12th INTERNATIONAL HERPETOLOGICAL SYMPOSIUM ON

CAPTIVE PROPAGATION & HUSBANDRY

NEW YORK - NEW JERSEY METROPOLITAN AREA
June 15-18, 1988

Edited by Martin J. Rosenberg, Ph.D.

PROCEEDINGS OF THE

12th INTERNATIONAL

HERPETOLOGICAL SYMPOSIUM

on

Captive Propagation

And Husbandry

New York - New Jersey Metropolitan Area

June 15-18, 1988

Edited by Martin J. Rosenberg, Ph.D.

Front Cover: Reticulated python (*Python reticulatus*)
"Yellowhead" phase
Photo by William B. Love

Back Cover: African tent tortoise (*Psammobates tentorius tentorius*)
Photo by William B. Love

ACKNOWLEDGMENTS

Being a coordinator for a symposium which attempts to bridge the gap between zoo professionals, scientists, and herp breeders at all levels could be a harrowing experience. Fortunately, my job was made easier by a group of talented and dedicated individuals. I would like to take this opportunity to acknowledge the efforts of those who made the Twelfth International Herpetological Symposium a success.

Dave Hulmes, along with members of the New York Herpetological Society, were instrumental in making most of the local arrangements. Selecting a meeting site in the New York-New Jersey metropolitan area that was within the budgets of most of us was no easy chore. This area of the country was chosen because of the proximity of two of the world's outstanding reptile collections, the Bronx Zoo and the Staten Island Zoo. Dave was responsible for obtaining free entry into both zoos and arranging the Bar-B-Q and the private viewing of Jungle World at the Bronx Zoo and the memorable banquet at the Staten Island Zoo. He also arranged the visit to the American Museum of Natural History and the program by Dr. Richard Zwiefel of their Department of Herpetology. Dave's co-workers who deserve recognition are Bob Doremus for the bus arrangements and mugs, Ernst Hoffman for the logo design, Richard Linke for the reams of information on New York and attractions, and Bill Poueymirou and Adam Zweig for providing refreshments at the hospitality room.

Brian P. Backner and Brett C. Stearns, who co-chaired the Program Committee, were responsible for the quality and variety of the papers presented and the outstanding workshops (six workshops were held, the most yet). They spent many hours organizing a program that was informative and at a level of sophistication we can all be proud of.

Dr. Richard Ross' tribute to Joseph Laszlo at the Staten Island Zoo was greatly appreciated.

I would like to extend a note of gratitude to Richard A. Hahn and Martin J. Rosenberg, who certainly have been instrumental in the success of the symposium throughout the years and who offered me support and the benefits of their wisdom in coordinating this event.

Thanks must go to three wives, Mary Anne Hahn, Pat Hulmes, and Elaine Uricheck, who spent many hours at the registration and sales tables, and who provided support and encouragement to their respective spouses.

It was a pleasure working with all the people who made the symposium a success. For me, it was a truly rewarding and memorable experience.

Finally, compliments are again due to Cyndee Richards, secretary in the Department of Biology at Case Western Reserve University. Cyndee has once again produced a high quality and professional publication of which contributors and symposium attendees can be proud.

Michael J. Uricheck, Ph.D.
Symposium Coordinator

INTRODUCTORY COMMENTS

For both naturalists and conservationists, 1988 held out several terrifying promises. For the first time, "The Greenhouse Effect" and "Ozone Layer" became buzzwords in the American vernacular. Less well popularized is the plight of the tropical rainforests where, in 1988, some 75,000 square miles of the Amazon River basin were slashed and burned. This translates to as many as five to ten species of plants and animals lost through extinction **per week**. Herpetoculture has made prodigious strides in the last two decades, but with long-term habitat destruction and global warming trends, we now face a race against time, with a need to propagate as many species as possible in captivity before the wild populations are eliminated. Sadly, the lost species include many which have not yet been discovered as well as those already known.

Contained in this volume are the thoughts and techniques of many successful herpetoculturists. Use of their experiences, perhaps in modified form, may allow one more species to be saved from extinction. It is toward this end that we have all worked so hard in organizing and running this 12th International Herpetological Symposium and in preparing this volume. The importance of our mission as herpetoculturists cannot be overstated. I hope you will use the information contained within these pages to guide you toward success in your herpetological husbandry and propagation efforts -- and I wish you good luck in this most worthwhile endeavor.

Brian P. Backner, M.D.
Program Co-chairman

CONTENTS

12th INTERNATIONAL HERPETOLOGICAL SYMPOSIUM On Captive Propagation and Husbandry New York-New Jersey Metropolitan Area June 15-18, 1988

SYMPOSIUM COORDINATOR

Michael J. Uricheck, Ph.D.
Department of Chemistry
Western Connecticut State University
Danbury, CT 06810
(203) 797-4232 (Office)
(203) 754-6839 (Home)

PROGRAM COMMITTEE

Brian P. Backner, M.D.
17 Margaret Road
Sharon, MA 02067
(617) 784-7264

Brett C. Stearns, Legal Counsel
California Academy of Sciences
2442 Sharon Oaks Drive
Menlo Park, CA 94025
(415) 854-2099

HOST COMMITTEE

David Hulmes
122 Second Ave.
Hawthorne, NJ 07506
(201) 427-0768

PROCEEDINGS EDITOR

Martin Rosenberg, Ph.D.
Department of Biology
Case Western Reserve University
Cleveland, OH 44106
(216) 368-3558 (Office)
(216) 451-1081 (Home)

EUROPEAN LIAISON

Quentin Bloxam
Curator of Reptiles
Jersey Wildlife Preservation Trust
Channel Islands, Great Britain
0534 61949

AUSTRALIAN LIAISON

Chris B. Banks
Keeper in Charge
Department of Herpetology
Royal Melbourne Zoological Gardens
P.O. Box 74
Parkville, Victoria 3052
Australia
(03) 347-1522

SYMPOSIUM SERIES DIRECTOR

Richard A. Hahn, President
Zoological Consortium, Inc.
13019 Catoctin Furnace Rd.
Thurmont, MD 21788
(301) 271-7488

Copyright 1989 by the International Herpetological Symposium on Captive Propagation and Husbandry
ZOOLOGICAL CONSORTIUM INC. THURMONT, MD 21788

THE SOLAR VIVARIUM
A Reptile Enclosure to Provide Natural Light

Ernst G. Hofmann

The solar vivarium described is designed to provide captive reptiles with unfiltered ultraviolet rays of daylight to an amount determined by available conditions such as latitude, season, compass orientation of placement, and manual control of the mechanics involved. However, during the period when outdoor temperatures are low, an artificial source of ultraviolet rays must be employed. Using a standard window as natural light entry, the solar vivarium offers simplicity in maintenance and can be modified for keeping various types of reptiles. The unfiltered daylight will supply vitamin D3, which is absolutely essential for balancing the complex homeostatic mechanism which prevents rickets in young reptiles and osteomalacia in adults (Phillips 1988). Optimum production of vitamin D3 is stimulated by exposure to 295-305 nm (Takada et al. 1979).

The exposure index (Fig. 1) indicates the degree of sun exposure to be expected according to compass location of the selected window. Latitude of location is of great importance for the degree of efficiency of UV exposure. The duration of the open thermocover (Fig. 2) must be carefully monitored to avoid either chilling or over heating.

Figure 2 illustrates the basic shape of the solar vivarium. The floor should be made of plywood, laminated or varnish coated for moisture resistance. Plywood of 1/4" thickness will do for the sides and top of the enclosure. Plastic windowscreening is recommended for the outward facing side. The indoor front portion of the solar vivarium, made of sliding glass, provides easy access to the interior. The hinged "Thermocover" over the screened area facing the open air (when the window is open) is the most crucial item of the solar vivarium. Plexiglas G-UVT 0.25" manufactured by Rohm and Haas with a transmittance of 0.9% of 250-550nm UV waves (Phillips 1988) is the most desirable material. Unfortunately the price is high and small quantities are virtually unobtainable. Lexan Thermoclear, made by General Electric for greenhouses has a high insulation rating but no UV transmittance. The suggested two sheet mylar frame cover offers a 0.4% transmittance of 350-550 nm waves. With these factors of limited transmittance we must deal with the difficult task of taking advantage of unobstructed UV access into the solar vivarium. The variable of latitude and local climatic conditions for each location must be taken into consideration. Figure 3 illustrates the "winter position A"; with the window down and the thermocover down, only the long infrared waves of the low winter sun will be of benefit.

Towards the warmer season "position B" can be approached gradually and temperature regulation controlled by operating window and thermocover. Maximum exposure to ultraviolet rays with minimum risk can be reached at the summer months with the enclosure at "position C."

The set-up of the interior depends largely on the type of reptile in captivity. A sand, barkmulch, humus mixture 2-4" thick will retain moisture for possible egg deposition and also retain warmth stored during the day. Placing large pieces of bark will provide cover under extreme conditions. The solar vivarium is especially suited for keeping aquatic turtles, but the water should be heated since small amounts are subject to rapid cooling.

There is considerable effort involved in operating this type of reptile housing but what can be more rewarding and aesthetically satisfying than providing natural light and the warmth of the sun for a creature removed from its natural habitat.

LITERATURE CITED

Phillips, J. A. 1988. A Holistic View of Reptile Physiology. Intl. Zoo Yearbook.

Takada, K., T Okano, Y. Tamura, S. Matsui, and T. Kobayashi. 1979. A rapid and precise method for the determination of vitamin D_3 in rat skin. Nutr. Sci. Vitaminol.

452 S. Ridgewood Rd.
South Orange, NJ 07079

Fig.1

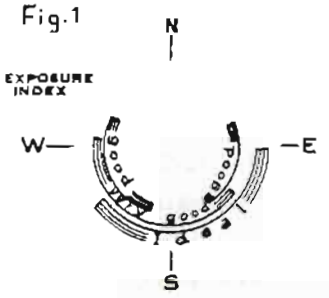
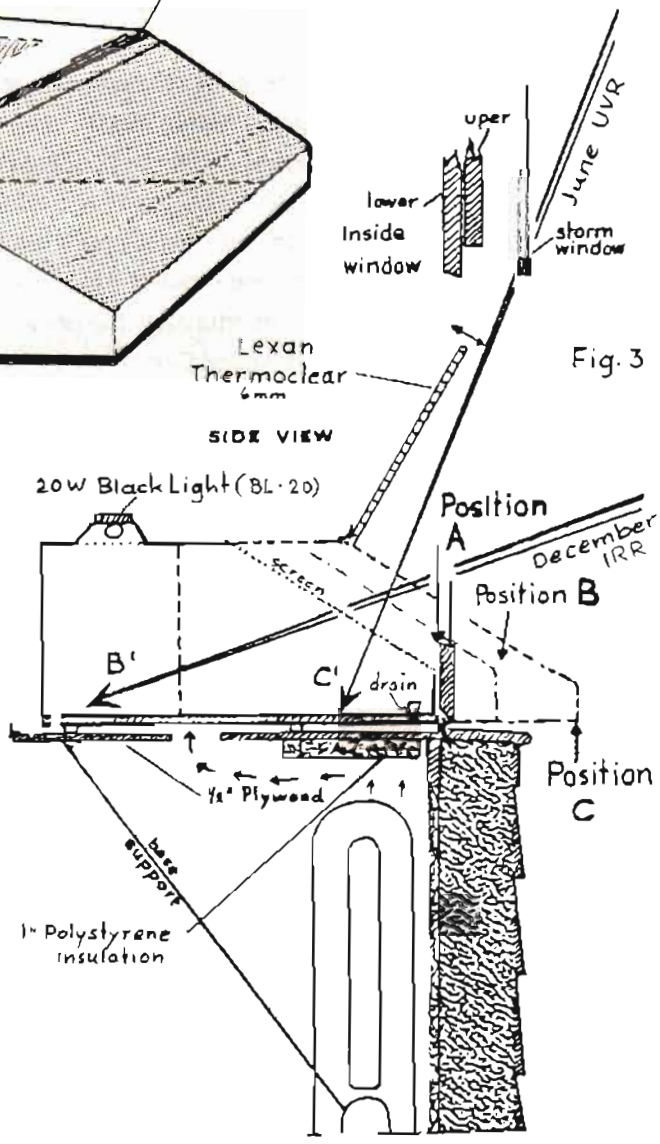


Fig.2

Fig. 3



DO YOU KNOW IF IT'S LEGAL?

William B. Allen, Jr.

I am not going to elaborate on the federal laws. These involve the Endangered Species Act, which covers federally endangered and threatened species of amphibians and reptiles. I am also not going to discuss the SSP (Species Survival Plan) in which participants can exchange captive-bred specimens. Both of these systems require permits, usually from both the sender and the receiver.

Each state has its own listing of species which will fall under one of the following categories: endangered, threatened, or species of special concern. Thus, some reptiles and amphibians do not have to be federally protected -- they are protected under the state laws.

Most states' rules and regulations require the collector to have a scientific collecting permit, which is usually available free or at a very nominal cost. This allows anyone with an interest in herpetology that wants to do a study to legally pursue it. However, permits are not granted to individuals who just want an animal as a pet. You must have a justifiable reason, such as scientific study or a propagation project. It often helps to offer a little extra on your part, such as the release of any surplus young that may be derived from your propagation program or studies.

As I mentioned, each state has its own regulations; but adjacent states may vary in their laws. The following chart demonstrates this. I have used nine species and four neighboring states to show the variation.

SPECIES	PA	NY	NJ	OH
Massasauga	END.	END.	NA	NP
Tiger Salamander	END.	END.	END.	NP
Bog Turtle	END.	END.	END.	NA
Blandings Turtle	END.	TH.	NA	NP
Eastern Mud Turtle	END.	NA	NP	NA
Spotted Turtle	NP	SC	NP	END.
Green Salamander	END.	NA	NA	END.
Timber Rattlesnake	SC	TH	END.	NP
Blue-Spotted Salamander	NA	SC	END.	END.

KEY: END. Endangered TH. Threatened SC Special Concern
NA Not Applicable NP Not Protected

Most states will not allow you to keep an endangered species, regardless of its origin. This means that you cannot bring an animal into a state that protects it, even though you obtained that specimen from a state where it was not protected.

For example, you cannot go into Ohio and collect massasaugas or tiger salamanders and bring them back into Pennsylvania, because they are on the endangered species list in Pennsylvania. There is a valid reason for this. You could very easily obtain a letter from a friend saying that he had given you a specified number of a certain reptile or amphibian that were protected in your state but not in his. These could die or be sold and you could then go out on your own and pick them up in your own state and use his letter as proof that he had sent them to you, should you be caught with them.

There are also laws in each state directing whether or not you can sell the animals or transport them from the state, as well as laws regulating the importation of animals in the state.

What I am trying to do here is to tell you not to rely solely on federal regulations, but to check with the state before you collect or ship anything. You can be in as much trouble with the state regulations being broken as you can with the federal regulations.

And I think if you read between the lines of the state regulations you will see that states also discourage collecting species of special concern.

As I mentioned before, it is not hard to obtain a permit if you have a valid reason. Permit requirements in most states require you to turn in a report at the end of the year on what you have collected and the disposition. Reports from the Pittsburgh Zoo include: kind and number of species seen, what was caught and kept, what was caught and released, and any other items that we feel may be of interest to the biologist that issues the permits. After all, this is a useful way to keep track of which species may be getting scarce and which are overabundant. Permit reports provide information which may suggest the need to limit (or <u>not</u> limit) collecting by private individuals.

Another thing I highly recommend when you collect in an area is that you get in touch with the local law enforcement officer and let him know that you are collecting in the area. There are two good reasons for this. One, he may be able to direct you to a better area for collecting than the one that you chose. Who knows the good areas better than the people who work in those areas? Two, an officer appreciates a call early in the day or evening so that he won't have to get out and go looking should he get a call at eleven or twelve o'clock at night that someone is out prowling in a marsh or field with a flashlight. He would know that it was you because you had called and let him know. That kind of consideration is well received by the field people. It shows that you are thinking of them. This is the kind of stuff that is taken into consideration when you meet them in the field. I should know -- I was a law officer for 20 years with the Game Commission.

These laws are not on the books to put the hobbyist out of business. They are designed to protect the remaining reptiles and amphibians from the commercial trade, both large scale and small scale. Herpetologists want to protect these animals and the law is trying to help us.

So, by taking a little time to write and get permits, you may be saving yourself from a night in jail and, possibly, a heavy fine.

The Pittsburgh Zoo
P.O. Box 5250
Pittsburgh, PA 15206

REPTILE ENCOUNTERS AT AUDUBON ZOO

Andrew T. Snider

In 1924, Audubon Zoo officially opened its doors to reptiles and amphibians. The Odenheimer complex, as it was and still is called, featured other animals in addition to herps, including fish, small mammals, and a separate facility for seals and sea lions. As was the custom of the day, animals were simply maintained. Little, if any, thought was given to long-term maintenance or propagation of specimens. Many native Louisiana species were exhibited with a smattering of exotics such as matamatas, boa constrictors, and an occasional python. Many years and many specimens later, much of the complex was closed to the public for remodeling into major parts of the zoo's master plan.

Next came the Louisiana Swamp exhibit. This facility opened in April 1984 amidst a thunder of applause from visitors and the zoo community as a whole. Heralded by the AAZPA as the Outstanding Exhibit of the Year, 1984, people flocked to the zoo to see swamp-dwelling animals residing in a true-to-life bayou setting.

An innovation of this remarkable exhibit is the use of the area culture to demonstrate how man and animal can coexist happily utilizing the same habitat. Twenty or more species of reptiles and amphibians are exhibited here including three-toed amphiuma, *Amphiuma tridactylum*, alligator snapping turtles, *Macroclemys temmincki*, as well as normal and leucistic forms of the American alligator, *Alligator mississippiensis*. Nine specimens of "white alligators" currently reside at Audubon Zoo. The first four were donated by a Cajun fisherman on 5 September 1987. Since then, five more from the same nest were placed on loan to the zoo by a private citizen and L. L. & E. (Louisiana Light & Electricity). Growth studies are currently underway concerning these remarkable animals and basking behavior will be the subject of future research.

The staff of the Louisiana Swamp Exhibit serve as experts on native Louisiana wildlife, thus providing a valuable service to the New Orleans general community. In addition, they work closely with the Gopher Tortoise Council to relocate tortoises brought to the zoo as donations.

In 1986, plans were finalized for what was to become the present day Reptile Encounter. Work progressed quickly and on 5 September, 1987, Audubon's newest major exhibit opened to an enthusiastic public. The facility is composed of 73 indoor exhibits and an outdoor entry exhibit for Chinese alligators, *Alligator sinensis*. The outdoor architecture was specifically designed to resemble that of the adjacent 1924 Odenheimer Complex.

The interior ambiance in the public area of Reptile Encounter is heightened by the use of low light levels, carpeted floors and walls, and acoustically tiled ceilings. Visitors are further assured of a pleasant visit by the comfortable climate-controlled atmosphere. Back-lit graphics throughout the building explain interesting facts about individual species as well as educational aspects of herpetological interest.

The exhibit area is divided up into several special hallways. Upon entering the building, the visitor finds him/herself in the Hall of Giant Reptiles. Green anacondas, *Eunectes murinus*, are the first to greet visitors with above and below water viewing of these magnificent animals. Several species to which Audubon has long-term commitments are also included in this area. The Dumeril's ground boa, *Acrantophis dumerili*, one of three SSP species found in Reptile Encounter, is located here. Also located in this area is a 45 x 20 ft exhibit for false gharials, *Tomistoma schlegeli*. The breeding group of this impressive crocodilian consists of 1.3 individuals. Two 6 ft-deep pools are located here as well as two heated nesting chambers and an off-exhibit quarantine pool area. Numerous colorful birds also inhabit this lush tropical stream setting. Several large lizard and tortoise species round out the first phase of the visit.

The Hall of Adaptive Animals was conceived as a means of teaching visitors various principles concerning herptiles. Nocturnalism, hibernation, and parallelism are all addressed here with various interesting exotic species. Matamatas, *Chelus fimbriatus*, and prehensile-tailed skinks, *Corucia zebrata*, are just two of the many species exhibited here. Reptile Encounter's first successfully bred species also resides in this area, the Madagascar giant day gecko, *Phelsuma m. grandis*. In March 1988 two specimens of this beautiful reptile hatched and it is our intention to continue propagating this interesting species. Three of our adult breeder males were confiscated, wild-caught specimens which add to the genetic integrity of the species in North American collections.

