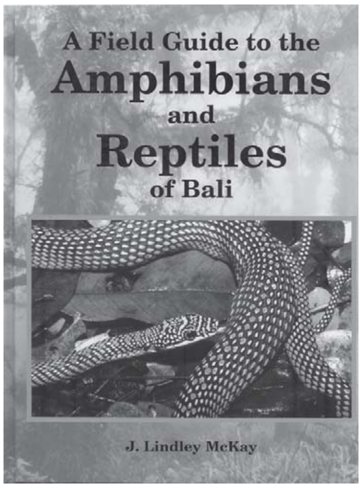
SAVAGE, J. M. 2002. The Amphibians and Reptiles of Costa Rica. University of Chicago Press, Chicago. xx + 934 pp.

SEÑARIS, J. C., AND J. AYARZAGÜENA. 2005. Revisión Taxonómica de la Familia Centrolemidae (Amphibia: Anura) de Venezuela. Publicaciones de la Comité Español del program Hombre y Biosfera-Red IberoMaB de de la UNESCO, Madrid. vii + 377 pp.

Herpetological Review, 2008, 39(1), 124–125.

A Field Guide to the Amphibians and Reptiles of Bali, by J. Lindley McKay. 2006. Hardcover. Krieger Publishing Co., Malabar, Florida (www.krieger-publishing.com). 146 pp. Hardcover. US $39.50. ISBN 1-57524-190-0.

JESSE L. GRISMER
Department of Biology, Villanova University
Villanova, Pennsylvania 19085, USA
e-mail: jesse.grismer@villanova.edu



A Field Guide to the Amphibians and Reptiles of Bali is a detailed overview of the 114 species of amphibians and reptiles known from this tropical island. Bali is a relatively small Indonesian island (128 × 76 km) located 5 km east of Java and 45 km west of Lombok. It is of biogeographical interest in that it lies immediately west of Wallace's Line and represents the most southeasterly extent of the range of many taxa. This book presents the most up-to-date list of amphibians since Iskandar (1998) and the first list of reptiles for the island since Brongersma (1934). The author states that the purpose of the book is to provide an easy identification for any species of the island's amphibians or reptiles. Was this goal accomplished? In many aspects it was, and anyone could use this book to identify most of the herpetofauna of Bali, but it may have fallen short in some areas.

In the introduction the author provides a useful map, and gives an overview of some of the classic works and more recent book length treatments that have been published on southeast Asian herpetology. In Chapter 2, "Making an Identification," the author does a good job in discussing how to make an identification with a specimen in hand and provides diagnostic characters to look for if the specimen was not captured but only observed. This chapter is accompanied by clear and precisely labeled illustrations of frog and tadpole anatomy, turtle shell morphology, and lizard and snake head scalation (lateral, dorsal, and ventral views), and instructions on how to perform certain scale counts. Chapter 3, "Explanation of the species Accounts," provides instruction on how to use the species accounts along with descriptions of the subsections within the species accounts. Chapter 4, "Environment of Bali," gives background on the ecological and geological make up of the island and a detailed description of the different types of habitats, including a list of species that are generally associated with each habitat. This chapter also discusses human impact on the island's natural vegetation.

Chapter 5, "An Overview of Amphibian and Reptile Fauna," is a discussion concerning the evolutionary origins of the fauna of Bali as a mixture of southeast Asian and Papuan elements. Here the author discusses the diversity of Bali compared to that of the surrounding islands and provides a general overview of the different types of lizard and snake ecologies observed on Bali. Chapter 6, "Traditional Uses and the Turtle Trade," is a discussion of the medicinal and commercial uses of the amphibian and reptile fauna. The majority of this chapter is concerned with the use of sea turtles as a food source and documents the growth and decline of this industry. It includes some disturbing anecdotes detailing the harsh treatment of these animals after they have been sold in the marketplace. In chapter 7 "First Aid for Snakebites," the author describes the types of bites one could receive from the different species of venomous snakes (cobras, kraits, pitvipers, and sea snakes) on Bali and the procedures to follow in case of a bite.