The next stop is the Hall of Venomous Reptiles. The dark, Chinese form of the king cobra, *Ophiophagus hannah*, greets visitors amidst his bamboo-strewn exhibit. West African gaboon vipers, *Bitis gabonica rhinoceros*, and reticulate gila monsters, *Heloderma s. suspectum*, are two species that have shown reproductive behavior in this area of the building. Two specimens of the bushmaster, *Lachesis muta stenophrys*, have recently been acquired for future reproductive studies.

The next area of the building explains the differences between four of the five classes of vertebrates: Amphibians, reptiles, birds, and mammals. This exhibit hallway is appropriately called What's in a Name? An egg display also helps the visitor understand the differences and similarities between these four groups.

A special exhibit containing highly endangered Atlantic Ridley sea turtles, *Lepidochelys kempi*, is next. The 6700 gallon coral reef tank contains 1.2 specimens of this impressive chelonian. Special graphics explain the plight of this species and the need for continuing efforts in support of legislation concerning the use of Turtle Excluder Devices, TEDs, by shrimpers in the Gulf of Mexico. It is hoped that by exhibiting these beautiful animals, Audubon can help the species through public education.

Twelve species of amphibians are highlighted in the next area of Reptile Encounter. Grotesque, unusual animals like the hellbender, *Cryptobranchus alleganiensis*, and tiny jewel-like frogs inhabit these exhibits. It is here that much future work will be done in the area of captive husbandry and reproduction. Presently, a group of 15 golden mantella, *Mantella aurantiaca*, are being tended to in anticipation of upcoming research. The blue and yellow dart poison frog, *Dendrobates tinctorius*, is a beautiful species with several distinct races. Audubon will be working with both the Surinam and French Guiana races of this variable and spectacular animal.

The grand finale of the tour is a simulated rock canyon containing 22 exhibits of one of nature's most impressive animals, the rattlesnake. Rattlesnake Encounter shows the visitor that these animals are not to be feared, but admired. Animals can be viewed on rocky outcroppings near your feet as well as on ledges above your head. This area is divided up into two sections: A warm side for tropical and desert-dwelling species and a cool side for temperate and montane animals.

A major innovation in Reptile Encounter is the use of refrigeration units in place of air conditioning in all "cool rooms," including Rattlesnake Cool, the Amphibian Room, and the hibernaculum. This allows for higher humidity than is possible with typical air conditioning. A continuously playing tape explains the natural history of the rattlesnake as well as many interesting aspects of these animals' lives. It ends with a plea for conservation, not destruction, of these spectacular examples of nature's wonders.

Behind the scenes Reptile Encounter is very impressive in its spaciousness and well-planned facilities. Thirteen large exhibits are monitored continually for temperature and humidity by computer. Photoperiods are also computer-controlled according to the animal's natural range. Smaller fiberglass modules are controlled by individual timers. Concrete floors and easily-cleaned trough drains make sanitation and cleanliness an easy matter. A tool room/shop area allows for continuous maintenance of facilities without having to heavily rely on the already overtaxed maintenance department. A live-feed room was provided to assure continuous live rodents, insects, etc. for those species that require those items. Large supplies of frozen food items are

also maintained in the spacious kitchen facilities. Separate large holding areas are available for off-exhibit venomous and non-venomous species. In this way, large groups of individual species can be obtained, held, and bred for proper genetic management of all potential long-term breeding programs.

All aspects of Reptile Encounter were designed with safety in mind. Animal safety is assured by an electronic security alarm system. Shift cages are utilized wherever they are needed, including large nonvenomous as well as venomous species. All rooms with the potential for holding venomous reptiles are equipped with snakebite alarm buttons. Annunciator panels then tell assistant personnel in which area of the building the bite has occurred. Nine varieties of snakebite sera are maintained at all times in the kitchen facilities of the reptile building. Several physicians from a local hospital are on call for all snakebite emergencies. All personnel working with venomous reptiles utilize a "red tag system." A portable red tag is placed on each venomous enclosure being worked. In the event that a snakebite victim passes out after being bitten, it would then be possible to tell which animal inflicted the bite. A snakebite procedure and an extractor kit are also located in each venomous-animal room.

Separate quarantine and hospital facilities are also provided in Reptile Encounter. All new specimens must be quarantined for a period not less than 30 days, during which time two fecal exams are taken to detect the presence of parasites. The hospital facility is used only for potentially pathogenic and/or infectious diseases. Both rooms have separate air conduction systems and entry ways from the rest of the building. A hibernaculum is also available for all temperate species as well as those that need a certain "cooling down" period for optimal health and/or reproduction. Veterinary care for the entire collection is provided by an on-staff veterinary team. Excellent health care is therefore assured.

Research has already begun in the new facility. Paramyxovirus work has recently started in conjunction with the Houston Zoo and Dr. Elliott Jacobson of the University of Florida. This lethal virus has recently been shown to affect many species of reptiles including colubrids, boids, and elapids as well as the viperids, which were the first group shown to be affected. Male hormone levels of selected species will be tested in an upcoming project to detect reproductive peaks. It is hoped that various species that have thus-far been difficult to reproduce in captivity will someday be bred routinely through work such as this.

The Education Department at Audubon Zoo has routinely called upon the reptile staff to give lectures and demonstrations to school groups, TV audiences, and other members of this community. In addition, the staff is used as a constant source of information to the general public via seemingly continuous phone calls.

The future holds many interesting and exciting possibilities for the reptile department. It is hoped that within the next year, seven tuatara, *Sphenodon punctatus*, will be shipped by the New Zealand government to Audubon Zoo for future reproductive studies and possible exhibition. A total of five specimens of this "living fossil" are currently in the U.S. at the St. Louis Zoo and with more specimens possibly coming to the Audubon and Dallas Zoos, it is hoped that much will be learned about these wonderful creatures.

An upstairs breeding area will be constructed within the next two years to hold a variety of reptilian subjects which would otherwise not be worked with due to lack of space. Those of us at Audubon Zoo await these and other exciting possibilities. It is hoped that in the future, Audubon will be known not only as a nice place to visit, but as an excellent breeding facility for many species of endangered or otherwise threatened reptiles and amphibians.

Herpetology Department
Audubon Park and Zoological Garden
P.O. Box 4327
New Orleans, LA 70178

DENDROBATID FROGS: A WORKSHOP

Dale Bertram, M.D.

INTRODUCTION

How Popular is the Keeping of Dendrobatid Frogs as a Hobby?

Europe: Extensive interest – especially in the Netherlands and West Germany. The average European hobbyist has much more experience keeping these animals and has easier access to specimens (especially captive-bred specimens). I estimate that there are 200-300 "serious" collectors in West Germany and 100-150 in Holland. ISSD (The International Society for the Study of Dendrobatid Frogs) has members in Belgium, West Germany, Holland, Sweden, Switzerland, East Germany, Scotland, England, and Czechoslovakia.

U.S.: I estimate that approximately 50-60% of those hobbyists in the U.S. that express an interest in these animals are only nominally interested. After nearly a year of formal, and word-of-mouth, advertising, the U.S. membership of ISSD stands at only 55 (total membership = 86).

Asia: Attempts to advertise ISSD in Japan have so far yielded no responses.

South America: There are numerous "professional" herpetologists who are interested in dendrobatid frogs working in South America. "Hobbyists," as the term is used in the U.S., should be listed on Appendix I of CITES. ISSD has one member living in Colombia.

Africa: ?

Why Such Limited Interest?

1. Misconceptions regarding the difficulty of working with these animals.
2. Poor availability and high prices of specimens.
3. Past bad experiences on the part of those first attempting to get started; i.e., high mortality and expensive "pet trade" acquired specimens.
4. Lack of knowledge of principles of husbandry and breeding, especially basic "how to get started" concepts.

In This Workshop We Hope to be Able to Address at Least These Four Points

I. MISCONCEPTIONS

1. **Dendrobatids are very delicate animals.**

I have heard this general statement expressed all too often. These animals are beautiful and very photogenic. Most hobby-oriented books have at least a few photographs that never fail to stir interest. Because of their beauty, relative unavailability, and their unwarranted reputation of being poisonous and therefore dangerous, they have acquired a mystique. Like most generalities, this statement is too broad. Some species are in fact quite delicate; *Dendrobates histrionicus* is, in my opinion, not a beginner's frog. Others, for example *Dendrobates auratus*, are quite hardy and can tolerate a few mistakes. In any group of herps there are those species that are delicate and those that are not.

2. **Only "experts" can get these animals to breed.**

This is completely false. Often someone who has managed to get them to breed is considered an "expert" solely on the basis of this success. The truth is, if you can keep them alive for a year or two, they will breed eventually despite what you do or do not do for them.

3. **It is nearly impossible to get good, healthy specimens.**

It is difficult, but not impossible. Dendrobatids have been offered for sale less and less often the past few years. The recent addition of dendrobatids to Appendix II of CITES is likely to make this situation worse. If a person really is serious about establishing a breeding colony, he/she must go about it with the right attitude. The "pet trade" specimens will have some unavoidable mortality; this is simply a fact that must be dealt with. If you want a pair - buy four or six - and be satisfied if you end up with the pair you sought. Whenever possible buy captive-bred specimens from someone you know and trust.

4. **Dendrobatids do not ship well.**

Here again is a broad generalization; while it is not true that they *do not* ship well, it is all too often true that they *are not* shipped well. They must be very well protected during shipment, especially from temperature extremes. Unless you are willing to put up with higher losses, I do not recommend shipping them in the winter. The **24 hours and it is yours** guarantee offered by most wholesalers is inadequate. Shipping stress produces mortality for up to two or three weeks after the actual shipping. Do not be afraid to demand your own shipping specifications when ordering these animals; if a dealer is unwilling to comply with your specifications, then do not buy from him.

5. **Dendrobatids are endangered by over-collection and habitat destruction and therefore trafficking in them should be discouraged. This fact is supported by their recent addition to Appendix II of CITES.**

There is no scientific evidence to support this generalization. The addition of dendrobatids to the CITES list was done without the benefit of research that supports the notion that they are, in fact, threatened by trade. You are referred to the editorial entitled, "The CITES Conservation Circus" (N. Mrosovsky, 1988. Nature Vol. 331, 18 February). Preservation of species within genetically healthy captive populations is an expressed goal of ISSD. This can only be accomplished if access to wild populations by those with legitimate interest in preservation can be continued.

6. **No good "How-To" books on their care are available.**

Unfortunately, much of the literature pertaining to the husbandry and propagation of dendrobatids is written in German or Dutch. An excellent book written by a West German has recently been published in English and I highly recommend it! The title is: **Breeding Terrarium Animals**, by Elke Zimmermann. Also, the **ISSD** newsletters are extremely informative.

II. POOR AVAILABILITY AND HIGH PRICE OF SPECIMENS

Recently, frogs from Surinam and Guyana have been showing up on the American market; specifically, *D. tinctorius* and *D. trivittatus*. The price for *D. tinctorius* ranges from $25 - $50 each. I have received numerous phone calls from people who have purchased *D. tinctorius* from Florida wholesalers in the past few months. There seems to be significant problems with "nose rot" in these recent shipments.

D. auratus is commonly available for $25 - $40. It is feared that most of the adult *D. auratus* available are wild-caught frogs from Hawaii. Inasmuch as the geographic range of this species is quite limited in the Hawaiian Islands, over-collections could lead to future problems. No data exist on the effects of collection within the Hawaiian populations. *D. auratus* is a very commonly bred species and captive-born babies are relatively easy to find and are usually fairly inexpensive.

The availability of dendrobatids in Europe is greater than it is in the U.S., although the prices are at least as high if not higher. This is due to the large numbers of successful breeders. In addition to the commercially available animals, there is a significant amount of trade among fellow collectors. Getting to know the "right people" is an important part of specimen acquisition. ISSD is in the process of establishing a "Breeders Network" to facilitate sales, trades, and breeding loans among its members in the hopes that this friendly trade will result in more, better, and less expensive specimens for all.

III. PAST BAD EXPERIENCES

Do not underestimate the power of word-of-mouth as it circulates within herpetological circles. All too often I have heard the account of how some person stuck his neck out and spent $200 or $300 on "poison arrow frogs," only to watch them all die within a week. Stories like these are often heard at state herpetological society meetings. The result of the recounting of these bad experiences is the development of the mystique that I mentioned earlier. Often though, if you quiz the person telling such a tale, you will find out that the whole misadventure was actually an ill-conceived idea, poorly executed.

The buyer of dendrobatid frogs should not be an impulse buyer. I must confess that if I went to my mailbox and found a herp price list and a letter from the Pope, I would know which dendrobatids were listed before I got the scoop on the latest ecclesiastical developments. If you get one of these lists, notice that "Red & Black Poison Arrow Frogs" are listed, think to yourself, "I would sure like to give these a try," and then quickly try to set something up for them while you await the shipment, you might as well flush your money down the toilet. It is better to select a species, learn about its requirements, set up a suitable habitat, and then set out to purchase the animals. Do not waste these frogs!

IV. HOW TO GET STARTED

The first thing that the would-be keeper of dendrobatids must do is decide that he/she is willing to make the commitment that this hobby requires. It is not enough that you are intrigued by these animals and that you have decided that you would like to try keeping them. You must know what you are getting yourself into. You will need to be able to sustain an environment that does not come naturally in most of our homes. You will need to know how to, and be able to, produce a consistently reliable supply of small live food insects (and fruit flies by themselves are not enough). You should be willing to anticipate some losses when deciding what you want to buy; i.e., buy four to get two. Everything must be ready *before* the frogs arrive. A well-thought-out commitment will pay off in a hobby that is satisfying rather than frustrating.

The Specifics

Habitat: Since time and space are limited, we will discuss only the generic tropical terrarium. This will suit many of the species which are readily available including *D. auratus*, which is the species that I recommend starting with. The basic factors which must be considered are: temperature control, humidity control, ventilation, population density, and stress minimization. I do not think that lighting is a critical factor.

Temperature control, humidity control, and ventilation are all related. It is difficult to provide daytime temperatures of 78-80°F, night time temperatures of 68-76°F, and 80% - 100% humidity in a tank that has a screen top for good ventilation unless you live in a greenhouse. A single terrarium in your living room will require compromises that in the long run will lead to disappointing results. I suggest a separate "frog room"; I have two of them in my basement. The rooms are insulated completely and the walls and ceiling are covered with plastic to retain the humidity. They are heated with small non-fume producing space heaters and humidified with rolling belt type room humidifiers. The various climate control devices are all set on timers and the whole

operation runs fairly trouble free. Because the rooms themselves provide the right temperature and humidity, the tanks all have screen tops and the ventilating fans just circulate the healthy jungle air. The tanks should get a good sprinkling everyday and an automated system to accomplish this is a real time saver. Incidentally, this type of environment is great for growing pin-head crickets as well.

Population density is important. It is not necessary to have big tanks (though I prefer these myself) but the tanks should not be crowded. If 15 gallon tanks are used, only one pair of frogs should be kept per tank. This fits right in with the last concept, stress minimization. Overcrowding causes stress.

The most stressful thing for a frog is its owner. Once they are in their happy home, leave them alone. Resist the temptation to roust them out to admire their beauty or show them off to your friends. A frog that is handled will go into hiding for several days. So, if every few days you pick them up to admire them, they will always be stressed. If you never bother them at all you will find that they will be out and about in plain sight quite often. Give them plenty of covered hiding places and plant the terrarium densely.

Food: The keeper of dendrobatid frogs should culture both fruit flies and crickets. If you can culture springtails or other small insects, the added variety will be beneficial. The culturing of food insects must be attended to *religiously* and is, in fact, the major work of keeping dendrobatids. Before deciding to keep these frogs, you should think this over carefully. When it is -20°F in the middle of January and you run out of pin-head crickets, there are not many options open to you – and even fewer to your frogs. Insects should be dusted with vitamin supplements; I do that every other day. Supplementing the diet with "pasture plankton" in the summertime is a good practice. I do not think that you can feed them too much; obesity is not a common nutritional problem among amphibians in most collections.

One Virginia Terrace
Madison, WI 53705

HUSBANDRY AND BREEDING OF Agalychnis callidryas AT THE NATIONAL ZOO

Cecilia Chang

DESCRIPTION AND NATURAL HISTORY

Agalychnis callidryas (red-eyed treefrog) is a medium-sized treefrog found throughout Central America. The general coloration is a green dorsum, ranging from a leaf-green to a dark green, with some specimens sporting small white spots on the dorsum. The underside is a creamy white color. The flanks are blue with bars of yellow-orange. The hands and feet are orange. The most prominent feature is, of course, the bright red eyes, hence the common name, red-eyed treefrog.

Snout-vent lengths for males range between 40 - 50 mm; females are slightly larger, between 60 - 80 mm. Both sexes have a streamlined appearance, with the exception of females during the breeding season.

Field notes from observations recorded in Gamboa, Panama, in May-June 1986, and April-June, 1988, are summarized below.

Agalychnis callidryas is a nocturnal creature and has been known to breed throughout the wet season (April-November), but with pronounced activity in May-July. During the rainy season, colonies appear at various pools and ponds. Shortly after sunset, the males call from the trees above and descend to the ponds below. The call sounds like a series of two to three "clucks." Females appear later in the night to breed.

They are most abundant when rains have occurred throughout most of the afternoon. Sunset in Gamboa, Panama occurs around 1900 h. Females have been spotted at breeding sites no earlier than 2100 h, with the peak of breeding activity culminating at approximately 2400 to 0300 h.

Prior to amplexus, males call from the treelimbs or bushes overhanging the water. The females approach the calling males to amplex. Once in amplexus, the pair moves about the trees, and several clutches are deposited on the underside of leaves. The pair will move down into the water first, before climbing to the appropriate egg deposition site.

Although *Agalychnis callidryas* usually deposit egg clutches in the trees above, they will seek out any other available areas as long as they are close enough to the water. At the beginning of the rains, the ponds have yet to fill up and expand out to the tree-lined shore, and most of the overhanging branches are far out of reach from the ponds. Under such conditions, one can find *Agalychnis callidryas* on the ground in amplexus, with egg deposition often taking place on thin grasses or twigs on the edge of the pond. Many times, these clutches are deposited in a string, rather than in a mass, and adhere to the twigs or grasses. Later in the season, when the ponds have reached the surrounding forest line, the frogs move up into the trees and egg deposition will then occur within arms reach above the water.

During the breeding season, both sexes give off a pungent odor in certain situations. Single males, when harassed, will release this odor. An amplexing pair will not smell; if they are forcibly separated, however, the male will smell. Females after egg deposition will also smell. Presently, there is a study being conducted on the circumstances surrounding this odor, and there is some evidence that it is some type of release response.

The eggs deposited by *Agalychnis callidryas* are encased individually in a membrane and clustered in a gelatinous substance. The gel is sticky to the touch and adheres steadfastly to any surface. The eggs vary in color from a creamy white to a light blue. Based on random estimates at various sites, each clutch size varies from as few as 12 to as many as 70 eggs. Eggs within a clutch range from 0 to 100% fertility, with the average at about 70%. Generalized observations point to smaller clutch size at the beginning of the rainy season (April) and larger clutch size at the height of the rainy season (June-July).

Embryonic development can be first detected by sight on day 3 from egg deposition. By day 5, the embryos can be seen swaying and squirming in the egg capsules. Tadpoles hatch on day 7. The hatching process can be best described as a three-second mass squirming frenzy. Once the first tadpole hatches, all of the rest of the clutch will be out in three seconds, flipping and sliding down the leaves and landing in the water below. Once the tadpoles have landed in the water, they are fairly inactive for the first few days. Later, they can be seen feeding at the surface.

In the ponds that are breeding sites for *Agalychnis callidryas*, there are also numerous other amphibians breeding. Common species found are *Bufo marinus, Leptodactylus pentadactylus, Hyla rubra, Hyla ebraccata, Hyla rosenbergi, Phrynohayas venulosa, Physalaemus pustulosus, Smilisca phaeota*, and some of the *Centrolenella* species. It is unclear as to exactly who is eating whom in these ponds, but it can be assumed that *Leptodactylus pentadactylus* tadpoles will probably devour many of the above species of tadpoles, including *Agalychnis callidryas*. Other predators found at these ponds are fish, insect larvae, insects, and birds. Based on field data from a study of *Physalaemus pustulosus* tadpoles' survival and growth rates in the wild, the natural mortality of all tadpoles is extremely high. With the disappearance of water in temporary pools, and as well, the high number of predators, the tadpole survival rate is low, compared to the overall high rate of hatching success.