The following chapter, "Amphibians, Class Amphibia," begins with a key to all the frog species and their tadpoles; one in English and the other in Bahasa Indonesia. Each is accompanied by several diagrams of frog and tadpole characters useful for identifying certain species. This is followed by the species accounts for frogs. The species accounts consist of one or two small (8.5 × 5.8 cm) photographs of the species and a short (maximum one page) account with four sections. The first, "Description," is an adequate overview of the color pattern, snout–vent length, total length, tadpole morphology, and morphological variation within the species. The next section is "Habitat," describing the type of habitat and microhabitat in which each species occurs as well as its elevated range. Following this is "Biology," which discusses natural history, including activity times, call descriptions, and where tadpoles can be found. The last section, "Distribution," lists the major islands and mainland countries of each species' range throughout southeast Asia. One point of possible contention is that Iskandar (1998) records *Occidozyga lima* on Bali, where here and in Frost et al. (2006), *O. laevis* is the only species of *Occidozyga* recorded.

Chapter 9, "Reptiles, Class Reptilia," is organized similarly to the previous chapter. Each section (turtles, lizards, and snakes) has English and Bahasa Indonesia keys followed by illustrations of head scalation and other relevant morphological characters and species accounts mirror those for amphibians. The main difference between the treatments of the three reptile groups is in the emphasis on diet for snakes. In the appendix, there is a section called "Species Not Yet Recorded on Bali" in which species accounts are given for several species which the author believes have a high probability of being discovered on Bali due to their presence on nearby islands or their widespread distribution in the region. This is an interesting section but the inclusion of these species along with the confirmed species makes the keys unnecessarily long. Following the appendix is a glossary of terms used for describing amphibian and reptile morphology and certain terms in Bahasa Indonesia.

This book does have some shortcomings. The quality of photography makes it difficult to positively identify roughly 10% of the species (e.g., *Occidozyga laevis* [Plate 31], *Rana chalaconota* [Plate 35], *Rana nicobariensis* [Plate 39], *Eutropis rugifera* [Plate 77], *Sphenomorphus temminckii* [Plate 82], *Boiga cynodon* [Plate 94], *Enhydris plumbea* [Plate 106], *Gongylosoma baliodeirum* [Plate 108], and *Rhabdophis chrysargos* [Plate 123]). Many of the photographs are of individuals from outside of Bali. This is normally not a significant concern if the specimen was collected from an adjacent region, but in some cases the specimen photographed is from Australia, Sulawesi, or China where populations may look significantly different. An unfortunate oversight is the lack of a figure showing the details of foot morphology that are diagnostic for many species of reptiles and amphibians. The Literature Cited is divided into categories with citations listed under the category in which each was referenced in the text. This is a bit awkward as the reader must search in several places to locate any particular reference. Finally, without explanation the author chose to recognize recent taxonomic revisions for the Scincidae (*Mabuya*; Mausfeld and Schmitz 2005) and the ratsnakes (*Elaphe*; Utiger et al. 2002) but not for *Trimeresurus* (Malhorta and Thorpe 2004), or the paraphyletic genus *Rana* (Dubois 1987, 1992; Frost et al. 2006).

These drawbacks aside, this book is an informative field guide to most of the amphibians and reptiles of Bali and is a great stepping-stone for future work. It is also an indespensible piece of literature for anyone interested in southeast Asian herpetology in general.

Literature Cited

Brongersma, L. D. 1934. Contributions to Indo-Australian herpetology. Zool. Meded., Leiden 17:161–251.

Carranza, S., and E. N. Arnold. 2006. Systematics, biogeography, and evoltion of *Hemidactylus* geckos (Reptilia: Gekkonidae) elucidated using mitochondrial DNA sequences. Mol. Syst. Evol. 38:531–545.

Dubois. A. 1987. Miscellanea taxinomica batrachologica (I). Alytes 4:61–78.

———. 1992. Notes sur la classification des Ranidae (amphibians anoures). Bull. Mens. Soc. Linn. Lyon. 61:305–352.