HUSBANDRY

This species, until recently, has been considered a difficult species to maintain in captivity due to its seemingly delicate nature. When they are disturbed, they seem to succumb more easily to stress and its debilitating consequences than other species. Thus, our experiences with medical treatments of any sort often prove to be fruitless once an illness is detected. However, when left alone and properly maintained, they can thrive and breed in captivity.

Adult specimens can be kept in glass or fiberglass enclosures in moderate sized groups. When considering the type of cages, space, height, and the surface of the cages should be taken into account. Since these frogs tend to be easily stressed, overcrowding should be avoided. And because they are active climbers at night, the height of the cage should bear more importance than the amount of floor space. Any rough surfaces should be avoided.

At NZP, for a group of 6-10 adults, I use a 20-gallon long aquarium standing on its end, with the cage top hinged so that it can be used as a door.

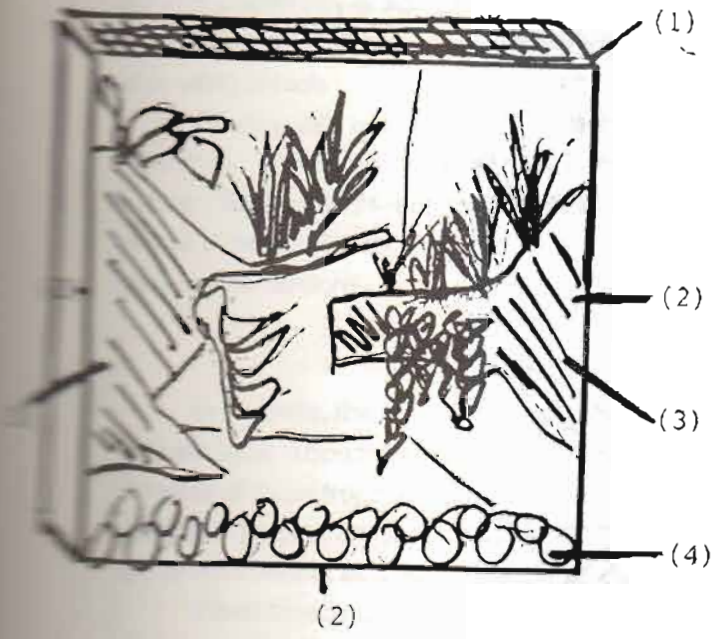
Cage furnishings can consist of any stalky, leafy plant such as *Philodendron*, bromeliads, or *Spathiphyllum*. Bamboo perches and branches are also suitable. The more plants and perches provided, the more hiding and climbing spaces are available for these frogs.

The type of substrate to use depends on the amount of energy one wishes to spend. Gravel or some other attractive rocks would be appropriate for any exhibit. For a holding cage, however, the substrate can be astroturf, mulch, sphagnum moss, or just a bare floor. Though lacking in aesthetic quality, astroturf is highly recommended for cages off exhibit because it is the easiest to clean and disinfect, and it retains moisture.

When setting up a cage for these frogs, it is important to keep in mind that though these frogs are found in the tropics, they live in a fairly dry environment except during the rainy season. Therefore, a bowl of water on the floor of the cage is sufficient. In conjunction with the astroturf as substrate, the cage will be moist enough.

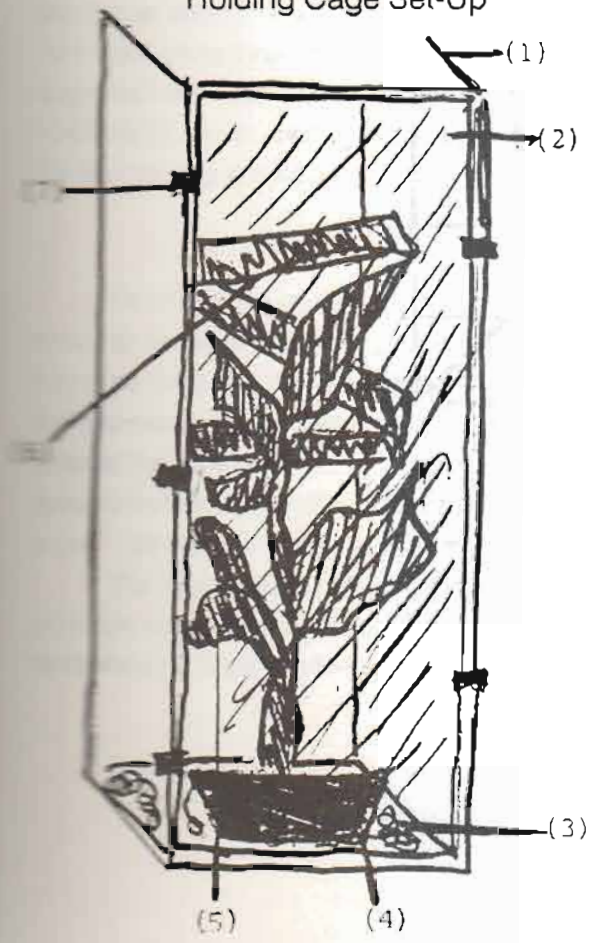
Lights used for these frogs can be strips of fluorescent Chroma 50s and black lights suspended over the top of these cages at a 12/12 L:D cycle. If the cage is big enough, provide a hot spot by suspending a 75 or 150 watt plant grow light over one corner of the cage. Air temperature should range from 75° to 90°F.

Exhibit Cage Set-Up



(1) 1/4 inch wire mesh top
(2) fiberglass walls, floor, and back door
(3) wood stumps on walls
(4) 5-8 cm gravel

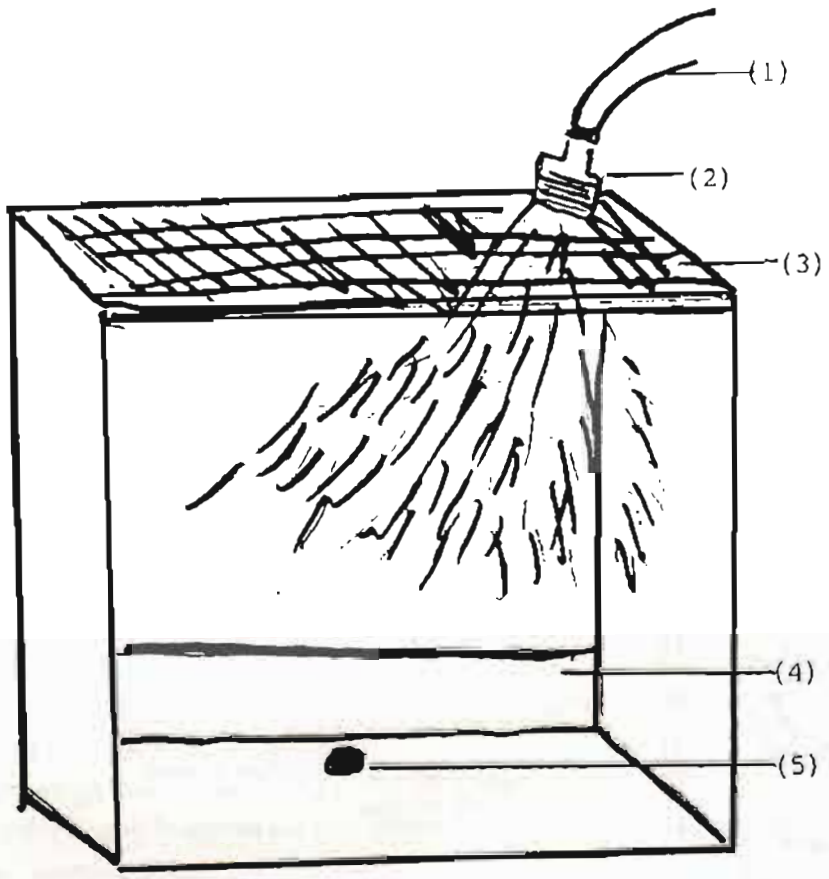
Holding Cage Set-Up



(1) Glass tank, 20-gallon long
(2) Screen mesh door
(3) Astroturf
(4) Glass bowl of water
(5) Plant cuttings
(6) Bamboo perches on walls
(7) Hinges

Showering Set-Up

(1) Hose suspended
(2) Shower head
(3) Screen top
(4) Overflow at back door
(5) Drain plug on floor



The daily maintenance of these frogs, as for all species of amphibians, should be conducted on a routine schedule. Thus, the walls, the floor, leaves, and astroturf should be cleaned in a consistent manner, usually every other day or so. On a weekly routine, individual specimens should be examined and the astroturf, if used, should be disinfected with a mild solution of bleach and water. Be sure that the item is rinsed and dried after using bleach.

Feeding can be done once or twice a week, depending on the plumpness of the frogs and their breeding activity. During the dry cycle, I only feed them once a week, while during the wet season I feed them twice a week. Adult and 1/4-inch crickets dusted with Pervinol powder are placed in a bowl and left in the cage for a couple of days. The crickets tend to stay in the bowl and retain more of the Pervinol dust on them than if they were strewn randomly in the cage.

REPRODUCTION

In captivity, the dry and wet cycle can be easily manipulated to encourage breeding activity in *Agalychnis callidryas*. Though changes inthe light cycle do not appear to have any significant effect on the breeding behavior of these frogs, a pronounced dry and wet cycle does. The essential factor in getting these and other species of frogs to breed is to provide a noticeable wet and dry cycle. Therefore, the frogs should be kept in a dry environment at all times, except when breeding is desired.

When breeding is to be induced, a hose with a shower attachment should be suspended over the top of the cage. (If the top of the cage is solid, use the side of the cage.) Allow water to overflow as the water is being lowered into the cage. Usually a few hours of showering per day is recommended for breeding conditioning.

Depending on the health of the individuals and the frequency of the breeding cycles, eggs should be discovered shortly. For example, at NZP, the first shower given to a group of frogs kept in a dry environment for several years threw the frogs into a breeding frenzy and eggs were found the following morning. But for frogs that have been showered repeatedly with only a few months of dry periods inbetween, reproduction was less likely to occur. One note to keep in mind is that if a few days of showering produces no breeding activity, it is best to stop and let the frogs dry out for a longer period.

EGG INCUBATION

Once the egg clusters are found, it is best to remove them to a separate tank so that careful observations can be made and the eggs are not disturbed. Usually, I remove the leaves with the eggs and tape the former to the sides of an aquarium. About 3-4 inches of dechlorinated water is left on the bottom of the tank and a sheet of glass or heavy plastic is placed over the entire top. If the eggs are found to be attached to the sides of the walls or tanks, I use a razor blade to remove the clutch and try to re-attach the clutch to a leaf. Sometimes the clutch will not adhere to the new leaf; in that case I float the leaf and clutch on a sponge in the water. I do not mist the eggs and I avoid getting water on the clutch.

The temperature inside the incubator is an important factor in hatching success. In sealed incubators with high humidity, temperatures of 82°F or above are too high. Embryos may die after day 5 at these high temperatures. Eggs incubated at 74° to 78° hatch out successfully.

Table 1.
Breeding Results for *Agalychnis callidryas* at the National Zoological Park

DATE	NO. OF EGGS IN CLUTCH*
8/04/84	3.3 adults to breeding set-up for showers
8/05/84	60-70
8/06/84	12
8/11/84	~ 60
8/13/84	20-30
	20-30
	30-40
8/31/84	30-40
	20-30
	20-30
	20-30
*The majority of eggs in 1984 clutches were fertile, but none hatched out. Temperature range in artificial incubator was 82° to 91°F. All embryos died in egg capsule on day 5.	
7/16/85	2.2 adults to showers
7/22/85	~ 10
6/04/86	11.9 adults to showers
6/11/86	30-40
	30-40
	30-40
	10-20
7/13/86	30-40
	30-40
11/13/86	10-20
	10-20
11/24/86	30-40
	~ 10
4/03/87	11.9 adults to showers
4/04/87	20-30 infertile
	20-30 infertile
	20-30
	20-30 Infertile
4/05/87	30-40
	10-20
	10-20
	30-40 infertile
6/25/87	30-40
	30-40
c6/26/87	10-15 2nd generation, 1st breeding

TADPOLE CARE AND DEVELOPMENT

Once the eggs hatch, the tadpoles take a couple of days to absorb the yolk sac before they will feed. After several days, they tend to suspend at a 45° angle, moving the tips of their tails. When they are disturbed, they swim frantically.

The tadpoles are fed a variety of fish food products in powdered form. The food items are all commercial products easily obtained directly from wholesalers. Equal parts of the following are fed to the tadpoles daily:

Tetra-min Staple Flakes
Tetra-min Conditioning Flakes
Krill Flakes
Freeze-dried Brine Shrimp
Freeze-dried Tubifex Worms
Freeze-dried Plankton
Freeze-dried Bloodworms

These food items are ground up in a coffee mill and Pervinol powder is added at approximately 1/5 of the total volume.

Of the many clutches of tadpoles that *Agalychnis callidryas* reared at NZP, those that were housed in tanks with water changes exhibited low frequency of deformities and mortality. These tadpoles grew at a fast pace and metamorphosed with no mortality. However, several clutches reared in other filtration devices experienced slow growth rates and massive deformities and mortality. Charcoal, sponge magnet, and undergravel filters were tested. These filters appeared to have a detrimental effect on growth of the tadpoles. Since these tadpoles are filter feeders, filtration devices may be removing the small particles of nutrients essential to their growth. Further studies are needed, however.

The period from hatching to metamorphosis ranged from 32 to 53 days. Those that metamorphosed in the shorter periods were reared in tanks with daily water changes, while those that took longer to metamorphose were reared in tanks with filtration devices. The shorter the period between hatching and metamorphosis, the lower the deformity and mortality frequency.

FROGLET CARE AND DEVELOPMENT

The froglets appear plump for a few day after metamorphosis. They often change from a bluish-gray to a maroon-brown color. It is unknown why they exhibit these strange colors, and then change colors, in their first two weeks. Their eyes are round and are colored yellow, as in the tadpoles. After a period of two weeks, the eyes turn red and the dorsal color turns green. After the initial two weeks, the behavior as well as the color of the froglets greatly resemble that of the adults. Sexual dimorphism can be detected at 5-6 months of age and calling can be heard at seven months.

Department of Herpetology
National Zoological Park
Washington, DC

AN UPDATE ON SPINDLY LEG SYNDROME IN FROGS

James H. Marlett

This paper is an informal update of our continuing battle to eliminate spindly leg syndrome in anuran reproduction. Spindly leg syndrome (SLS) is a condition in metamorphosing frogs characterized by thin forelimbs which are either weakened or completely useless. This condition has appeared in many collections.

We have become more and more convinced that at Sedgwick County Zoo & Botanical Garden, SLS is related to diet in the parent frogs (Marlett, Eichler, and Hagemeister 1987). During the 1987 breeding season we made a concerted effort to maximize vitamin and mineral supplementation in our dendrobatid frogs. Their ration of pinhead crickets was dusted daily with Theralin powdered pet vitamins and with Osteoform, a calcium, vitamin A, and vitamin D supplement. Tadpoles were fed a free-choice diet of a blended mixture of 48 g Purina Trout Chow, 48 g Tetramin Staple Diet flake fish food, 1 g Theralin, and 2 g Osteoform. The result was that no normal dendrobatid frogs were produced in 1987. Very few eggs actually hatched and any tadpoles surviving long enough to have front legs exhibited SLS.

During that time we also saw an increase in deaths among apparently healthy young and adult frogs, particularly *Dendrobates tinctorius*, in which death was preceded by a generalized loss of coordination followed by paralysis of the hind limbs. Ultraviolet light has been reported to prevent this condition (J. Murphy, pers. comm.). Ultraviolet light had no noticeable effect in our case.

While there was no evidence that this phenomenon was related to our breeding failure, both problems led us to consider the possibility of over-supplementation, particularly of vitamins A and D, which are known to be toxic, and calcium, which is known to interfere with the absorption of some nutrients (Lewis, undated). In November of 1987, Osteoform supplementation was reduced to twice a week and multivitamin supplementation was reduced to the other five days. We have experienced no more deaths related to paralysis of the hind limbs since that time.

In January of 1988, after the reduction of Osteoform to twice a week, the firm of Allen and Baer Associates, Inc., analyzed diets at Sedgwick County Zoo. It was their opinion that calcium, phosphorus, vitamin D, and vitamin A were being supplied to the anurans in the correct ratios, but they were still being fed in excessive amounts. However, we have made no further supplement reductions at this time.

We were also concerned that some micronutrients may not have been available to the frogs, either because they were not present in a useable form or they were not being absorbed. It was decided that when our supply of Theralin was exhausted, we would replace it with Nekton-Rep, a vitamin and mineral supplement designed for reptiles and amphibians. In addition to vitamins and minerals, Nekton-Rep contains the amino acids L-lysine and L-methionine. Nekton-Rep-Color, which we received through a shipping error, contains the carotene pigments canthaxanthin and apocarotenal. We realized that it was not good science to make more than one change at a time in the entire group of frogs under study, but we also felt that these changes were indicated for our overall husbandry program.

Thus, the 1988 breeding season for *Dendrobates auratus* began with Osteoform supplementation reduced from daily to twice a week and multivitamin supplementation reduced from daily to five times a week. The multivitamin had been changed from Theralin to Nekton-Rep-Color. All other facets of culture remained the same as those used in 1987 and the same frogs were used for breeding. Tadpoles were fed the same diet as in 1987 except that Nekton-Rep-Color was used in place of Theralin. All other conditions, including water quality, remained the same.

In 1988, of the eleven froglets metamorphosed so far, none has exhibited spindly leg syndrome. This contrasts sharply with 1987, in which no normal *D. auratus* were produced.

With the help of St. Francis Regional Medical Center in Wichita, Kansas, we have attempted to perform histological examinations on some of the many preserved SLS frogs which have been generated both in our collection and in other collections. Unfortunately, amphibian tissues apparently autolyse rapidly and none of the specimens had been properly preserved for histopathic examination. This phase of our investigation has not been very revealing except for two differences noted between SLS frogs and the only normal, wild caught juvenile specimen we had at our disposal. The first difference is that the compact tissue of the forelimb bones appears to be much thicker in SLS frogs than in our normal specimen. Calcium staining is being performed to determine if there is a difference in calcium deposition. The second is that bone marrow observed in the forelimbs of SLS specimens was formed of fatty tissue typical of mature bones.

The marrow observed in the normal frog was active hematopoeitic tissue typical of young bones. While we do not believe it to be the case, it is possible for both of these characteristics to be artifacts of sectioning different locations on the bone. Clearly, more histological data are called for.

LITERATURE CITED

Lewis, L. D. Undated. Harmful effects of common dog feeding practices. Mark Morris Associates, Topeka, Kansas. 5 pp.

Marlett, J. H., V. B. Eichler, and M. Fellows-Hagemeister. 1987. Spindly leg syndrome in frogs: present knowledge and future directions to elucidate its causes. 11th International Herpetological Symposium on Captive Propagation and Husbandry. Zoological Consortium, Inc. Thurmont, Maryland. pp. 145-150.

Sedgwick County Zoo & Botanical Garden
5555 Zoo Boulevard
Wichita, KS 67212

BREEDING AND YEAR-ROUND MAINTENANCE OF THE EUROPEAN FIRE SALAMANDER (Salamandra salamandra) IN AN OUTDOOR ENCLOSURE

John C. Brunner

INTRODUCTION

Nine adult (5.4) fire salamanders (*Salamandra salamandra*) were acquired from Mr. John Pickett in 1978. They were said to have originated in the northern Pyrenees Mountains of Spain.

By the description and the distribution maps in Steward (1969) I determined that these specimens were probably of the subspecies *terrestris*. *Terrestris* is a medium-sized race, slightly shorter and more slender than the subspecies salamandra. My males averaged 5 inches total length. The females averaged 5-1/4 inches and were slightly more stocky in the body than the males. Sex can readily be determined since the lips of the cloaca are more protuberant in the male. In *terrestris* the yellow markings on the back form two parallel stripes. Some of my specimens had broken stripes and some had solid stripes, both running together partway down the tails.

According to Steward (1969) the main requirements of these salamanders are shade and moisture. They also prefer moderately elevated terrain with leaf litter under deciduous forests. Maruska (1982) classifies *Salamandra* as a moderate to cool temperature preferring salamander (45° - 72°F).

HOUSING

I kept these salamanders in an outdoor pen in Montrose, Pennsylvania for several years. Our elevation in northern Pennsylvania is 1700 feet. It is hilly terrain, largely covered with deciduous hardwood forest. This environment apparently approximated their natural habitat. They were kept outdoors with no supplemental winter heat and were brought in three or four times yearly (in the summer) to check their condition and offer supplemental (controlled) feedings when necessary.