Frost, D.R., T. Grant, J. Favovich, R. H. Bain, A. Haas, C. F. Haddad, R. O. De Sa, A. Channing, M. Wilkinson, S. C. Donnellan, C. J. Raxworthy, J. A. Campbell, B. L. Blotto, P. Moler, R. C. Drewes, R. A. Nussbaum, J. D. Lynch, D. M. Green, and W. C. Wheeler. 2006. The amphibian tree of life. Bull. Amer. Mus. Nat. His. 297:1–370 + 1 folding cladogram.

Iskandar, D. T. 1998. The Amphibians of Java and Bali. Research and Development Centre for Biology-LIPI, Bogor.

Malhorta, A., and R. S. Thorpe. 2004. A phylogeny of four mitochondrial gene regions suggests a revised taxonomy for Asian pitvipers (*Trimeresurus* and *Ovophis*). Mol. Phyl. Evol. 32:83–100.

Mausfeld, P., and A. Schmitz. 2003. Molecular phylogeography, intraspecific variation and speciation of the Asian scincid lizard genus *Eutropis* Fitzinger, 1843 (Squamata: Reptilia: Scincidae): taxonomic and biogeographic implications. Organisms Div. Evol. 3:161–171.

Utiger, U. , N. Helfenberger, B. Schätti, C. Schmidt, M. Ruf, and V. Ziswiler. 2002. Molecular systematics and phylogeny of old world and new world rat snakes, *Elaphe* auct., and related genera (Reptilia, Squamata, Colubridiae). Russ. J. Herpetol. 9:105–124.

PUBLICATIONS RECEIVED

***Chamaeleo calyptratus*, the Yemen Chameleon,** Second Edition, by Wolfgang Schmidt. 2007. Natur und Tier-Verlag, Münster, Germany (www.ms-verlag.de). 96 pp. Hardcover. € 18.50 (approx. US $27.00). ISBN 978-3-86659-036-6.

This is an English translation of the original German language popular book on the biology and husbandry of *Chamaeleo calyptratus*, a large and charismatic chameleon in which males bear a huge casque. These impressive lizards have become relatively common in the private collections of terrarium keepers and this book was written to provide guidance to prospective owners, based on the author's years of experience with Yemen Chameleons. Basic information is provided about the evolution of chameleons, the habitat of the Yemen Chameleon, and distinctive features of this and other chameleons (color and color change, casque, tail prehension, independently mobile eyes, tongue protrusion). Advice is provided about selecting and sexing Yemen Chameleons and mating behaviors, copulation, and oviposition are discussed and illustrated by color photos of captive animals. Much of the text is devoted to information about housing individuals of this species (they should be kept alone and provided with good ventilation and adequate possibilities for climbing), feeding them (diet should be varied and vitamin-enriched), and regulating their environment (temperatures should reflect those seen in nature, with both diel and seasonal variation; Yemen Chameleons are particularly suitable for limited free-ranging activity in outdoor enclosures). The book concludes with brief comments on disease and other veterinary concerns and a 66-entry bibliography. This small book serves as a concise guide to *Chamaeleo calyptratus* and its captive care and breeding and is recommended reading for anyone considering maintaining this species in captivity. Unfortunately, most of the references to further sources of information are specific to Germany and this English edition suffers from some instances of awkward translation and some typographical errors. Nonetheless, the book contains a great deal of information gathered over the many years that this species has been successfully bred in Europe and the 75 color photographs and non-technical text make it accessible to a wide audience.

Amphibien an einem Stillgewässer in Peru mit einer illustrierten Checklist der Amphibien und reptilien des unteren Río Llullapichis, by Andreas Schlüter. 2005. Edition Chimaira, Frankfurt am Main, Germany (www.chimaira.de). 347 pp. Hardcover. € 49.80 (approx. US $74.00). ISBN 3-930612-51-8.