The pen was located in the partial shade of a large sugar maple tree. In addition, the pen was surrounded by a thick stand of Japanese Knotweed (*Polygonum cuspidatum*), which formed a dense shade canopy over the pen from 1 June until mid-October.

The pen housing the adults consisted of 1/2" plywood forming a 4' x 8' rectangular vertical wall; it was sunk 6 inches into the ground, with 10 inches remaining above the ground. The top edge had a 2-inch screen lip stapled to protrude horizontally into the pen area to prevent salamanders from climbing out. When not attending the pen, a cover was put on top of the vertical walls. The cover was constructed of 2 x 3s in a framework covered with 1/4-inch hardware cloth. The pen was, therefore, open to whatever wind, rain, snow, and sun that was available.

At one end of the pen, a hibernaculum was constructed. This consisted of a 12" diameter hole dug 12" deep (at which point bedrock was encountered). This hole was filled with stones in such a way as to allow passages and cavities that would give salamanders access to the bottom of the hole. In severe winters (to -2°F) none of the *terrestris* were lost to frost. In winter, several inches of leaves would be spread over the stone hibernating pit to add insulation and reduce the depth of frost penetration. A number of times in early spring when the ground was still frozen at the surface, rain would cause standing water to accumulate to a depth of a few inches in the pen and remain water-covered for several days. Somehow the salamanders survived these events and emerged a few weeks later. The soil in the pen area was rich humus and well drained. Once frost had left the ground, water would drain away from the pen within a day or two.

At the other end of the pen (inside), a small "pond" was constructed by lining a small (12" diameter; 6" deep) depression with mulch plastic and filling it with water. In this "pond" the adults would deposit their offspring. A number of hiding places were provided, consisting of flat stones, boards, and small logs.

A few inches of leaf litter were provided year-round to encourage the growth of naturally occurring small insects and earthworms as a source of food for the salamanders. On the inside vertical walls of the 4' x 8' pen was stapled a 6" band of aluminum flashing which served to inhibit the escape of crickets from the pen. Both crickets and earthworms were offered in the pen at irregular intervals, perhaps every other week during the warmer months. In addition, as mentioned, three or four times per summer the salamanders were brought indoors briefly (24 hours) to an aquarium where they could be observed and fed a meal of earthworms, mealworms, and/or crickets. In spite of a rather severe (cold) climate in Montrose, *Salamandra* were observed active and feeding as late as 3 November and as early as 19 March.

As a testament to the hardiness of this species and success of the hibernaculum refuge constructed in their pen, two active males were spotted on the surface on 19 March 1983 during a 45°F rain; by 24 March nighttime temperature was 12°F. No salamanders were lost due to freezing, in spite of having removed most of the leaf litter cover from over the hibernating stone pit.

BREEDING

The mating behavior as described by Steward (1969) was only observed once, in October 1984. Mating is said to occur at all times of the warmer part of the year, but usually occurs in spring or early summer. Mating occurs on land.

My first recorded birth was of 25 live young born in May 1982. Prior to the birth, a particularly plump female was noted in the outdoor pen and she was brought indoors to observe her more closely. She was placed in a 10-gallon aquarium with shallow water (1" deep) at one end and flat stones at the other. A few days after being brought inside, the female gave birth to 25 young. The young were about one-inch long with gills and four well-developed legs. The tails were flattened vertically and the color was brownish, speckled with small black dots.

After the birth and a feeding, the female was returned to the outdoor pen, while an attempt was made to rear the young in the 10-gallon aquarium. In spite of frequent trips to ponds and swamps with bucket and net, I found it difficult to provide an adequate diet to the larvae. They would feed on a variety of live aquatic animals such as *Daphnia*, tubifex, and mosquito larvae. Like Wisniewski and Paull (1986) in their early broods, I had poor growth rate and high mortality. Approximately one half of the brood made it to metamorphosis, but then the young became even more difficult to feed. Eventually, two survived from the brood, mainly because they were returned to the adult's outdoor pen where they were "on their own" to find small food items. Three years later (in May 1985) these two survivors were three inches long.

No young were produced in 1983, probably because of a deficiency of food available to the adults the previous summer.

In 1984 at least two of the four females in the outdoor pen showed signs (plumpness) of being gravid. In early June 1984, 41 live larvae were produced (probably two litters). The larvae were deposited in the small "pond" in the adult pen. In anticipation of their birth, a larger pond had been excavated a few yards from the parents' pen. This rearing pond was eight feet in diameter and six-to-ten inches deep. It was built in the same shady environment as the adults' pen, within a thick stand of Japanese Knotweed. The pond was well stocked with small aquatic animals and insect larvae prior to the introduction of the *Salamandra* larvae. The pond was surrounded and enclosed by a vertical plywood wall approximately 12 inches high and sunken into the ground a few inches. A horizontal lip of screening (two inches each side)

was stapled along the top of the plywood wall. No attempt to cover this rearing area was made due to the large area (approximately 200 sq. ft.) enclosed. It was hoped that the plywood wall would keep potential predators out of the rearing pond and, of course, keep the fire salamanders in the area.

Of the 41 larvae born in 1984 and put in this rearing pond, an estimated 30 or more successfully metamorphosed. They began metamorphosing on 24 July, slightly under two months since birth. As the summer progressed it seemed that fewer and fewer of the metamorphosed young could be found in the leaf litter and under logs and stones in the rearing pen. As a precaution, seven young were removed to the covered pen of the parents. Here they did well, growing two inches within one year (doubling of length since metamorphosis).

The remainder of the young left in the rearing pen gradually disappeared. Their disappearance was almost certainly the result of predation by green frogs (*Rana clamitans*) and, though less likely, garter snakes (*Thamnophis sirtalis*). Garter snakes had a difficult time crossing the plywood fence around the rearing pond. Often they would be found outside the fence apparently trying to get in. Only on two occasions were garter snakes found *inside* the fence. Once a freshly dead garter snake was found inside the pen. It had no visible marks on its body. Possibly it died from trying to eat a poisonous *Salamandra*, though none was found in the stomach. On one other occasion a live garter snake was found inside the rearing pen. It was forced to regurgitate a freshly swallowed fire salamander. Both the snake and the salamander appeared unharmed.

The green frogs were probably the main reason most of the young salamanders disappeared from the rearing pen. The frogs had no trouble entering the enclosed rearing pond (over the one-foot-high fence). When I became suspicious of the frogs (several were in the pond area), I dissected one of the larger frogs and found the partially digested remains of a small salamander. Plans were made to raise the height of the rearing-pond fence.

The final breeding of my fire salamanders was in May 1985. Over a period of several days, beginning on 17 May, one-or-more of the four adult females produced 44 live and seven dead young (larvae) in the small pond in the adult pen. The 44 live were transferred to the larger rearing pond. All went well for about one month when most of the larvae began to develop a bloated condition, almost as though a gas bubble were formed under the skin around the abdomen. Most of these died. It is not certain, but heavy rains in early summer may have raised pond acidity, having some effect on this swollen skin condition. Rainwater pH measured in Montrose, Pennsylvania in May 1988 measured 5.7 and 4.8 on two different occasions. Rearing pond water after heavy rains measured pH 6.3. After four days of no rain the same pond measured 6.5. The survivors which metamorphosed in 1985 once again disappeared. Attempts were made to reduce the predatory green frog population, but they were not completely successful.

CLIMATOLOGICAL FACTORS

Although many problems were met in the rearing of young fire salamanders, the survival and breeding of the adults for a number of years in a seminatural outdoor environment might be explained by some climatological comparison between Montrose, Pennsylvania and the salamander's natural range in Europe.

Seasonal day lengths (photoperiods) are nearly identical for Montrose and Northern Spain, since both are approximately at 42° North latitude. Day length in June is about 15 hours and in December about 9-1/2 hours.

Since my *terrestris* were said to be collected in Northern Spain, weather data were found (*World Almanac - 1988*) for Madrid (to the southwest) and for Geneva (to the northeast).

Table 1. Climatological comparison of Madrid, Spain; Geneva, Switzerland; and Montrose, PA

	Madrid		Geneva		Montrose	
Elevation	2,188 ft.		1,329 ft.		1,700 ft.	
Average Yearly Precipitation	16.5 in.		33.9 in.		41.4 in.	
	Max.	Min.	Max.	Min.	Max.	Min.
Normal January Temperature (°F)	47	33	39	29	30	12
Normal July Temperature (°F)	87	62	77	58	83	60
Recorded Extremes (°F)	102	14	101	-1	100	-29

From the data in Table 1, it can be seen that the climate of the native range of *Salamandra salamandra* is similar to that of Montrose, Pennsylvania ... and to a large portion of northeastern United States. The main differences in climate are the greater precipitation in Montrose and colder normal and extreme temperatures of winter. This would be expected due to the prevailing easterly winds in the northern hemisphere, bringing a more severe continental climate to central and eastern United States as opposed to the more moderate, coastal climate of Western Europe.

CONCLUSION

A year-round outdoor enclosure is a practical and low maintenance method of keeping and breeding European fire salamanders. Success would depend on having a somewhat similar climate to the native range of this species. Shade, moisture, and a retreat from winter frost and summer heat are of greatest importance. Rearing young could be successful if problems of water quality and predation are monitored and kept under control.

LITERATURE CITED

Hoffman, M.S. 1988. The World Almanac and Book of Facts. Newspaper Enterprise Assoc., Inc. Ballantine Books

Maruska, J. 1982. The reproduction and husbandry of salamanders in captivity. AAZPA 1982 Regional Proceedings.

Steward, J. W. 1969. The Tailed Amphibians of Europe. Taplinger Pub. Co. N.Y.

Wisniewski, P. J. and L. M. Paull. 1986. Breeding and rearing of the European fire salamander. Int. Zoo Yb. 24/25:223-226.

RD 2, Box 158
Montrose, PA 18801

CAPTIVE MANAGEMENT AND BREEDING OF Heosemys grandis

Allen Foust

DESCRIPTION

Heosemys grandis, the Asian giant land turtle, is a large emydid from Thailand, Burma, Cambodia, and Malaysia (Pritchard 1979). These are mountainous countries with peaks to 5.8 km which roughly span the same latitudes occupied by South America.

The turtles grow to a carapace length of 43.2 cm and mass of about 6.8 kg. The carapace is brown and the plastron is yellowish with a fan-like pattern of dark brown lines on each scute which tend to fade with age. The toes are webbed and the animals display a tendency to be aquatic, although they are comfortable on land.

Our adults were acquired as juveniles in 1975.

HOUSING

From mid-May to mid-October the adults are kept outdoors in a public display. The area is approximately 56 m^2 with a 3 m diameter pool, 45.7 cm deep. The enclosure contains plantings of sedge, cattail, water hyacinth, and various flowering plants and weeds. Both the adults and the young keep to the water. If the female is seen on land too often, she is probably being harassed by the male and she is moved.

During the winter, housing becomes a little more troublesome because of the tortoises' size. The male is kept in an enclosure on the floor. Even though the room temperature is maintained at 29.4°C, the floor temperature is much cooler, requiring that two 250-watt heat lamps be provided. Straw bedding is used as a substrate and is deep enough for the turtles to burrow under. A water dish large enough to accommodate the male is provided. He spends most of his time in it or buried under straw in a cooler corner. In past years, the female preferred to climb out of the enclosure and sleep under a refrigerator in the kitchen. She would appear every few weeks when thirsty or hungry, usually when the floor was being washed. Now she is confined to a climb-proof pen or to a box of straw as insurance against her walking out an open door.

FEEDING

During summer, food is thrown into the pool and around the enclosure. Diced fruits and vegetables and a commercial meat mixture which we fed to our birds of prey is distributed on land and fish and meat are thrown into the water. I do not have to wait to see if they are eating as they are the biggest turtles in the pen and are quite able to fend for themselves.

They prefer meat but will eat anything. In winter I concentrate on feeding the female as much as she will eat as this is the time when she is forming eggs.

BREEDING SEASON

October is the beginning of the breeding season. The pair is put in an enclosure with a shallow pool of water. The male jumps the female immediately and copulation usually occurs within five minutes and may continue for as long as 20 minutes. After copulation the turtles are returned to their respective homes until the next day when the procedure is repeated. About 4 or 5 matings over a 2-week period seem to be sufficient. After mating they are kept apart until March or April, when the female has finished laying and the male's interest is less intense.

LAYING

The eggs can be expected from November through March (Table 1). In about 3 to 4 weeks after the last mating I move the female to our indoor public exhibit where she has access to water and land for nesting. A heat lamp is suspended about 30 cm above the area most frequently used for nesting. The exhibit is shared by about 40 water turtles which use the same nesting beach.

On one occasion the female had been sitting in shallow water for several days. As she usually spends her time under water I considered this to be unusual behavior and placed her on land by the heat lamp. Within 20 minutes she had constructed a nest and had begun to lay eggs. I have observed her nesting twice and both times it was the standard turtle nesting. The nest was dug with alternating hind feet, the eggs were laid and arranged in the nest by the hind feet, the nest was filled in and then pounded down.

On two occasions oxytocin was used to induce laying. On 1 March 1987 1 cc (20 U.S.P. units per cc.) of oxytocin was injected in a muscle of the hind leg with a tuberculin syringe. She was put on dirt in the same place that she had nested previously. Within an hour she had moved down to a ledge in the water and laid 9 eggs, all of which were recovered.

In the 1987-88 breeding season the female was not moved to the warm public display but allowed to form her eggs in a colder pen. By January 1988 she aborted an egg. Oxytocin in the usual dosage was administered in February and eggs were dropped on 8, 9, and 11 February. The female was put into the greenhouse for the remainder of the winter where a last egg was found in a nest, in March. None of the 9 eggs laid in 1988 was viable (Table 1).

Table 1.
Egg Production in *Heosemys grandis*.

Laying Date	Broken (b)/ Decomposed (d)	Infertile	Dead in Egg Full-Term	Hatched	Total Eggs Laid	Incubation Time (Days)
01/02/81	6 d				6	
02/03/82	9 d				9	
11/09/85		2		5	7	98
12/27/85		5		2	7	125-133
01/19/87		1	2	4	7	105-108
02/08/87 03/01/87	1 b		5	4	10	108-113
01/18/88 02/08/88 02/09/88 02/11/88 03/10/88	1 b	 2 3 2 1			9	

EGG MEASUREMENTS

The eggs are hard shelled, approximately 5 x 3.5 cm, and have masses from 31 to 46 g (Table 2). Variation in egg mass occurs from year to year. This is probably due to the amount of food eaten and the temperature at which the turtle was kept during the formative period.

TABLE 2.

Masses of *Heosemys grandis* eggs from 1985, 1987, and 1988 clutches.

Date	Masses of Individual Eggs (g)	Average Mass
11/09/85	46, 45, 44, 43, 43, 38	43.16
12/27/85	39.5, 39.4, 39, 38.5, 36.6, 36.5, 34.5	37.71
01/19/87	41.3, 41, 40.5, 39.8, 36, 35,3, 34.3	38.31
02/08/87	31	36.18
03/01/87	40, 38.2, 38.1, 37.7, 37.1, 36.3, 35.2, 35.1, 33.1	
02/88 - 02/11/88	35.8, 35.6, 35, 34.3, 33.6, 32.8, 28.8	33.67
03/10/88	33.5	

METHOD OF INCUBATION

After laying, the eggs are collected, weighed, and marked with the date. They are then placed in a plastic shoebox on a mixture of vermiculite and water in a 1:0.8 ratio (by mass). A lid is loosely affixed and the shoebox is put in an incubator with a circulating fan and maintained at 27.7°C. At this temperature, the eggs hatch in 98 to 113 days (Table 1).

DEVELOPMENT

Candling reveals the first signs of life at about one month. A small pink ring near the top of the egg is observed to enlarge daily until it encompasses the entire egg. During incubation the egg gets darker and develops air pockets. By the end of the third month I start looking for shadows of heads or toes so that I can determine which is the head end. Sometimes a head retracting or a foot trying to push away the light can be revealing. I mark an "X" over an air pocket at the head end and give it a quick whack with a heavy knife handle. After picking away pieces of shattered shell, the head and front feet are exposed. Any excess fluid is daubed away to make breathing easier.

Occasionally the inner membrane is still intact. If it is discolored it can safely be torn with a pair of tweezers; otherwise it should be put in a container between damp paper towels, covered loosely and returned to the incubator. The opened eggs are treated the same way. They are checked occasionally to be sure that they are not having any problems, such as hatchlings rolling unhatched eggs around, eggshells cutting into or constricting a hatchling or that the paper toweling is not drying out. Hatching is usually complete within 10 days of opening.

CARE OF HATCHLINGS

The hatchlings continue to be maintained as above until the yolk on their bellies is absorbed. After leaving the egg the babies expand and flatten.

Because flattened turtles have a difficult time righting themselves if they fall on their backs, I prefer to keep them on land. A plastic dish pan with a few inches of dampened sphagnum moss makes a good nurs-

The babies eat on land or in water and will consume any animal or vegetable matter. They will even eat from one hand while being held by the other.

GROWTH

Hatchlings have masses ranging from 25 to 40 g. Growth is rapid at temperatures above 26.6°C and by the time they are two years old they range from 934 to 1489 g.

The adult male and female have masses of 7711 and 4082 g, respectively. During their second summer and winter they were kept outdoors and moved to cooler quarters in winter and, as a result, their growth slowed dramatically.

I believe that they could be grown to sexual maturity in 6 years.

COMMENTS

I consider *Heosemys grandis* to be an easy species to keep and breed in captivity. They have also been bred at the Columbus Zoo in Ohio and the Kansas City Zoo in Missouri.

As of 1 January 1987 there were 14.14.35 in 19 zoo and private collections (Slavens 1987).

LITERATURE CITED

Pritchard, P. C. H. 1979. Encyclopedia of Turtles.

Slavens, F. L. 1987. Inventory of Live Reptiles and Amphibians in Captivity.

Turtle Back Zoo
560 Northfield Ave.
West Orange, NJ 07052

POPULATION ECOLOGY OF THE WOOD TURTLE (Clemmys insculpta)

Steven D. Garber

Wood turtles (*Clemmys insculpta*), along with both of their eastern North American congeners, spotted turtles (*Clemmys guttata*) and bog turtles (*Clemmys muhlenbergi*) are declining in numbers throughout their range. The bog turtle is widely recognized as the rarest turtle in North and the spotted turtle is now recognized as rare, threatened, or endangered in many areas. The wood turtle has been overlooked as another species of special concern. Life history data are required if this species is to receive attention and then protection. Because wood turtle populations are thought to be declining in numbers, this study was an attempt to determine whether the populations are stable, self-sustaining, and require any management.

Significant wood turtle habitats need to be adequately protected. For this to occur, we need to learn about the ecology of these organisms. Here, I will present data from two practically contiguous populations that are separated by a man-made reservoir. Population sizes and numbers of turtles per unit area were determined and movements and behavior (particularly overwintering) were noted.

METHODS

Two populations were observed over a 13-year period from 1977 to 1989. During that time 126 wood turtles were part of a mark and recapture study. Each turtle was notched along the marginal scutes; time and location were recorded, as were sex, length, mass, and over 40 other variables. The results reported here come from over 1,000 total captures.

The study sites are located along the Sargent River, which has been artificially divided by Lake Chamberlain, a 1.6 km (1 mi) long reservoir separating the two turtle populations. The reservoir was constructed during the period lasting from 1890-1892, and then the dam was raised 12.2 m (40 ft) in 1958-1959. Both turtle populations were geographically and genetically isolated from one another 99 years ago. Before then, they were probably part of one larger population. There are minor physiological and behavioral differences in the turtles from each population, which are elaborated upon below.

Animals from the two populations never come in contact with each other. All the turtles remain relatively near the river, seldom moving more than 1,500 m from where they overwinter. Their overwintering behavior is not physiological hibernation; rather, they remain in a relatively alert mode throughout the winter. Their eyes are often open and they move about on occasion.

Considerable overwintering site fidelity occurs from winter to winter (Garber 1988). The turtles overwinter in the river and use it as a primary route when traveling to their terrestrial feeding sites, usually in wet meadows and adjacent fields. The wood turtles also pass through, and on occasion spend considerable amounts of time in, adjacent woodlands.

The habitat above the reservoir consists of a shallow stream ranging from 3 to 5 m in width and seldom more than 1 m in depth. It passes through a deciduous forest. Downstream from the reservoir there is a much greater proportion of field habitat. This is a primarily old field that is maintained by the South Central Connecticut Regional Water Authority. Above the reservoir the stream fluctuates in temperature and flow according to the weather and precipitation. Below the reservoir there is an intermittent flow from the spillway, as well as a steady flow from pipes leading from the bottom of the lake. When the water in the reservoir gets too high, the pipes are opened more and a surge cascades down the river, gouging much of the stream's bottom and banks. Such flooding often coincides with the times when the turtles are in the river. Due to the flooding, the

sites where turtles overwinter differ significantly above and below the reservoir. Downstream the turtles overwinter in deeper, more protected pools, while above the reservoir the hibernation sites tend to be smaller, shallower pools.

Supplementary unpublished field observations and data were provided by John Behler of the New York Zoological Society and David Collins of the Burnet Park Zoo, both of whom have been studying wood turtles in New York State.

Wood turtles, like many species of turtles, have distinct annuli on their scutes that last many years before wearing smooth. If the turtles are caught when still young enough to be aged, their ages were extrapolated throughout the rest of their lives, even after the annuli had worn smooth. With a long term study such as this, it is possible to establish the age-group distribution for the entire population. If turtles were first captured when already of a sufficient age that their shells were too smooth to accurately age, they were included in the age class of the old turtles with an unknown age.

RESULTS

The two populations vary phenotypically from one another. The turtles above the reservoir have more yellow streaking on their carapace and more dark spots on the anterior scales of their forelimbs. In addition, above the reservoir, the older individuals have much wider heads than those turtles living below the reservoir.

The age distribution of each of the populations shows a relatively flat distribution of numbers of turtles from ages 6 through 16. The beginning of the histograms and the ends are blank because it was not possible to locate all the hatchlings and very young turtles. It was not possible to age the very old turtles whose shells were worn smooth, though they were sexed and included.

While the survivorship curves are similar for both populations, the primary difference is that there were fewer turtles above the reservoir than below in each of the age classes. During years 6 through 16 (age 6-16), above the reservoir there were 1.5 turtles per year and below the reservoir there were 2.28 turtles per year. But the size distributions, masses, ages, and sex rations were all similar.

Because most of the hatchlings that are caught once are never recaptured, it is presumed that mortality must be high. Based on such extremely high mortality during the first several years, and assuming several females laid approximately eight eggs each with high hatching success, the left sides of the survivorship curves were extrapolated. High mortality of hatchlings and juveniles appears to be the norm for these populations.

The greatest mortality rates level off when the turtles reach the age of about six years, with a length of approximately 10.5 cm (carapace length) and a mass of 250 g. The mortality rates dropped and most then survived to maturity and beyond.

The turtles reached maturity at an average age of 14 years at a carapace length of 17 cm and mass of 600 g. The longest turtles were 19 cm and the heaviest turtle was 900 g. Upon reaching maturity, they live another 12 to 18 years. However, the ages of the oldest turtles were not known, so the right-hand side of the survivorship curve was also filled in with what seemed to be a conservative extrapolation. It could be that some of the turtles live longer than 30 or 35 years, but it is too early in the study to support such a contention with data.

Oliver (1955) recorded a wood turtle that lived 58 years in captivity, but that may be an isolated incident; under ideal conditions many animals are known to live longer in captivity than in the wild. There are also other examples of turtles that lived 50 to 70 years (Biegler 1966), but most of the data from natural

populations, rather than from captive individuals, indicate that longevities of 20 to 30 years are the norm (Gibbons 1987). If many of these turtles live much longer than 26 to 32 years, then there would have been more of the older age classes. Therefore, it would seem that nearly all the turtles die by the time they reach the age of about 35.

Above the reservoir, 37% of the captured turtles were juveniles. Below the reservoir, 44% were juveniles. Juveniles were turtles less than 14 years old that could not yet be positively sexed. Above the reservoir the sex ratio of the adults was 2.4:1 (females to males) and below the reservoir the sex ratio was 1.5:1 (females to males).

In comparison, the wood turtles collected by David Collins in Oneonta, New York had 18% juveniles and a sex ratio of 1.3:1. He may have had fewer juveniles because they are small and difficult to find. Since he was mostly catching adults, the sex ratio that coincides with the more females to males is in accord with the Connecticut findings.

DISCUSSION

Both populations of wood turtles appear to be self-regulating, each having approximately the same number of turtles staggered through each age class from 6 to 16. Each of these populations is isolated from other wood turtles in the region. Other populations in the area were smaller than those studied along the Sargent River. Informal observations over the years seemed to indicate the smaller populations were declining in numbers. Roads and vehicular traffic appear to represent a major factor in this decline. It is readily apparent that a loss of just one or two turtles a year to vehicles (or collecting) could easily be enough to drive a population to extirpation. These turtles need rivers in which to overwinter, wet meadows as well as the adjacent fields to feed in, and woods , which they occasionally travel through during the summer.

In addition to the results presented here, other investigators who shared their unpublished wood turtle data concurred that the populations they had studied were also quite small. Neither Behler (personal communication), Collins (personal communication), nor my data had numbers exceeding 100 living wood turtles. If you consider how many painted turtles (*Chrysemys picta*) or how many snapping turtles (*Chelydra serpentina*) occur in a pond, the wood turtle numbers are comparatively low.

Geneticists believe that such small, isolated populations need to exchange genes periodically. Otherwise, over many generations, they would risk developing an inbreeding depression that could have a detrimental effect on the future of those populations. Polygynous animals such as these wood turtles would suffer more rapidly from inbreeding depression than would monogamous species (Chesser 1983). If wood turtles have always existed in small populations, they may show little inbreeding depression.

To keep the rate of inbreeding low, a constant rate of exchange between the populations is necessary, since there is effectively no more genetic exchange occurring between most wood turtle populations. For these populations to survive we may have to artificially exchange turtles, or their genes, among wood turtle management units. To date, conservation strategies seldom include manipulating genetic exchange between wild populations. Rather, it is usually felt that preserving isolated refugia is sufficient; such an assumption may be invalid. If we are going to preserve the genetic diversity found in a region, we will have to manage the smaller, isolated populations more carefully. Genetic diversity for a region may actually be better maintained through many small populations with differing gene frequencies than by allowing "superior" (at least temporarily) genotypes or genes to dominate in all the populations of a region, which could happen with artificial gene exchange between sites.

Many turtle species have been shown to have temperature-dependent sex determination with regard to the incubation temperatures of the eggs (Yntema 1976; Pieau and Dorizzi 1981; Pieau 1982; Vogt and Bull 1984; and Standora and Spotila 1985). However, Bull (1985) demonstrated that there is no effect of temperature on sex determination of wood turtles, and the clutches studied all hatched out with a one-to-one ratio. It is therefore unlikely that wood turtles will compensate for a decline in their numbers by increasing the ratio of females to males.

To recruit new turtles from nearby populations may require considerable planning because Carroll and Ehrenfeld (1978) showed that displaced wood turtles attempted to home to their initial capture point. Therefore, it may be necessary to limit movement to just the eggs or hatchlings.

Head-start programs have begun with spotted turtles (Collins, personal communication). However, raising turtles rapidly by keeping them indoors for the first two years, promoting growth up to six times its normal rate, may have its problems. Head-start programs may adversely affect a small population with many closely related individuals, adding to the problems of inbreeding depression. If gravid females are taken from the population several years in a row, and the young are head-started, then, when released, if the turtles remain in the general area, the chances are considerably greater that they will breed with one another. The effect will be even more inbreeding. More siblings will mate with siblings, and siblings will mate with parents, rather than maintaining what could have been the previous system where the young males mated with the older females.

CONCLUSION

Wood turtle populations studied have been found to be small and isolated. Hatchling mortality is thought to be high. Compounded with high losses due to road kills and collecting, this has led to a dire conservation management situation. Before populations can be managed, they must be identified, and their ecology needs to be understood. It has been shown that females outnumber males in this study. Inherent genetic factors relating the species' age group distribution with the sexual system that may help preserve the population structure are discussed.

LITERATURE CITED

Biegler, R. 1966. A survey of recent longevity records for reptiles and amphibians in zoos. Int. Zoo. Yearbook. 6:487-493.

Bull, J. J. and R. C. Vogt. 1985. Non-temperature dependent sex determination in two suborders of turtles. Copeia 1985(3):784-786.

Carroll, T. E. and D. W. Ehrenfeld. 1978. Intermediate-range homing in the wood turtle, *Clemmys insculpta*. Copeia 1978(1):117-126.

Chesser, R. K. 1983. Isolation by distance: Relationship to the management of genetic resources. *In*: Genetics and Conservation (Ed: C. M. Schonewald-Cox et al.) Benjamin/Cummings Pub. Menlo Park, CA.

Garber, S. D. 1988. Migratory behavior in turtles. Plastron Papers 19(13):15-21.

Gibbons, J. W. 1987. Why do turtles live so long? BioScience 37(4):262-269.

Oliver, J. A. 1955. The natural history of North American amphibians and reptiles. Van Nostrand Reinhold. Princeton, NJ 359 pp.

Pieau, C. 1982. Modalities of the action of temperature on sexual differentiation in field-developing embryos of the European pond turtle *Emys orbicularis* (Emydidae). J. Exp. Zool. 220:353-360.

Pieau, C. and M. Dorizzi. 1981. Determination of temperature sensitive stages for sexual differentiation of the gonads in embryos of the turtle *Emys orbicularis*. J. Morphol. 170:373-382.

Standora, E. A. and J. R. Spotila. 1985. Temperature dependent sex determination in sea turtles. Copeia 1985(3):711-722.

Vogt, R. C. and J. J. Bull. 1984. Ecology of hatchling sex ratio in turtles. Ecology 65(2):582-587.

Yntema, C. L. 1976. Effects of Incubation temperatures on sexual differentiation in the turtle *Chelydra serpentina*. J. Morphol. 150:17-27.

Department of Biological Sciences
Nelson Hall
Busch Campus
P.O. Box 1059
Rutgers University
Piscataway, NJ 08855

WOOD TURTLE (Clemmys insculpta) BEHAVIOR OBSERVED IN CAPTIVITY

Steven D. Garber

MATING AND COURTSHIP BEHAVIOR

From observations in captivity and in the wild, mating behavior occurs almost exclusively in water, although I once found two adults on top of one another on land, but near the water. They may have been in that position when the stream was higher and the water subsequently abated.

Males follow females under water until mounting begins. Male clasping ensures the position will remain secure, with his claws holding the anterior and posterior parts of the female's carapace. His concave plastron fits her convex carapace. They remain together for a long period, though actual coitus occupies much less time.

FEEDING BEHAVIOR

When brought in from "hibernation," wood turtle scats indicate that water and mucus pass through their digestive tracts, but little actual food passes. Likewise, they show little inclination to take food unless kept inside for longer periods. This might be more a reflection of acclimatizing to captivity, rather than any indication of their feeding behavior, or lack thereof, during the winter. Raw meat will be taken, particularly chopped meat, but one must be careful not to give them too much as it can cause bloating. There is little inclination to take lettuce and most other greens, but they readily take fresh strawberries as well as frozen and thawed strawberries. During the summer, on many occasions, I have found wood turtles with parts of, or whole, slugs in their mouths. I have never fed them slugs in captivity.

AGONISTIC BEHAVIOR

Open-mouth gaping behavior was observed when turtles were out of water. Similar behavior has been observed with their congener, the Pacific pond turtle (*Clemmys marmorata*). This behavior has been directed toward me from the turtles in the wild that were part of a study, and therefore were seeing me each day. I have also seen wood turtles in captivity direct this behavior toward one another.

AGGREGATIVE BEHAVIOR

On land wood turtles sometimes move together as well as feed together. Hatchlings and juveniles are sometimes seen grouped before dispersing. One of the most interesting behaviors observed to date has been aggregative behavior during the winter. To determine if this might occur in captivity, I created a similar environment that could be manipulated. A bathtub with a drain and faucet to control both the amount and temperature of the water was constructed. No aggregative behavior was seen under either dry or wet (water temperatures ranged from 15-20°C) conditions. But when the tub was filled with cold water (2.5°C), the turtles were stimulated to aggregate.

AQUATIC VS. TERRESTRIAL BEHAVIOR

Hatchlings in the wild are seldom found in water. Rather, they occur primarily near the water's edge, perhaps near where they hatched out, though I have yet to determine where the turtles being studied ovipo-sit. The oldest turtles are quite heavy and can come to the surface to breath by walking to shallow water and then extending their neck. If placed in water that is too deep, in captivity, the effort of coming to the

surface to breath may be too much and can result in drowning. This happened to one of my turtles, though it did recover after having gone completely limp. Although the recovery appeared complete, this turtle has never been recaptured.

DISEASE

During winter, wood turtles remain in streams where temperatures hover just above freezing all winter. They eat nothing, or almost nothing, when brought inside and kept there for days, weeks, or months before releasing them (sometimes this was necessary when waiting for biotelemetric equipment to arrive). The turtles easily become sick and sometimes die. Most commonly, their eyelids puff up, which can result in blindness, or they contract respiratory infections. Such illnesses are very uncommon in wild populations.

Department of Biological Sciences
Nelson Labs
Rutgers University
Piscataway, NJ

CAPTIVE MANAGEMENT OF EASTERN Clemmys AT ZOO ATLANTA WITH EMPHASIS ON Clemmys muhlenbergii

Dennis W. Herman

INTRODUCTION

The eastern emydid turtles of the genus *Clemmys* are favorites among both the private breeders and zoological institutions. This popularity is most likely attributed to their relative rarity in nature and the intelligence they display in captivity. The largest member of *Clemmys* is the largely terrestrial wood turtle, *Clemmys insculpta*; the most aquatic and most handsome member of the group is the spotted turtle, *Clemmys guttata*; and the smallest is the semi-aquatic bog turtle, *Clemmys muhlenbergii*, which is intermediate in size between the other two.

Several zoos and private breeders currently maintain one or more of these species, while very few keep all three in their collections (Slavens 1985). The bog turtle appears in fewer collections due to its protected status in much of its range. Most of the bog turtles presently in zoo collections are either grandfathered animals or those kept under state permits for captive breeding purposes (Herman 1987a; Tryon 1988). In most cases, private breeders will not divulge their sources for this species, although several reptile dealers recently offered 'legal' bog turtles for sale.

Zoo Atlanta (formerly Atlanta Zoological Park) has maintained the eastern *Clemmys* periodically since 1965, but had success only with *C. muhlenbergii*. While putting together the expansive reptile collection for the new Reptile House (opened 1966), the former curator purchased groups of wood, spotted, and bog turtles. Most of these turtles died after a few years with the exception of 1.1 bog turtles purchased in 1967. Zoo Atlanta did not keep either *C. guttata* or *C. insculpta* for nearly ten years, until 1978, when 2.2 spotted turtles were purchased. Wood turtles were eventually added to the collection in 1983-84 through breeding loans. The techniques for captive management used at Zoo Atlanta are variations on those used elsewhere, with some added features.

CAPTIVE MANAGEMENT OF ADULTS

Housing

Our adult groups of *Clemmys* are housed in individual artificial bogs that closely resemble their natural habitat (see illustration in Herman and George 1986). The individual set-ups for each species are as follows:

Wood Turtle. Wood turtles (2.2) are housed in an enclosure that measures approximately 760 x 250 cm (25 x 8 ft) with a sunken 950 l (250 gal) metal stock tank for swimming, mating, and hibernating. This tank has a fresh-water source that trickles through it to simulate a stream. The turtles are able to burrow into the mud and gravel substrate to escape cold winter temperatures. The entire enclosure is landscaped with a variety of berry-producing plants and weeds for forage and cover. A perimeter fence with an overhang prevents escape from climbing, at which wood turtles are adept.

Spotted Turtle. Spotted turtles (2.2) are kept in a 180 cm diameter (6 ft) metal stock tank buried to a depth of 46 cm (18 in) for insulation. This set-up is identical to that of the bog turtle and will be discussed in that section.

Bog Turtle. Bog turtles (5.9) are housed in various sized metal and fiberglass tanks. The enclosure dimensions and the number of bog turtles per enclosure are: 1.1 (original pair) are housed in a 180 cm-diameter metal stock tank, 1.3 (North Carolina - population #1) are housed in a 200 cm-diameter (7 ft) metal stock tank, 1.4 (North Carolina - population #2) are housed in a 120 x 150 x 80 cm high (4 x 5 x 2.5 ft) fiberglass tank, 1.1 (Virginia) are housed in a 120 x 120 x 60 cm high (4 x 4 x 2 ft) fiberglass tank, and 1.0 (progeny of ZA's original pair) is housed in a 100 x 120 x 60 cm high (3.5 x 4 x 2 ft) fiberglass tank.

Each bog is constructed using the same basic design whether round or rectangular. Plumbing is achieved using 1/2-inch PVC tubing with freshwater supplied from the tap. An overflow drain is installed in each bog and the water exits into the zoo's sewer system so that a completely open system is maintained. Turn-valves at the intakes are used to regulate water flow. Only a small trickle is maintained. Since the water flows constantly, winter freezing has never been a problem. Each tank's substrate consists of a 5 cm layer of pea gravel topped by a 10-to-15 cm layer of a peat and sphagnum moss mixture. The water level is maintained 5 to 8 cm above this peat mud layer. The land area is built up with a mixture of long fibre sphagnum moss, ground peat, and sand over a pea gravel base. The bogs are landscaped with live sphagnum mosses, sedges, bog rushes, and other native bog species. Two rivulets in each of the large bogs flow from the intake through the land area into the water area. The water area covers nearly one-third of the set-up. Sheet metal overhangs on the corners of the rectangular tanks prevent escape by climbing.

Each bog is managed by selective cutting periodically through the year, especially in the summer. Trimming excess sedges from the perimeter of the tank prevents the turtles from using the vegetation as a bridge to freedom. Burning of each bog takes place during February. This practice rejuvenates the bogs and it clears them of dense dead vegetation that hinders the growth of the carnivorous plants in each bog. Each spring the fresh green of the sprouting vegetation makes for a beautiful bog.

BEHAVIORAL OBSERVATIONS

Natural behaviors have been observed for each species in the outdoor bogs. Each turtle is located during hibernation so that periodic monitoring can be done. The observed behaviors for the most part parallel those observed in nature. Seasonal temperatures have been recorded periodically since the first bog was constructed in 1979. Air, water, and substrate (hibernacula) temperatures were recorded daily in one bog between November 1982 and March 1983: Air, 4-18°C; water, 6-15°C; substrate, 4-14°C. Since winter temperatures are variable in Atlanta, the extreme fluctuations were expected.

Activity

The earliest emergence from hibernation was observed in the wood turtles; more accurately, they never truly hibernated. They were often observed basking on sunny winter days in January and February. The spotted turtles were observed active in late January, with increased activity during February and March. The bog turtles leave their respective hibernacula early (January), but surface activity has not been earlier than 16 February. By the end of March most of the *C. muhlenbergii* are actively seen basking and foraging for food. This activity is six to eight weeks earlier than observed for wild turtles in North Carolina (Herman 1986a).