This book stems from the doctoral work conducted by Andreas Schlüter at a site on the lower Río Llullapichis in Amazonian Peru. Although the author has published many of his findings in the 30 years since the field work was initiated, this volume represents a comprehensive, if much belated, summary of his study as a whole, which examines the biotic and abiotic causes for the comparatively low frog diversity in the study site—a shaded pond in montane rainforest. The entire metazoan fauna of the pond is characterized and 71 anuran species were identified at the site, 30 utilizing the pond itself. Species accounts are presented for each of these 30 frogs. The information summarized includes synonymy, geographic distribution, size, recognition characteristics, breeding call, diet, reproductive mode and development, activity period, relationship to the pond, and autecological description. These accounts are variably accompanied by line drawings of a selected specimen, a sonogram, distribution map, and black and white photograph(s) of adults, larvae or eggs. These are supplemented by a section of 100 half page color photos illustrating the amphibians of the pond and remaining study site. Niche segregation of anuran mating calls is considered and a remarkable once-a-year mass concert is described and analyzed with respect to its chorus structure. The book also considers responses to predators, the adaptations of eggs, and the ethology and spatial niche partitioning of larvae. A checklist of the herpetofauna of the lower Río Llullapichis is provided and 65 color photos illustrate the reptiles of the area, which are otherwise peripheral to the work as a whole. The book concludes with a literature cited with about 350 entries. Despite the fact that the dissertation upon which this book is based was completed in 1984, the data presented remain valuable, particularly in a comparative light, as other anuran communities in Amazonia have subsequently been investigated. This book will certainly be of interest to amphibian ecologists and ethologists as well as herpetologists with general interests in Amazonia. Short English and Spanish abstracts provide an overview for non-German speakers.

Timber Rattlesnakes in Vermont and New York: Biology, History, and the Fate of an Endangered Species, by Jon Furman. 2008. University Press of New England, Lebanon, New Hampshire (www.upne.com). 224 pp., 12 color illustrations, softcover. US $24.95. ISBN 978-1-58465-656-2.

This highly readable book, targeting a non-professional audience, details the natural history and conservation status of *Crotalus horrridus* near the northeastern extreme of its historical range. In this region, Timber Rattlesnakes are highly endangered, in large part due to a long history of bounty hunting, which Furman covers in fascinating, albeit disturbing, detail. A number of bounty hunters are profiled, and some agreed to be interviewed for the book. Recent and ongoing conservation conflicts are presented, as well as future prospects. The author, an "accomplished naturalist" (from the press release), benefited from collaborations with field researchers William Brown, Marty Martin, and Randy Stechert; their cooperation ensured that the biological information presented was accurate and current. This book is likely to appeal to a broad audience of herpetologists, conservationists, and readers with an interest in wildlife of the northeastern U.S.

Inseparable Friends in Life and Death, the Life and Work of Heinrich Kuhl (1797–1821) and Johan Conrad van Hasselt (1797–1823), Students of Prof. Theodorus van Swinderen, by Charles Klaver. 2007. Barkhuis, Groningen, The Netherlands (www.barkhuis.nl). ix + 105 pp. Softcover. € 25,00 (approx. US

$37.00). ISBN 978-90-77922-316.

The history of herpetology is rife with personalities who made major contributions during very short lives. Among the most tragic of such deaths were those of two young friends who had met at the University of Groningen in the Netherlands, Heinrich Kuhl and Johan Conrad von Hasselt. Both were accomplished and respected zoologists by the age of 20 and were personally acquainted with the leadings scientists of the day, including Cuvier, Lichtenstein, and Temminck. In 1820 both men were appointed as delegates of the Natuurkundige Commissie voor Nederlandsch Indië (Commission for the Study of the Natural Sciences of the Netherlands East Indies) and departed for Java, where Kuhl died in September 1821 and Van Hasselt two years later. In their brief period of collecting, Kuhl and van Hasselt sent thousands of specimens back to Europe, among them 300 amphibians and reptiles of 90 species. Many were new and were described by them in published letters to colleagues in Europe. Another herpetological project of Kuhl's, completed before his departure for Asia, but never published, was the identification of the herpetological illustrations in Seba's *Thesaurus*.