Reproduction

Wood turtles have been bred by several zoos and private breeders (Slavens 1985). Zoo Atlanta obtained 2.3 wood turtles in 1983-84 through breeding loans. One female was found dead in 1986, the victim of rodent predation. Combat leading to courtship has been observed each year, with copulation observed in

October and November. Nesting has not been observed, although ground sniffing by females has been witnessed. The earliest egg deposition I have recorded was on 30 May. An average clutch of five eggs is usually laid by our wood turtles. One clutch (N=5) was weighed and measured and the following data were recorded:

Length (mm):	27.5 - 33.5, $\overline{X}$ = 34.0 mm
Width (mm):	20.2 - 22.6, $\overline{X}$ = 21.3 mm
Mass (g)	6.4 - 7.9, $\overline{X}$ = 7.3 g

The eggs were incubated in a medium of sand and peat moss at a temperature of 26.7 - 27.8°C. The only successful hatching of wood turtles at Zoo Atlanta occurred on 13 July 1987 after 54 days incubation. The neonates (N=4) were weighed and measured one day post parturition:

Carapace length (mm):	32.6 - 35.4, $\overline{X}$ = 34.0 mm
Plastron length (mm):	27.3 - 29.4, $\overline{X}$ = 28.2 mm
Mass (g):	6.4 = 7.9, $\overline{X}$ = 7.3 g

Spotted turtles have also been successfully bred in several zoos and private collections (Paull et al. 1983; Slavens 1985). In 1978 Zoo Atlanta purchased 2.3 *C. guttata*, the first since 1969. After an adjustment period in one of the outdoor bogs, copulation was observed. Nesting was observed only once, although the females annually deposit from 2 to 4 eggs. The eggs were always left in the nest for incubation under natural conditions. This was usually successful until 1985 when the outdoor set-ups were invaded by the imported red fire ant (*Solenopsis invicta*) and predation on recently pipped eggs was observed. The nests are now located as soon as possible and the eggs removed to be incubated indoors. Measurements and masses of a recent *C. guttata* clutch (N=2) are as follows:

Length (mm):	31.3 - 32.6, $\overline{X}$ = 32.0 mm
Width (mm):	16.7 - 17.3, $\overline{X}$ = 17.0 mm
Mass (g):	5.5 = 5.6, $\overline{X}$ = 5.55 g

The spotted turtle eggs were incubated in a medium of sand and peat at a temperature of 20-28.9°C. The masses and measurements of neonates hatched in 1984 (N=2) after 63 days of incubation were:

Carapace length (mm):	26.8 - 27.8, $\overline{X}$ = 27.3 mm
Plastron length (mm):	23.1 - 23.6, $\overline{X}$ = 23.3 mm
Mass (g):	4.1 - 4.4, $\overline{X}$ = 4.2 g

In 1973 bog turtles were first bred in captivity at the Bronx Zoo (Anonymous 1974). Since then they have been bred at other zoos (Tryon and Hulsey 1977; Herman 1980; Slavens 1985) and in private collections (Warner 1974; Slavens 1985). One pair (1.1) of bog turtles was purchased by the zoo in 1967 and is still reproductively active. These turtles have surpassed the longevity record reported by Bowler (1977) and are at least 25 years old. They have successfully produced eggs and offspring annually since 1974. The female has deposited a total of 56 eggs during this time. Her clutches have ranged from two to seven eggs annually ($\overline{X}$ = 3.7), and she has deposited multiple clutches in 1985 (Herman 1983; Herman 1986c). After the location of the

bog turtle nests, the eggs were removed, weighed, measured, and returned to the nest cavity for natural incubation. Incubation temperatures of one *C. muhlenbergii* nest (clutch = 4 eggs) were recorded daily between 20 May and 20 July 1986 at 0900 h, 1300 h, and 1700 h:

0900 h:	14.4 - 26.7°, $\overline{X}$ = 24.8°C
1300 h:	22.2 - 34.4°, $\overline{X}$ = 28.4°C
1700 h:	22.8 - 32.2°, $\overline{X}$ = 28.7°C

In 1985 several bog turtle nests were destroyed by an oppossum, *Didelphis marsupialis* (Herman 1986b). The following year predator-proof wire cages were constructed and placed over each nest. Unfortunately, imported red fire ants invaded the bogs and destroyed the emerging neonates as they pipped their eggs. One of these destroyed nests contained an egg with fully developed twins (Herman 1987b). Beginning with 1987, all bog turtle eggs are removed from the nest and incubated in a 19 l (5 gal) aquarium in a sand, peat, and sphagnum moss medium. Incubating temperatures fluctuate between 20-28.9°C. A clutch of five eggs laid 30 May 1988 was weighed and measured:

Length (mm):	28.5 - 31.5, $\overline{X}$ = 29.7 mm
Width (mm):	15.8 - 16.3, $\overline{X}$ = 16.2 mm
Mass (g):	4.6 - 5.0, $\overline{X}$ = 4.8 g

Masses and measurements of five neonates that hatched in 1987 were:

Carapace length (mm):	26.6 - 28.5, $\overline{X}$ = 27.6 mm
Plastron length (mm):	21.6 - 22.7, $\overline{X}$ = 22.3 mm
Mass (g):	4.6 - 4.9, $\overline{X}$ = 4.8 g

FEEDING

Feeding regimes differ slightly among the three species. Since the adults are housed outdoors year round, they forage for and feed on a variety of invertebrates and plants. The wood turtles are fed salads bi-weekly, which consist of chopped fruits, vegetables, and whole skinned mice, with Pervinal^{R} multivitamin powder (St. Aubrey/Division of 8-in-1 Pet Products, Inc., NY, NY) added. The spotted turtles and bog turtles are supplemented bi-weekly with either newborn mice, crickets dusted with bone meal, or earthworms. The bog turtles have been observed feeding on the leaves of day flowers and grass-of-Parnassus growing in the enclosures. Occasionally they are fed strawberries and watercress.

CAPTIVE MANAGEMENT OF NEONATES

Housing

Neonate *Clemmys* are now reared together in an aquarium that measures 122 x 60 x 30 cm high. This rearing set-up is very similar to the larger outdoor bogs. An undergravel filter is covered with a layer of coarse gravel. A peat mud layer is created by floating ground peat moss and sphagnum moss on the water until it becomes water soaked and sinks to the bottom. This usually takes 3-4 weeks, but is facilitated by the filtering action of the undergravel filter. One end of the set-up is landscaped and planted with live sphagnum mosses, sedges, and bog rushes. Light is provided by four 40 W Vita-Lites^{R} (Duro-Test Corp., North Bergen, NJ) placed 30 cm above the land surface. Three 150 W incandescent plant lights provide hot spots for basking.

Ultraviolet light is essential to neonate turtles and is provided by one 250 W/110 V UV lamp on a timer. Fresh water is added to the set-up as needed due to evaporation. An overflow drain is used to periodically flush the set-up.

Feeding and Growth

The neonates are fed a calcium-enriched diet two or three times weekly. This diet includes Frog or Turtle Brittle® (NASCO, Fort Atkinson, WI), chopped mice or newborn mice, minnows, pond snails, slugs, dusted crickets, and earthworms. Frog Brittle is fed to the smaller turtles and Turtle Brittle to the larger neonates or juveniles. Occasionally Purina Trout Chow (Purina Feed Co., St. Louis, MO) is fed to the juveniles because it has a higher fish oil content than the Turtle Brittle. Frog/Turtle Brittle currently makes up 40-50% of the young *Clemmys*' diet. The following is the feeding record of one neonate *C. muhlenbergii* over a 2-1/2 year period:

Food Item	No. Feedings	%
Frog Brittle	76	35
Crickets	50	23
Newborn Mice	41	19
Minnows (Fish)	20	9
Earthworms	16	8
Chopped Mice or Meat	9	4
Pond Snails	3	1
Vegetation	1	-
TOTAL	216	99%

Growth rates of neonate *Clemmys* are scant at the zoo, except for *C. muhlenbergii*. Spotted turtle and wood turtle growth rates have only been recorded over a six month period, while growth rates of *C. muhlenbergii* were recorded (six month intervals) over a 2-1/2 year period. Below are the mean growth rates of the three *Clemmys* for comparison:

C. insculpta (N=3)	*C. guttata* (N=2)	*C. muhlenbergii* (N=6)
CL: 9.1 mm	CL: 4.5 mm	CL: 6.9 mm
PL: 7.9 mm	PL: 2.9 mm	PL: 5.2 mm

All of the neonates were fed through the winter months and not hibernated. The wood turtles grew faster than either the spotted or bog turtles.

Mean growth since hatching for the above six bog turtles over a two year span was:

After One Year	After Two Years
CL: 22.1 mm	CL: 30.8 mm
PL: 18.7 mm	PL: 26.9 mm

These growth rates are more accelerated than those I have measured in wild bog turtles (Herman, pers. obs.). This is expected for captive, non-hibernating turtles. Arndt (1972) maintained two hatchlings which grew an average of 3 mm during four months. Sachsse (1974) reported a growth rate of 13.5 mm over a six month period for one hatchling while Warner (1974) recorded growth in two neonates at 4 mm each in one month. Tryon and Hulsey (1977) recorded 33 mm mean growth in two neonates over a 12 month period at

Fort Worth Zoo. Humidity is an important factor in the growth rate of *C. muhlenbergii* according to Zovicklan (1971). The rearing set-up at Zoo Atlanta provides moderate to high humidity for the growing neonates. The growth that I have recorded for captive bog turtles falls within the range reported by others.

CAPTIVE PROPAGATION/RELEASE PROGRAM

Two of the outdoor bogs house bog turtles that were collected under permits from the North Carolina Wildlife Resources Commission for a captive propagation project. Bury (1979) expressed concern over mixing native stocks for release, thus affecting genetic variability. This is a valid point in our opinion and we have taken it into account in our projects. The NC bog turtles are from two different wild populations so they are housed in separate bogs. One set-up houses 1.4 bog turtles while the other has 1.3 bog turtles. Any progeny produced from these turtles are monitored and fed for several months to one year, after which they are released back into their parents' native bogs. To date, five neonates have been returned to the wild from this program.

A head-start project was initiated in 1982 and annual permits from North Carolina allow me to collect up to five gravid bog turtles and return them to the zoo for egg and neonate data. The females are released soon after egg deposition at their points of capture. After an adjustment period, the neonates are returned to their native bogs. Six neonates have been returned to the wild in North Carolina, Tennessee, and Virginia as a result of this project.

SUMMARY

Habitat destruction is the primary cause for the decline in eastern *Clemmys* populations. Collecting for the pet trade is a major secondary threat, especially for the threatened wood turtle and the rare bog turtle. Captive breeding programs for these turtles are necessary to alleviate pressures on wild populations. The knowledge gained from studying these interesting and often misunderstood chelonians is invaluable. It is important for zoos, as well as private turtle breeders, to work closely with the various state nongame agencies and get actively involved in the conservation of the eastern *Clemmys*. Maybe this involvement will help insure that the *Clemmys* will survive into the next century.

ACKNOWLEDGMENTS

I wish to thank the staff of the Department of Herpetology for their assistance and encouragement. I am grateful to Howard Hunt for his helpful comments and advice from the project's infancy to the present.

LITERATURE CITED

Anonymous. 1974. Rare bog turtles breeding at the Bronx Zoo. Def. Wildl. News 49(1):64.

Arndt, R. G. 1972. Additional records for *Clemmys muhlenbergi* in Delaware, with notes on reproduction. Bull Md. Herpetol. Soc. 8(1):1-5.

Bowler, J. K. 1977. Longevity of reptiles and amphibians in North American collections. SSAR Misc. Pub. 6:32.

Bury, R. B. 1979. Review of the ecology and conservation of the bog turtle, *Clemmys muhlenbergi*. USDI, Fish & Wildlife Ser. Special Scientific Report - Wildlife No. 219. 9 pp.

Herman, D. W. 1980. Atlanta Zoo hatches rare bog turtles. AAZK Animal Keeper's Forum 7(9):198.

_____ 1983. Life history notes: *Clemmys muhlenbergi*. Reproduction. SSAR Herp. Review 14(4):122.

_____ 1986a. Herp-of-the-Month: *Clemmys muhlenbergii* (Schoepff) bog turtle. N. C. Herpetol. Soc. Newsletter 9(2):3-5.

_____ 1986b. Life history notes: *Clemmys muhlenbergi*. Nest predation. SSAR Herp. Review 17(1):24.

_____ 1986c. Life history notes: *Clemmys muhlenbergi*. Reproduction. SSAR Herp. Review 17(1):24.

_____ 1987a. The diminutive bog turtle: A species headed for extinction? ZOOM, Publ. Friends of Zoo Atlanta 1(1):4-5.

_____ 1987b. An incident of twinning in the bog turtle *Clemmys muhlenbergii* Schoepff. Bull. Md. Herpetol. Soc. 23(3):122-124.

Herman, D. W. and G. A. George. 1986. Research, husbandry, and propagation of the bog turtle *Clemmys muhlenbergii* (Schoepff) at the Atlanta Zoo. *In:* S. McKeown et al. (eds.) Proceedings of the 9th International Herpetological Symposium on Captive Propagation and Husbandry. Zoological Consortium, Inc. Thurmont, MD. i-vi + 265 pp.

Paull, R. C., D. N. Riemer, and A. L. Smith. 1983. Spotted turtle, *Clemmys guttata* (Schneider 1792). Turtle Trust Occas. Paper. 12:10-11.

Sachsse, W. 1974. Zum Fortflanzungsverhalten Von *Clemmys muhlenbergii* Bei Weitgehender Nachamung Der naturlichen Lebensbedingungen Im Terrarium (Testudines, Emydidae). Salamandra 10(1):1-14.

Slavens, F. L. 1985. Inventory of Live Reptiles and Amphibians in Captivity. F. and E. Slavens, Seattle, WA. 341 pp.

Tryon, B. W. 1988. The rare little bog turtle of East Tennessee. Tenn. Wildl. 11(4):6-9.

Tryon, B. W. and T. G. Hulsey. 1977. Breeding and rearing the bog turtle *Clemmys muhlenbergii* at the Fort Worth Zoo. Int'l. Zoo Yearbook. 17:125-130.

Warner, J. L. 1974. Demise of the Muhlenberg? Conn. Herpetol. Soc. Bull. 4:1-2.

Zovickian, W. H. 1971. Humidity as a growth factor in hatchlings of *Clemmys muhlenbergi.* Bull. Md. Herpetol. Soc. 7(4):93-95.

Department of Herpetology
Zoo Atlanta
800 Cherokee Ave., SE
Atlanta, GA 31315

THE RADIATED TORTOISE SPECIES SURVIVAL PLAN

William Holmstrom

The radiated tortoise (*Geochelone radiata*) is one of the most beautiful tortoises on Earth. Highly desired by professional and amateur animal collectors, this tortoise is also sought by the people of Madagascar for cultural and economic reasons (Groombridge 1982; Andrianariva, pers. comm.). Although its wild status has not been recently and thoroughly investigated, the radiated tortoise appears to be stable in the core of its range, but shrinking from the east and northwest (Groombridge 1982). The radiated tortoise has been placed on Appendix I of the Convention on International Trade in Endangered Species (CITES) and is listed as "Vulnerable" in the International Union for the Conservation of Nature (IUCN) Red Data Book (Groombridge 1982).

Perhaps the primary threat to this tortoise's survival is the economic situation of the Malagasy Republic government. Although the radiated tortoise has been officially protected in Madagascar since 1961 (Groombridge 1982), funds are lacking to implement its protection, and exploitation for food and the pet trade continues today (Andrianariva, pers. comm.).

A background of the Species Survival Plan (SSP) should be briefly outlined before describing the specifics of the radiated tortoise SSP.

The American Association of Zoological Parks and Aquariums (AAZPA) is the professional organization to which virtually all of the major zoos and many of the smaller zoos in this country belong. One of the concerns of the AAZPA is wildlife conservation on an international level. The conservation wing of the AAZPA is the Wildlife Conservation Management Committee (WCMC). To help combat the alarming and accelerating rate of global extinction of species, the WCMC conceived of the Species Survival Plan in the late 1970s and early 1980s.

The goals and philosophy of the SSP program have been outlined in detail by Meritt (1980) and Foose (1981), and Hudson (1983) presented an overview of the SSP application to reptiles. In short, the SSP attempts to maintain and reproduce in captivity critically endangered species through cooperative breeding programs. The ultimate goal is to maintain genetically viable populations in captivity and to reinforce weakened natural populations or repopulate original habitat where possible. Realistically, all species cannot be considered for SSP, and criteria had to be adopted for choosing target species.

"The three basic criteria for inclusion are:

1. A breeding nucleus of the species or subspecies must be available for captive management.
2. The species' or subspecies' continued existence in the wild must be in some degree of peril as defined by the IUCN, International Counsel for Bird Preservation, U.S. Fish and Wildlife Service, or reliable field report.
3. There must be available an organized group of captive propagation professionals with sufficient support to develop and carry the species or subspecies program to captive preservation status" (AAZPA, no date).

To date, some 40 studbooks are active, either at the regional or international level. Among these are four reptilian and one amphibian SSPs. These five are: Radiated tortoise, Chinese alligator (*Alligator sinensis*), Dumeril's ground boa (*Acrantophis dumerelli*), Aruba Island rattlesnake (*Crotalus unicolor*), and Puerto Rican crested toad (*Peltophryne lemuri*).

Other reptile and amphibian species are under consideration and a special advisory group has been formed to evaluate the crocodilian situation as a whole and recommend additional SSPs. Additionally, a process is now under way to evaluate all reptile and amphibian species for future consideration.

The radiated tortoise population in the United States fits the criteria for inclusion in the SSP perfectly. I began informally to compile data for the studbook in September of 1984. The Radiated Tortoise Species Survival Plan was instituted by petition to the SCMC by the New York Zoological Society in November, 1985. I was appointed studbook keeper at that time. By late 1985, when the SSP was formalized, there were 13 institutions and one private party participating in the program, and records on 179 tortoises were included. Tortoise locations were identified through Frank Slaven's *Inventory of Reptiles and Amphibians in Captivity. International Zoo Yearbook*, and through communication with tortoise owners. The response from the private sector was disappointing. Only two of the nine private collectors contacted responded, one negatively.

A historical perspective of the North American collection from 1973 through 1987 shows an increase of 200 animals during that period (Table 1).

Table 1.
Radiated Tortoise Population Profile, 1973 - 1987

YEAR	POPULATION	# BIRTHS	# DEATHS	% MORTALITY
1973	36	2	1	2.7
1974	42	7	0	0.0
1975	48	6	0	0.0
1976	56	8	0	0.0
1977	58	2	0	0.0
1978	64	6	3	4.7
1979	71	10	2	2.8
1980	76	7	1	1.3
1981	88	13	1	1.1
1982	138	10	1	0.7
1983	154	17	6	3.9
1984	165	17	6	3.6
1985	179	18	1	0.6
1986	217	38	7	3.2
1987	236	26	5	2.1

The year 1973 was chosen as a starting point since that is the first year that radiated tortoises were reproduced in this country. The tortoises in U.S. collections up to that point arrived from varied sources, but one group of 49 (Behler, pers. comm.) animals imported by Robert Baudy in 1969 (Baudy 1970) is still well represented (Baudy only reported importing 22 animals in his 1970 publication but provided John Behler with data on 49 individuals that he brought into the U.S. in August 1969). As many as 19 individuals may still be living. The exact number is difficult to determine since ownership of a number of these animals has proven hard to trace.

The Gladys Porter Zoo and Dr. William Zovickian were first to reproduce radiated tortoises in the U.S., in 1973. The Memphis Zoo and Aquarium produced their first hatchling in 1978. The New York Zoological Society (NYZS) collection also began reproducing that year in Gainesville, Florida, under the care of Dr. Walter Auffenberg. In 1979 the NYZS group was moved to its present location on St. Catherines Island, off the coast of Georgia, and continues to reproduce the species there. Zoo Atlanta produced its first young radiated tortoise in 1981.

In 1985 a second major source of wild stock entered this country. Fifty-seven radiated tortoises were smuggled out of Madagascar in 1982 and confiscated in Hong Kong. These were transferred to the Jersey Wildlife Preservation Trust in England by direction of the Malagasy government; 38 of the tortoises subsequently were distributed among five U.S. institutions.

The Lincoln Park Zoo in Chicago was added to the list of breeding institutions in 1988. Good reproductive success has continued at Gladys Porter Zoo and at St. Catherines Island. More than 90 young have been produced by these two institutions in the past three years alone. Eggs are being produced at Riverbanks Zoological Park in Columbia, SC, and at the Institute for Herpetological Research in Stanford, CA. Reproductive success at these two facilities is anticipated in the near future.

But reproductive success is not the only goal of SSPs. Ultimately, the goal is to preserve as much genetic material as possible in a manageable captive population, while maintaining demographic stability. This is achieved through formulation and implementation of a master plan for the species. The master plan is a long range, selective breeding schedule. Cessation of breeding over-represented stock and disposal of unneeded offspring are necessary considerations of the master plan in order to keep the population at a manageable level. Formulation of the master plan is one of the responsibilities of the Propagation Group. Each SSP has a Propagation Group elected from among its participating institutional representatives. The current Radiated Tortoise SSP Propagation Group is given in Table 2.

Table 2.
Radiated Tortoise SSP Propagation Group

William Holmstrom, NYZS, Species Coordinator/Studbook Keeper
John McLain, San Antonio Zoo
Brett Stearns, Institute for Herpetological Research
Tom Huff, Reptile Breeding Foundation
Pat Burchfield, Gladys Porter Zoo
Dennis Herman, Zoo Atlanta
Sean McKeown, Fresno Zoo
Charles Beck, Memphis Zoo
Julian Duval, Indianapolis Zoo
Ron Goellner, St. Louis Zoo
John Behler, New York Zoological Society (ex officio member)

Advisors:
Dr. William Zovickian, Sharon, CT
Dr. Joseph Flanagan, Houston Zoo
Dr. Ian Swingland, University of Kent, England
John Iadarosa, St. Catherines Island Survival Center

Three propagation group members are selected each year, serving a three-year term. Advisors are appointed by the species coordinator with the approval of the propagation group.