Charles Klaver's biography of Kuhl and Van Hasselt provides a glimpse of the promising careers of the two men, who are unknown to most herpetologists. Although short, the biography and its detailed endnotes, both of which are fully indexed, are a wealth of information about Kuhl and Van Hasselt, their achievements, and the European zoological community in the early 19th century. A bibliography of the 66 publications of the two (1817–1845) is an invaluable tool, as the works of Kuhl and Van Hasselt are among the most miscited in systematic herpetology. A bibliography of more than 230 entries supports the biography. An appendix lists the valid names proposed by Kuhl and Van Hasselt (101 genera, species, and subspecies in all major tetrapod groups, as well as teleost fish and mollusks) and those zoological taxa named in their honor (31). A second appendix is a synopsis of the nomenclatural status of the herpetological names authored by Kuhl and Van Hasselt in their published letters from Java. This includes 32 names (including errors, nomina nuda, junior synonyms and valid names), among them such familiar taxa as *Megophrys*, *Rhacophorus*, *Homalopsis*, and *Ptychozoon*. The book is illustrated by 16 figures (four herpetological) representing portraits, animals, and images of manuscripts and published works.

Inseparable Friends is highly recommended to anyone with an interest in the history of zoology or in the fauna of the Indonesian region. However, it provides particular insight into the herpetological aspects of Kuhl and Van Hasselt's careers (in part because the author's first career was as a chameleon systematist).

Die Nilweichschildkröte *Trionyx triunguis*, by Dieter Gramentz. 2005. Edition Chimaira, Frankfurt am Main, Germany (www.chimaira.de). 166 pp. Hardcover. € 44,80 (approx. US $66.00). ISBN 3-930612-41-0.

This is a German language monographic work on the trionychid turtle *Trionyx triunguis*. It includes a synonymy, list of vernacular names, phylogenetic tree, and an extensive section on morphology, including carapace and plastron size and proportion, osteology, and soft anatomy (largely drawn from the earlier works of Girgis). Coloration, blood chemistry, karyology and both sexual and ontogenetic variation are also reviewed. This large softshell turtle (to 85 cm or more carapace length) has a huge distributional range — from the eastern Mediterranean south along the Nile into East Africa and from Senegal to northern Namibia in West Africa. Most of the author's observations were derived from his own extensive field work in Gabon and especially in Turkey. As might be expected for a species with such a broad range, ecological and reproductive parameters are highly variable. Water bodies of all sizes, flow rates, depths, acidities, and salinities, from small lakes to the ocean are utilized by *T. triunguis*. Data are presented on reproductive parameters (timing, egg and clutch size, hatching success) for populations from Turkey and Gabon. Factors threatening the Turkish populations are considered in some detail. These include habitat destruction, accidental deaths caused by fishing nets and boat propellers, as well as intentional killings by those who perceive the turtles as pests. As of 2005 there were no effective measures of protection, and relict inland populations at Dalyan and Dalaman, Turkey (totaling only a few hundred individuals) were particularly imperiled.

Table and figure legends are given in both German and English and an extensive six page English summary is provided, providing English speakers with access to all of the author's major findings. The book is illustrated with 129 figures, 69 of them in color. Most of these are adequate, but a few (e.g., Figs. 83, 111) are not in focus. In addition to adults, juveniles, eggs, and habitats, photos show dead and damaged turtles reflecting negative human impact on Turkish populations of *T. triunguis*. Numerous graphs and tables present original data relating to activity, nesting, incubation, and other aspects of life history. The bibliography cites 136 references. There are occasional typographical errors in both the English (e.g., scull instead of skull, p. 50) and Latin technical terms (e.g., Opithoticum instead of Opisthoticum, p. 18) but otherwise the text is largely free of errors. This monograph will, of course, be of interest to cheloniophiles, but also to herpetologists and conservation biologists with special interests in Turkey.

A New Herpetological Circular from the Society for the Study of Amphibians and Reptiles
Herpetological Circular 37

SCIENTIFIC AND STANDARD ENGLISH NAMES OF AMPHIBIANS AND REPTILES OF NORTH AMERICA NORTH OF MEXICO, WITH COMMENTS REGARDING CONFIDENCE IN OUR UNDERSTANDING

COMMITTEE ON STANDARD ENGLISH AND SCIENTIFIC NAMES

This is the official name list of American Society of Ichthyologists and Herpetologists, The Herpetologists' League, and the Society for the Study of Amphibians and Reptiles. The list incorporates all taxonomic changes since the 2000 edition. This is a must for anyone working with North American herpetology. 84 pages. January 2008. ISBN 978-0-916984-74-8

Prices: Prepublication (SSAR members only)		After May 31, 2008	US $12.00
first copy -	US $8.00	USA Shipping : First copy	US $2.00
additional copies	US $6.00	Additional copies	US $1.00

Send orders to: Breck Bartholomew, SSAR Publications Secretary, P.O. Box 58517, Salt Lake City, UT 84158-0517, USA. Telephone / fax 801-562-2660; email: ssar@herplit.com. Make checks payable to "SSAR." Overseas orders must be paid in USA funds using a draft drawn on American banks or by International Money Order. Orders may be charged to VISA or MasterCard (Account number and expiration date must be provided). SSAR membership details and a complete list of Society publications can be obtained on request from the Publications Secretary.

anfibios y reptiles del estado de Coahuila, México

amphibians and reptiles of the state of Coahuila, Mexico

Julio A. Lemos Espinal
Hobart M. Smith

CONABIO

Amphibians & Reptiles of the State of Coahuila, Mexico

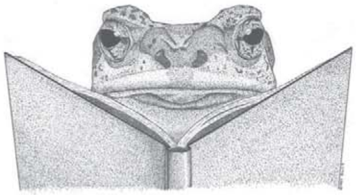
Julio A. Lemos Espinal & Hobart M. Smith

The most comprehensive text on the amphibians and reptiles of the state of Coahuila, Mexico. Contains taxonomic keys, in depth species descriptions, color photographs, gazetteer, dot distribution maps, and localities of collection. Complete text in both Spanish and English

Specifications: xii, 552 pages, 8½ x 11 inches. Paperback (ISBN: 970-9000-40-3) $90.00 + postage.

Bibliomania!

P.O. Box 58355, Salt Lake City, UT 84158-0355, USA
Phone/Fax: +1-801-562-2660
e-mail: breck@herplit.com **Herplit.com**



SSAR Committee Chairs and Coordinators

CHAIRPERSONS

Standard English and Scientific Names
BRIAN I. CROTHER
Department of Biological Sciences
Southeastern Louisiana University
Hammond, Louisiana 70402, USA

Conservation
STEPHEN C. RICHTER
Department of Biological Sciences
Eastern Kentucky University
Richmond, Kentucky 40475, USA

Grants-In-Herpetology
ERIK R. WILD
Department of Biology
University of Wisconsin-Stevens Point
Stevens Point, Wisconsin 54481-3897, USA

JOSHUA M. KAPFER
Natural Resources Consulting, Inc.
119 South Main Street, PO Box 128
Cottage Gove, Wisconsin 53527, USA

Kennedy Student Award
LYNETTE SIEVERT
Department of Biological Sciences
Emporia State University
Emporia, Kansas 66801, USA

Metter Memorial Award
JOSEPH J. BEATTY
Department of Zoology
Oregon State University
Corvallis, Oregon 97331-2914, USA

Meetings
HENRY R. MUSHINSKY
Department of Biology
University of South Florida
Tampa, Florida 33620-5150, USA

Nominating
KIRSTEN E. NICHOLSON
Department of Biology, Brooks 217
Central Michigan University
Mt. Pleasant, Michigan 48859, USA

Resolutions
RICHARD WASSERSUG
Anatomy Department
Dalhousie University
Halifax, NS B3H 4H7 Canada

Seibert Awards
PATRICK OWEN
Department of EEO Biology
The Ohio State University at Lima
Lima, Ohio 45804, USA

Student Travel Awards
DAWN S. WILSON
Southwestern Research Station
American Museum of Natural History
Portal, Arizona 85632, USA

Relations with Herpetologists at Zoological Parks
CLAY GARRETT
Ft. Worth Zoo
1989 Colonial Parkway
Ft. Worth, Texas 76110, USA

Webmaster
RAUL E. DIAZ
University of Kansas Medical Center
Lawrence, Kansas 66160, USA
e-mail: lissamphibia@gmail.com

COORDINATORS

Elector
ROBIN M. ANDREWS
Department of Biology
Virginia Polytechnic Institute
& State University
Blacksburg, Virginia 24061-0406, USA