At this juncture the Radiated Tortoise SSP has published the first edition of the studbook and is about to begin work on a master plan for the species. Additionally, a handbook on the husbandry and captive reproduction of radiated tortoises is in preparation. John Iadarosa, John Behler, John McLain, and Pat Burchfield are preparing this publication and, when completed, we hope to make it available to anyone interested.

The studbook is the instrument through which all registered SSP animals are recorded and tracked. Data are tabulated on each page of the studbook in column form. The first column shows the studbook number of the individual. This number is assigned chronologically by the length of time in captivity and remains with the animal. The second column is the sex, if known, and the third is the tortoise's in-house number or name at its current location. Birthdate, if known, is listed and sire and dam are noted by studbook number, by "wild," or by "unknown." Animal transfers are traced in the location column along with the date of transfer and the in-house identification at that location. Death dates are recorded. The status column will contain 'S' if the animal is active in the program, 'A' if alive but inactive, 'U' if its condition is unknown, and 'D' if dead. The comment section contains any useful information, such as breeding condition, injuries or medical problems, ownership, cause of death, etc. Comments are listed on a separate printout.

Other reports available from this particular program are Listing by Location, Population Age Structure, Offspring of Sire, Offspring of Dam, Age of Parents, Institutional Listing, and Mailing Labels.

The software package I am using was created by Andrew Odum at the Houston Zoo and is called the Houston Zoological Gardens Studbook Management System. It is one of several available to the studbook keeper and is favored by those keeping the reptile studbooks. Software for genetic and demographic analysis is also available.

Currently there are 27 institutions and six individuals participating in the Radiated Tortoise SSP. The expansion of the program has been rapid, mainly due to the availability of young through the breeding programs at St. Catherines Island and Gladys Porter Zoo. A number of institutions lack space for a breeding group of adults, but have expressed interest in participating as rearing facilities. The increased participation by the private sector is primarily the result of encouragement from the United States Department of the Interior Permit Office. Applicants for a Federal Wildlife Permit are urged by the Permit Office to participate in the SSP, although participation is not mandatory. Willingness to do so, however, is taken into consideration by the office when making their decision to grant or to deny a permit and the Radiated Tortoise SSP Species Coordinator has been called upon to comment on permit applications.

We anticipate continued growth of the captive population and increasing participation by zoos and private collectors. The studbook may become international in scope at some point in the future. In the meantime, however, several projects are planned for the Radiated Tortoise SSP. A master plan must be completed so that we can move forward with the aim of preserving genetic material in the captive population. Funding must be secured for preliminary and in-depth field studies to determine population levels and ecological parameters of the species. Ways and means will be explored to promote conservation of the radiated tortoise in Madagascar. And a finished studbook will be published detailing the natural history, population status, and history of the U.S. collection. With the help of a dedicated propagation group and the continued support of the radiated tortoise owners, I feel confident that the SSP can contribute to the continued well-being of this species.

LITERATURE CITED

AAZPA. No date. Species Survival Plan of the American Association of Zoological Parks and Aquariums. A guidebook to SSP. 24 pp.

Baudy, R. 1970. In quest of *Geochelone radiata*. International Turtle and Tortoise Journal, 4:19.

Foose, T. 1981. An update on the Species Survival Plan (SSP) of the AAZPA. Proc. Ann. Conf. AAZPA, pp. 8-15.

Groombridge, B. 1982. The IUCN Amphibia-Reptilia Red Data Book, Part 1, pp. 85-88. IUCN, Gland.

Hudson, R. 1983. The Species Survival Plan (SSP) and its application to reptiles. Proc. 7th Annual Symposium on Captive Propagation and Husbandry, pp. 1-9.

Wemmer, D. 1980. A Species Survival Plan for AAZPA. Proc. Ann. Conf. AAZPA, pp. 69-75.

Department of Herpetology
New York Zoological Park
Bronx, NY 10460

CURRENT AND FUTURE DIRECTIONS IN TORTOISE PROPAGATION AT THE JACKSONVILLE ZOOLOGICAL PARK

John R. Meyer, C. Douglas Page, & Celeste W. Moore

INTRODUCTION

Although tortoises have been present in the herpetological collection at the Jacksonville Zoo for the past 20 years, the efforts directed towards their husbandry and propagation have undergone an evolutionary process. Prior to 1982, the tortoise collection was relegated to a minor status, primarily because of the inadequate facilities (Williams 1986); in addition, efforts of zoo personnel were directed towards propagation of boid snakes, particularly members of the genus *Epicrates*.

With the arrival of David Collins as Reptile Curator in 1982, new impetus was given to upgrading the tortoise collection, particularly the Aldabra tortoises. The Jacksonville Zoo had exhibited these giant tortoises since 1968, and after two years of study and manipulation, three were successfully hatched in 1984 (Collins 1984).

Following Collins' departure in 1985, Ralph Williams assumed curatorial responsibility and continued the development of the tortoise program. Particularly noteworthy were additional hatchings of Aldabra tortoises in 1985 and 1986, as well as large numbers of hatchling leopard tortoises and red-footed tortoises in 1986 (Williams 1986). When Williams departed in 1986, curatorial responsibility was assumed by one of the authors (Meyer) and the continued development of the tortoise program has been the combined effort of all three authors. The following discussion summarizes the developments of the past two years and indicates future directions of the tortoise program at the Jacksonville Zoo.

Aldabra Tortoises (*Geochelone gigantea*)

Following the success of the 1984-86 hatching seasons, the Aldabra tortoise breeding program remained the same, with the exception of the intermittent exposure periods of the two males to both of the females at two week intervals. In 1986 and 1987, the males were left with the females for three to four weeks at a time from May to October. Although attempted copulations by both males were frequent and some intromissions were observed, oviposition did not occur. Egg formation was monitored using radiography at four week intervals from October through January both years. This was supplemented by frequent observations for laying activity.

The 1988 breeding season will be initiated in early June, with a return to the successful two week introduction/separation schedule. This will be augmented by the addition of an adjacent enclosure that will triple the size of the adult Aldabra tortoise exhibit. Females will continue to be restricted to their original nesting area when laying season begins. Plans are being made to acquire an additional male and two females, but these will not be introduced to the group until after the 1988 breeding season.

The winter quarters of both sexes will remain unchanged (Collins 1984) as they have proven adequate in the past. Heat is provided in the separate male and female houses by two overhead 250 watt heat lamps. Temperatures recorded during this past winter (Fig. 1) indicate the internal body temperatures maintained by one female indoors under varying outdoor air temperatures.

Of the 15 young that hatched between 1984 and 1986, three have been retained at the Jacksonville Zoo to monitor growth and development. Of the remainder, two did not survive; one was sent to the Norfolk Zoo and nine were sent to the Life Fellowship Sanctuary. Juveniles from 1984 and 1985 were moved from indoors to our off-exhibit tortoise runs in the summer of 1986; a 1986 hatchling was moved to these quarters in early 1987. This facility has allowed their free access, except during winter, to exercise and grazing. Growth records (Fig. 2) for these three individuals, all females, indicate a rapid rate of growth; frequent measurements and evaluation indicates that this growth rate is not producing shell abnormalities.

The three juveniles have outgrown their quarters of the past two years, and in early June of 1988 they were relocated to a new exhibit with increased area, indoors and outdoors, and an earthen pool. This new facility will accommodate the growing young tortoises, as well as provide the visiting public the opportunity to watch their development.

Red-footed Tortoises (*Geochelone carbonaria*)

A group of 3.3 adults of this South American species was maintained at Jacksonville from 1977 until 1986, at which time they were relocated to Silver Springs Reptile Institute. A total of 43 young were hatched between February and July of 1986, most of which were sent to various zoos and individuals in the United States. More detailed information on breeding, hatching, and rearing of this species may be found in a paper by Williams (1986).

Leopard Tortoises (*Geochelone pardalis*)

Four adults (1.3) of this African species were maintained at various times between 1983 and 1987, when the remaining breeding pair was relocated to the Audubon Park Zoo. One individual was hatched in 1985, and 51 in 1986; more detailed information may be found in Williams' (1986) report. Various zoos and individuals throughout the country received most of the hatchlings of this species. The 1985 juvenile male and a 1986 juvenile female have been retained to monitor growth and shell development to maturity, at which time breeding of this species may be resumed. Growth of these two from hatching to the present is depicted graphically in Figure 3.

Radiated Tortoises (*Geochelone radiata*)

One adult pair of the Malagasy radiated tortoise, acquired on loan in 1986 from the St. Louis Zoo, is currently in the Jacksonville Zoo collection. These are both wild caught animals and have been in captivity for over 25 years without ever having successfully reproduced. Due to a bone deformity in the hind legs of the female, normal elevation of the shell for copulation is impossible. Frequent breeding attempts were observed in 1987 and seven infertile eggs were produced. The pair has been recently relocated from the tortoise runs to a new enclosure with a pool and rock outcroppings to provide possible aids for successful copulation. Investigations are underway to evaluate the feasibility of artificial insemination in this female, and efforts are being made to acquire additional breeding age females.

African Spurred Tortoises (*Geochelone sulcata*)

Three males, hatched at the San Antonio Zoo in 1979, were acquired on loan in 1985. Since their acquisition they have been housed in the off-exhibit tortoise runs, but their size had reached a point where this enclosure was no longer adequate. Between the time of their arrival in 1985 and the present, the mass of each has increased from 1.5 to 14.8 kg, 1.6 to 17.0 kg, and 2.0 to 12.5 kg, respectively. The three were recently

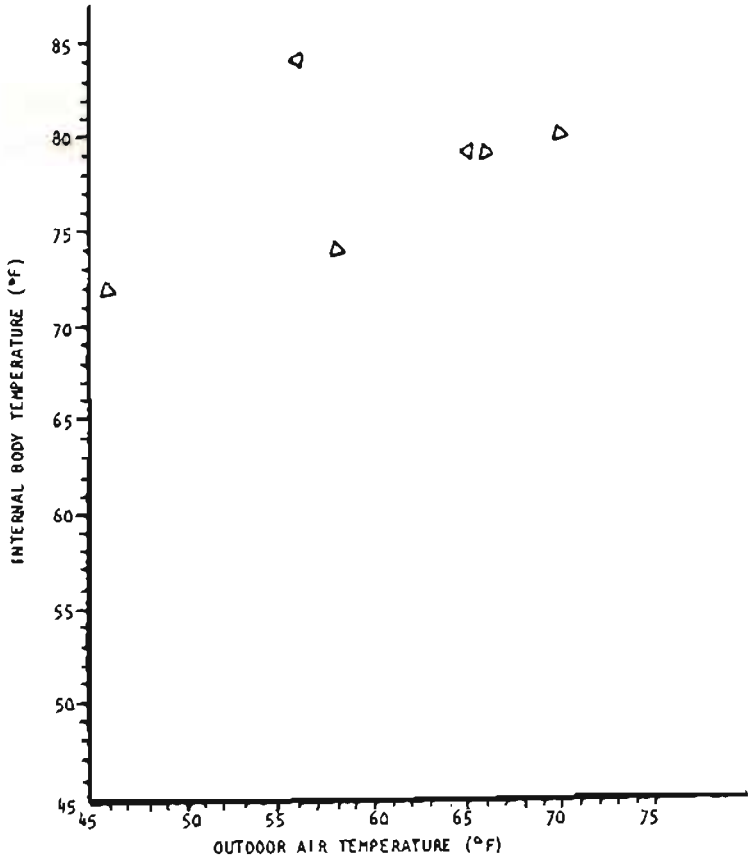


Figure 1. Internal body temperature maintained by female Aldabra tortoise in heated indoor quarters under varying outdoor air temperatures.

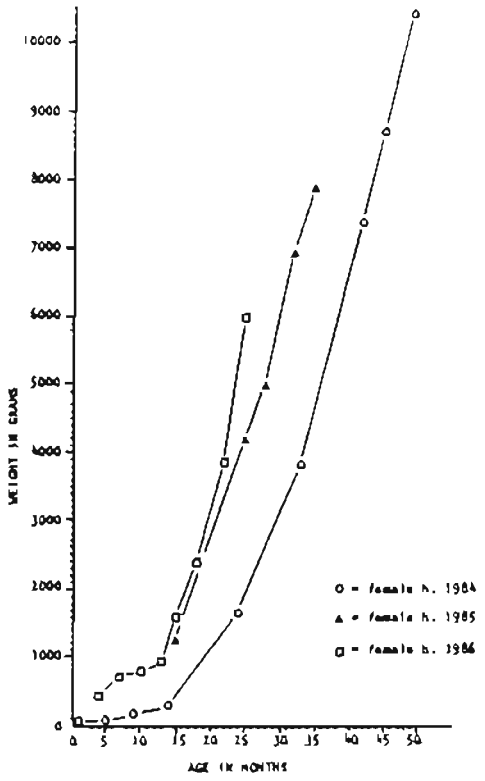


Figure 2. Growth of three Aldabra tortoises hatched at the Jacksonville Zoological Park.

HOUSING

Following hatching, all tortoises are located in the tortoise rearing room, where they are initially housed in 6" x 12" x 3" plastic boxes with pelleted rabbit feed as substrate. Depending on the tortoise and as growth dictates, they are transferred to five gallon terrariums with pelleted rabbit feed substrate until the age of two years. During the summer of the second year, the young tortoises are transferred to the off-exhibit tortoise runs, where they have access to exercise yards and free choice grazing on bahia grass or winter ryegrass. A more detailed description of this facility may be found in Williams (1986).

Winter heating in the off-exhibit tortoise runs consists of two overhead forced air heaters and one to two 250 watt heat lamps positioned over each cubicle. A steam vaporizer is utilized to counteract some of the drying effects of the forced air heaters; during a four week period in January and February 1988, while outdoor relative humidity readings averaged 84.5%, the indoor readings averaged 54%. During this same time period, while morning low temperatures outdoors ranged from 33° to 65°F, the indoor air temperature ranged from 58° to 79°F and the temperature beneath the rubber mat substrate ranged from 68° to 81°F. Within this facility during this time period, two adult star tortoises registered internal body temperatures ranging from 72° to 90°F (Fig. 5), while the three juvenile Aldabra tortoises registered internal body temperatures ranging from 72° to 91°F (Fig. 6).

Successful tortoise propagation and acquisition of animals has dictated the expansion of tortoise holding, rearing, and exhibit facilities. In June of 1988, new quarters to hold and exhibit the juvenile Aldabra tortoises and the adult radiated tortoises were opened. The inside areas are roomier than the previous quarters for these species, and the outside enclosures provide for a more varied environment. Total space for the radiated tortoises increased from 91 to 141 sq. ft., while the Aldabra tortoises have gone from 156 to 312 sq. ft.

Two 48" x 48" windows were placed on the south side of this new tortoise building for the winter solar heating effect; instead of glass, acrylic panels (Polycast UVT, Polycast Technology Corp.) were installed for winter use. This product has been shown to transmit medium wavelength UV light (295 nanometers) at 80% of that transmitted through air (Ullrey et al. 1986). In addition to vitamin D synthesis, other benefits of solar radiation include enhancement of physiologic processes and improved health of the integument of the chelonians (Behler 1987).

FEEDING

Tortoise keepers appear to vary widely in the precise diets and feeding regimens offered their charges, but a review of several references (Behler 1983; Blakely 1966; Burchfield et al. 1980; Burchfield et al. 1987; Davis 1979; Dickinson 1982; Frye 1981; Kirsche 1979; Louwman 1982; Samour et al. 1986; Throp 1969) indicates that the following generalizations may be made:

1. A diet high in roughage derived from fiber is desirable to maintain proper digestive tract function and prevent metabolic disturbances.
2. High protein diets are to be avoided, especially if they are at the same time low in fiber and high in moisture. Commercial diets range in their protein content from 14.3% (ZuPreem Herp Diet) to 35% (Zeigler Turtle Diet). The ideal for tortoises may be somewhere in the middle, as Frye (1981) recommends a diet of 19% protein for rehabilitating reptiles; diets containing 13% protein have proven adequate for growth and maintenance of snapping turtles (Frye 1981).
3. Adequate calcium must be provided and the proper balance with phosphorus maintained. In snapping turtles, diets containing 0.3% calcium and 0.2% phosphorus have proven adequate for growth and maintenance (Frye 1981). Two commercial diets provide levels ranging from 0.5% calcium, 0.4% phosphorus (ZuPreem Herp Diet) to 1.2% calcium, 1.0% phosphorus (Zeigler Turtle Diet).

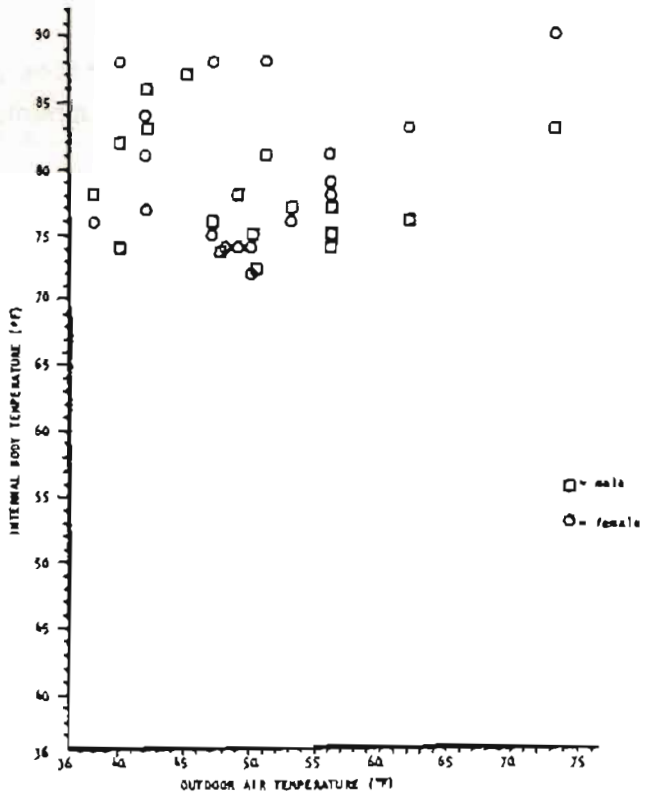


Figure 5. Internal body temperature maintained by two star tortoises in heated indoor quarters under varying outdoor air temperatures.

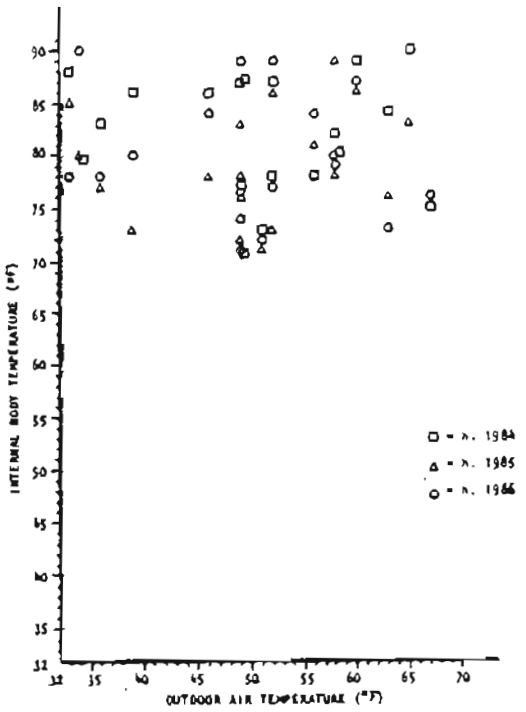


Figure 6. Internal body temperature maintained by three juvenile Aldabra tortoises in heated indoor quarters under varying outdoor air temperatures.

4. Frequency of feeding varied with the type of diet, with daily offerings of fruits, vegetables, greens, and hay being common, while two-to-three times a week was the rule for supplementation with higher protein (especially meats) diets and vitamin/mineral additives.

Diets fed to the tortoises at the Jacksonville Zoo are essentially unchanged since the early 1980s. The adult Aldabra tortoises are given their diet (Table 1) two times a week, and are either supplemented with alfalfa hay or have access to fresh pasture year-round. Results of a recent analysis of the Aldabra tortoise diet are included in Table 1; protein on a dry-matter basis was 20.4%, calcium 5.3%, and phosphorus 1.1%. Although no problems have been noted with this level of calcium, we are reviewing the diet for possible changes in the mineral supplementation.