Symposium Coordinator
RICHARD D. DURTSCHE
Department of Biological Sciences
Northern Kentucky University
Highland Heights, Kentucky 41099, USA

Information for Contributors

Herpetological Review is a peer-reviewed quarterly that publishes, in English, articles and notes of a semi-technical or non-technical nature, as well as book reviews, institutional features, commentaries, regional and international herpetological society news, and letters from readers directed to the field of herpetology. Articles reporting the results of experimental research, descriptions of new taxa, or taxonomic revisions are not published in ***HR***, but should be submitted to the ***Journal of Herpetology*** (see inside front cover for Editor's address). To submit a manuscript to ***HR***, please consult the SSAR webpage at:

<http://www.ssarherps.org/pages/HRinfo.php>

Reprints

Reprints of notes or articles published in ***HR*** should be ordered using the forms provided to authors as the issue goes to press. Proofs are not sent to authors of Geographic Distribution or Natural History notes; these authors should indicate (at time of submission) whether reprints are desired and if so, the appropriate order form will be sent prior to publication. These are also available upon request from the Editor.

Advertising

Herpetological Review accepts commercial advertising. Rates and copy information are available from the SSAR web page (http://www.ssarherps.org/pdf/Adrates.pdf).

Herpetological Review (ISSN: 0018-084X) is published quarterly (March, June, September, and December) by the Society for the Study of Amphibians and Reptiles at Central Michigan University, Department of Biology, 217 Brooks Hall, Mt. Pleasant, MI 48859, USA. Periodicals postage paid at Mt. Pleasant, MI 48859 and at additional mailing offices. **POSTMASTER:** Send address changes to *Herpetological Review*, Allen Press, Inc., P.O. Box 1897, Lawrence, KS 66044, USA. All rights reserved. No part of this periodical may be reproduced without written permission of the Editor. Regarding missing or damaged issues, please notify *Herpetological Review*, Allen Press, Inc., P.O. Box 1897, Lawrence, KS 66044, USA (tel. 785/843-1234 or e-mail: herps@allenpress.com).

SSAR Publications

SSAR publishes seven different series: *Journal of Herpetology, Herpetological Review, Facsimile Reprints in Herpetology, Contributions to Herpetology, Herpetological Circulars, Herpetological Conservation*, and the *Catalogue of American Amphibians and Reptiles* (see below for CAAR details). SSAR members receive pre-publication discounts on all *Facsimiles* and *Circulars* and on volumes in the *Contributions* and *Herpetological Conservation* series. A complete pricelist of Society publications is available at: <http://www.herplit.com/SSAR/>

Catalogue of American Amphibians and Reptiles

The ***Catalogue*** consists of loose-leaf accounts of taxa prepared by specialists, including synonymy, definition, description, distribution map, and comprehensive list of literature for each taxon. Covers amphibians and reptiles of the entire Western Hemisphere. Available by subscription only. See inside front cover to start annual subscription. Use the prices below to order back issues.

COMPLETE SET: NUMBERS 1 – 840		US $460
INDEX TO ACCOUNTS 1 – 400: Cross-referenced, 64 pages		$6
INDEX TO ACCOUNTS 401 – 600: Cross-referenced, 32 pages		$6
SYSTEMATIC TABS (Ten tabs to fit binder: "Class Amphibia," "Order Caudata," etc.)		$6
IMPRINTED POST BINDER (Note: one binder holds about 200 accounts)		$35
INCOMPLETE SET: NUMBERS	1 – 190	$75
	191 – 410	$85
	411 – 840	$320

To order: make checks payable to "SSAR" and mail to Breck Bartholomew, SSAR Publications Secretary, P.O. Box 58517, Salt Lake City, Utah 84158, USA (fax 801/453-0489). e-mail: ssar@herplit.com.

ISSN 0018-084X

The Official News-Journal
of the
Society for the Study of
Amphibians and Reptiles

Herpetological Review

Volume 39, Number 1
March 2008

ARTICLES

TECHNIQUES

AMPHIBIAN CHYTRIDIOMYCOSIS GEOGRAPHIC DISTRIBUTION

BOOK REVIEWS