All other tortoises in the collection receive the Prepared Tortoise Diet (Table 2) *ad libitum* three times a week, and they also (with the exception of hatchlings) have access to alfalfa hay or fresh pasture. This diet was also recently analyzed, and results (Table 2) on a dry-matter basis were protein 19.4%, calcium 2.4%, and phosphorus 1.2%. Growth rates of young tortoises are currently being monitored, and restriction of the amount offered may be necessary; the current formulation is believed to be adequate.

Hatchling tortoises, maintained indoors for the first one to two years, are fed the Prepared Tortoise Diet three times a week *ad libitum*. In addition, they are offered fresh greens or alfalfa sprouts on alternate days, following a soaking in fresh water. Growth and shell development are closely monitored. After four years of using this regimen on five species (*Geochelone elegans, G. carbonaria, G. gigantea, G. pardalis, Gopherus polyphemus*) no obvious problems have been noted and dietary revision has been unnecessary.

One additional dietary factor of known importance is vitamin D_3. During the past 20 years, the herpetological community has sought ways to correct deficiencies in the quality of light offered to animals housed indoors (Behler 1987). Emphasis has been placed on finding a safe and reliable artificial light source for the photobiogenesis of vitamin D_3 in the epidermis of the animal; results have been questionable at best (Ullrey et al. 1986; Watkins 1987). Vitamin D_3, the form that is active in reptiles, is required for proper calcium metabolism and prevention of metabolic bone disease (Frye 1981; Fowler 1986).

The Prepared Tortoise Diet at the Jacksonville Zoo has been analyzed for vitamin D_3, revealing a concentration of 2500 IU/kg of diet. As previously noted, this diet is fed to all tortoises regardless of their exposure, if any, to solar radiation. Animals that lack exposure to ultraviolet (UV) irradiation require a dietary source of the vitamin; there is, however, confusion over the amount that is considered adequate (Fowler 1986). Oversupplementation or hypervitaminosis D can be toxic and is associated with certain pathologic lesions, such as metastatic calcification (Frye 1981). To date, we have not had any indication of abnormal calcium metabolism in our tortoises, either clinically or at necropsy. In two commercial diets, the amounts of vitamin D_3 range from 607 IU/kg (ZuPreem Herp Diet) to 2700 IU/kg (Zeigler Turtle Diet).

LITERATURE CITED

Behler, J. 1983. Tortoise propagation at St. Catherines Wildlife Survival Center. AAZPA Annual Proc. 1983:87-94.

_____. 1987. Ultraviolet light and reptile propagation. AAZPA Annual Proc. 1987:162-169.

Blakeley, R. 1966. Notes on new diets for birds and reptiles at Chicago Zoological Park. Int. Zoo Ybk. 6:105-106.

Burchfield, P., C. Doucette, and T. Beimler. 1980. Captive management of the radiated tortoise, *Geochelone radiata*, at Gladys Porter Zoo, Brownsville. Int. Zoo. Ybk. 20:1-6.

_____, C. Hairston, and S. Huntress. 1987. Captive management of the Galapagos tortoise, *Geochelone elephantopus* ssp., at the Gladys Porter Zoo. AAZPA Ann. Proc. 1987:151-157.

Collins, D. 1984. Captive breeding and management of the Aldabra tortoise, *Geochelone gigantea*. Proc. 8th Ann. Reptile Symposium on Captive Propagation and Husbandry. 1984:36-45.

Davis, S. 1979. Husbandry and breeding of the red-footed tortoise, *Geochelone carbonaria*, at the National Zoological Park, Washington. Int. Zoo. Ybk. 19:50-53.

Dickinson, P. 1985. Maintenance, behaviour, and breeding of African spur thighed tortoise. Int. Zoo News 194:3-19.

Fowler, M. 1986. Metabolic bone disease. *In*: M. Fowler (ed.), Zoo and wild animal medicine. 2nd ed. W. B. Saunders Co., Phila. pp. 69-90.

Frye, F. 1981. Biomedical and surgical aspects of captive reptile husbandry. Veterinary Med. Publ. Co., Edwardsville, KS.

Kirsche, W. 1979. The housing and regular breeding of Mediterranean tortoises, *Testudo* spp. in captivity. Int. Zoo Ybk. 19:42-49.

Louwman, J. 1982. Breeding the six-footed tortoise, *Geochelone emys*, at Wassenaar Zoo. Int. Zoo Ybk. 22:153-156.

Samour, J., C. Hawkey, S. Pugsley, and D. Ball. 1986. Clinical and pathological findings related to malnutrition and husbandry in captive giant tortoises (*Geochelone* species). Veterinary Record 118:299-302.

Throp, J. 1969. Notes on breeding the Galapagos tortoise, *Testudo elephantopus*, at Honolulu Zoo. Int. Zoo Ybk. 9:30-31.

Ullrey, D., M. Strzelewicz, and S. Kaupp. 1986. Skylights and artificial lights for photobiogenesis of vitamin D_3. Proc. Amer. Assoc. Zoo Vet. 1986:66-67.

Watkins, B. 1987. Artificial lighting and vitamin D synthesis in birds. Assoc. Avian Vet. Today 1(2):59-60.

Williams, R. 1986. Tortoise reproduction at the Jacksonville Zoological Park. AAZPA Regional Proc. 1986:126-134.

Jacksonville Zoological Park

P.O. Box 26767

Jacksonville, FL 32218

Table 1. Adult Aldabra tortoise diet and analysis (dry matter) results. Jacksonville Zoological Park. Amount for one tortoise 2 x week.

3 large sweet potatoes (quartered)			
4 large apples (quartered)	Percent protein	=	20.4
4 large carrots (chopped)	Percent fat	=	10.2
2 whole bananas (peeled)	Percent fiber	=	2.9
1 head lettuce or greens (chopped)	Percent ash	=	17.5
4 cups bean sprouts	Percent Ca	=	5.3
1-1/2 lb. Bird of Prey Diet (Nebraska)	Percent P	=	1.1
4 Tbsp Vionate powder			
2 Tbsp oyster shell powder			

Table 2. Prepared tortoise diet and analysis (dry matter) results. Jacksonville Zoological Park. Fed ad libitum 3 x week

5 cups Purina Rabbit Ration (soaked 2:1 ratio in water)			
5 cups sweet potato (grated)	Percent protein	=	19.4
5 cups carrot (ground)	Percent fat	=	8.3
5 cups apple (grated)	Percent fiber	=	8.3
5 cups banana (peeled & chopped)	Percent ash	=	10.8
2-1/2 lb. Bird of Prey Diet (Nebraska)	Percent Ca	=	2.4
3 Tbsp Vionate powder	Percent P	=	1.2
3 Tbsp Bone-All powder			
2 cups alfalfa sprouts			

CAPTIVE MANAGEMENT OF THE TWIST-NECK TURTLE (Platemys platycephala), AT THE TULSA ZOOLOGICAL PARK

Darell C. Pickering

The twist-neck turtle, *Platemys platycephala*, is a small semi-aquatic turtle from northern Brazil, Venezuela, Colombia, eastern Ecuador, and Peru (Pritchard 1979). It has a very flat carapace which has a median groove. The plastron is large and flat. The neck is covered with barbels and there is a pair of barbels on the chin.

The Tulsa Zoological Park presently maintains a group of these turtles and the purpose of this paper is to give an account of their behavior and husbandry while at the Zoo.

HOUSING AND MAINTENANCE OF ADULTS

In 1981, 1.2 adult specimens were acquired. They were kept in a large stock tank until September 1982 when they were moved to their permanent exhibit. A third sub-adult female was added to the group in 1983.

The exhibit measures 2.4 x 1.7 x 2.4 m high and was originally a primate unit. It was renovated to simulate a tropical rainforest. Cement work was dyed and sealed with Aquapon to create a pool with a mud-like appearance. The small shallow pool, varying in depth from 9.0 cm to 25.4 cm, occupies approximately 1/3 of the available floor area. A gently sloping incline allows exit from the pool by the turtles to a basking and nesting area which measures 53.3 cm x 30.4 cm. This area is divided - half contains sand, half gravel and plantings. The remaining floor area has been rendered inaccessible to the turtles by steeply inclined walls at the end of the nest area and the remaining sides of the pool. This was done for exhibition purposes and keeps the turtles in the front half of the exhibit. The following chart shows carapace lengths and widths as well as masses of the four turtles.

	CL (cm)	CW (cm)	MASS (g)
Male	15.9	10.2	374
Female	14.4	9.7	361
Female	14.1	9.7	351
Female	12.4	8.8	220

Green crested basilisks (*Basiliscus plumifrons*) and Cuban treefrogs (*Osteopilus septentrionalis*) are also housed in the exhibit and have access to all parts of it.

The exhibit has a 138 x 138 cm skylight which is removed and replaced with screening during warm months, April - September. This allows unfiltered sunlight to enter the exhibit. Additional light is supplied by two four-foot Durotest Vita-lites® mounted at the front of the exhibit 2.0 m from the pool surface.

The exhibit is sprayed with water and the pool is overflowed daily. The pool is drained and cleaned once weekly, more often if conditions necessitate.

Ambient air temperature fluctuates seasonally with the winter range being 14° to 27.5°C and summer range being 13° to 35.5°C. Water temperature ranges seasonally from 15° to 27.5°C.

DIET

Adults are fed four times weekly: routine food items include earthworms, crickets, mealworms, waxworms, troutchow and a turtle gelatin diet similar to one described by Tryon (1978). Our gelatin consists of spinach, carrot, marmoset diet, Birds of Prey^{R} diet, green beans, shrimp meal, rabbit chow, monkey chow, bone meal, Vionate^{R}, and vitamins C and E.

Additional foods given are: minnows, pink mice, live brine shrimp and Birds of Prey^{R} diet.

All crickets are dusted with Vionate^{R}/bonemeal and a drop of Linatone^{R} vitamin supplement is given once a month in a food offering to each turtle.

The turtles are allowed to eat as much as they will consume. Mealworms and waxworms are limited to six or seven per turtle.

Plaster of paris blocks are kept in the water at all times.

COURTSHIP

Breeding and courtship appears year round, with higher occurances in the months of May, June, October and November. Courtship by the male begins with the male smelling the tail, vent, and rear marginals of the female. This is generally followed by a mount of the female's carapace by the male, who secures a podal clasp of her anterior and posterior marginals. Very rapid head and neck movements by the male to one side and then the other occur over the female's head and neck as he attempts to immobilize her or get her to withdraw into her shell. Biting and nipping can occur, generally by the male towards the female's head and neck. Females have also been observed to perform the rapid side-to-side head and neck movements while a mount by the male was occurring or when approaching another female. During the courtship described, the male actively performs a tail/vent search with his tail and extrusion of the intromittant organ may occur with no intromission attained.

A female mounting another female has been observed twice.

There seems to be a definite preference by the male for the largest female, with 56 recorded mounts and/or copulations between these two. The second largest female has 37 recorded mounts and or copulations. Courtship has never been observed with the smallest female. Gestation has not been determined.

The male can become excessively aggressive and is removed from the exhibit for several weeks during these times. Females will usually remain on the land area for extensive periods to avoid him. There is no schedule for these separations.

EGG LAYING

Egg laying has occurred in all months, except for January and October. Fertile eggs have been laid in March, May, June, September, November, and December.

Female *Platemys* show an expanded appearance between the carapace and plaston posteriorly several days or weeks before laying. Females exhibit prelaying behavior by smelling the nest area and by frequent diggings with the front feet. Unobserved egg laying or attempts are evident from the gravel substrate kicked or pushed into the pool by their activities. During nesting, females prefer to dig head first, at an angle, into the substrate to excavate a depression then move into typical egg laying posture over the depression. Eggs are usually left partially exposed. One egg was laid in the pool. A single egg is laid. Mean length is 52.8 mm (range = 49.0 - 56.5 mm); mean width is 27.5 mm (range = 27.0 - 28.6 mm); mean mass is 28.2 g (range = 22.3 - 29.6 g).

Since 1982, two eggs have been laid per year except 1983, when four eggs were laid. It has not always been determined as to which female is laying, but in most years each of the two older females has laid one egg. Of 14 eggs laid, nine have been fertile and five infertile. Seven of the nine fertile eggs have hatched. Six of the last seven eggs have been fertile and all have hatched.

The smallest female has never been observed laying or exhibiting nesting behavior. The largest female has laid four fertile eggs. The second largest female has laid two eggs, both of which were fertile.

INCUBATION

Eggs are removed from the exhibit after laying, then weighed and measured. A one-gallon glass jar is filled approximately 1/3 full of vermiculite and water of equal mass is added (Tryon 1975). The egg is placed on top of the vermiculite in the same position as it was found in the nest. The jar is sealed with a lid and placed into an incubator.

The incubator used is a large styrofoam box; an aquarium heater placed in a gallon jar filled with water maintains temperature and humidity. Temperature averages 27.7°C but fluctuates between 25.5° and 32°C.

As incubation progresses, fertile eggs change color from an off-white or pink to a pure white. This change begins as a white band in the center of the egg which progresses to one end and then more slowly to the opposite end. This may take several weeks.

Average incubation is 130 days (range = 119 to 143 days) for seven eggs that have hatched.

HATCHLINGS

Mean carapace length for young is 52.1 mm (range = 48.5 - 54.0 mm). Carapace width is 34.5 mm (range = 32.0 - 37.0 mm). Masses averaged 17.6 g (range = 14.9 - 19.7 g).

Upon hatching, young are left in the incubation jar until all yolk and amniotic membranes are absorbed or dropped and the hatchling is actively moving about. Young are placed into a rearing tub measuring 45 x 25 x 20 cm. Blasting sand is used for substrate and a flat rock is provided for basking. Water depth is maintained at 2.5 to 5.0 cm. A halved flower pot provides a refuge which the young use much of the time. A plaster of paris block is kept in the tubs of all *Platemys*. Vita-lites[R] are suspended 15.0 cm from the water's surface and a 60 watt incandescent bulb is located over the basking rock.

Hatchlings are fed the same diet as that given to the adults. They prefer live, moving foods at first but quickly accept prepared foods. Once established the young are housed together in plastic sinks.

SUMMARY

Platemys platycephala appears to be reproductively active year-round. The single egg and large hatchling size show a survival strategy of unigravida reproduction which is similar to several other chelonians.

Factors I believe have contributed to our present success with this species are: diet, exposure to unfiltered sunlight, periodic separation of the male from the females, good husbandry, and an exhibit design and layout that provides an unstressful environment to the turtles.

ACKNOWLEDGMENTS

I would like to thank Russell D. Grimpe and Linda C. Putnam for reviewing the manuscript and contributing to collection of obervational data over the years. I would also like to thank Jody Olewine for typing this manuscript.

PRODUCTS MENTIONED IN TEXT

Aquapon[R] - polyamide-epoxy concrete sealer manufactured by PPG industries, Inc. Coatings and Resins Division, Pittsburgh, PA 15222. Vita-lite[R] - full spectrum fluorescent tube made by Duro-Test Co., Bergen, NJ 07407. Birds of Prey Diet[R] - raptor diet prepared by Central Nebraska Packing, Inc., North Platte, NE 69101. Vionate[R] - Vitamin powder manufactured for Rich Health, Inc. Nutritional Research Laboratories, P. O. Box 18253., Irvine, CA 92713. Linatone[R] - Vitamin and Mineral supplement made by Lambert Kay, Cranbury, NJ 08512.

LITERATURE CITED

Pritchard, Peter C.H. 1979. Encyclopedia of Turtles. T.F.H. Publications, Inc. Neptune, NJ. pp. 778-779

Tryon, B.W. 1975. How to incubate reptile eggs: a proven technique. Bull. New York Herp. Soc. 11 (3-4):33-37.

Tryon, B. W. 1978. Some aspects of breeding and raising aquatic chelonians, Part 1. Herp. Review 9 (1):15-19.

Tulsa Zoological Park
5701 East 36th Street North
Tulsa, OK 74115

BEHAVIOR AND ECOLOGY OF THE ENDANGERED ENDEMIC: BERMUDA ROCK LIZARD (Eumeces longirostris)

Steven D. Garber

Bermuda's native terrestrial herpetofauna is limited to one species, the Bermuda rock lizard (*Eumeces longirostris*). The extreme isolation, small size, and recent geologic origin of Bermuda makes it unlikely that small lizards could survive such an oceanic journey, and the chances of establishing a population lasting hundreds of thousands of years is virtually impossible. Yet, this appears to be what happened.

E. longirostris was first described as an endemic form peculiar to Bermuda by Edward Drinker Cope in 1861. In a revision of the *Eumeces* genus, Taylor (1936) implied that this species reached Bermuda from the American mainland. In conversations I had in 1977 with this legendary herpetologist, shortly before his death, Taylor indicated he based his taxonomic decisions on the puzzling morphological peculiarities of *E. longirostris*. Unsure of this lizard's closest relative, he made the most conservative choice possible, stating circuitously that *longirostris* was closely allied to skinks occurring in the Eastern United States. He placed *E. longirostris* in a group all its own (monotypic group), leaving systematists in a quandary for the next 50 years. No one was sure whether the Bermudian skink had originated from African, South American, or North American stock.

Since Taylor's *Eumeces* opus, the development of new taxonomic techniques have enabled researchers to decipher the origins of certain species about which little was previously known. One deterrent to tackling some of these projects is that they require samples from around the world, as well as a working knowledge of the many species belonging to the group in question, the family Scincidae and the genus *Eumeces*. Alone, *Eumeces* is a formidable species assemblage for any systematist to study, considering it includes hundreds of species worldwide.

The extinction of many Bermudian species occurred during the Pleistocene when the Belmont Sea stood more than 100 feet above the current sea level, submerging most of Bermuda. This completely eliminated all the marsh habitats required by species such as the crane (*Baeopteryx latipes*) and the duck (*Anas pachysceplus*), which went extinct. But the skink, the soil burrowing cahow (*Pterodroma cahow*) and a finch were able to survive on the smaller land area, and are represented in later deposits.

The first report of any of these species came when Captain John Smith mentioned the presence of lizards on Bermuda in his "History of Virginia" (1623) where he wrote, "Lizards there were many and very large but now gone and it is said they were destroyed by the cat." From Smith's account, one would think a lizard larger than the skink lived there previously. Samuel Garman (1884) pointed out that Strachy (1610) and Rev. Hughes (1614) both failed to mention any lizards and would certainly have done so if some large species had been there. It is inconceivable that the skink living there now could have attained substantially greater lengths several hundred years ago. I have kept a series of Bermuda skinks in captivity from 1977 and at best, they only get about seven inches long.

Since the Bermudian fossil record lacks any evidence suggesting a large lizard, and since Smith based his description on the accounts of others, never having been in Bermuda himself, either the size of the lizard was exaggerated, or perhaps Bermuda was confused with another island. Considering how many people confuse Bermuda with the Bahamas, this is a distinct possibility. There is, in fact, a large lizard in the Bahamas, a ground iguana in the genus *Cyclura*.

Concerning Smith's statement that the lizards were numerous, we have little recourse but to discount his comments. Matthew Jones stated in 1859, before any amphibians or other reptiles were introduced there, that the Bermuda rock lizard was very common on old walls and in stone heaps in the cedar groves. Describing the status of these lizards in 1900 after the marine toad (*Bufo marinus*) was established, but before the arrival of any *Anolis* species, A. E. Verrill (1902) wrote, "this lizard which is a very active species, is by no means common, except in particular localities." He saw very few lizards except on Castle Island where they are common along the ruins of the old forts and also in the crevices of the cliffs. A few individuals were seen in certain places on the Main Island, in walls, but it was regarded as rare by the native, many of whom had never seen it at all.

Verrill's statement, particularly the reference to Castle Island and the shore cliffs, applies to the skink's status as it appears today, following the establishment of *Eleutherodactylus* and *Anolis* from the West Indies. David Wingate's observations (1965) indicate skinks may be more common inland than they appear, feeding undetected on the ground beneath vegetation and retreating under rubble, stones, and logs for cover.

Since the lizard is so rare, and has been for many years, I felt it was important to investigate the skink's behavior and to see if, as Wingate thought, they really are more common than people realize. I observed hundreds of individuals in the field and kept 35 in captivity from 1977 to present (Simbotwe and Garber 1979), and found that, although they spend some time undetected beneath the substrate, wherever they occur, they are found sunning and feeding, as well as interacting agonistically and sexually with their conspecifics.

Wingate's conclusions were, in my opinion, too optimistic. While some lizards occur in coastal areas and inland quarries, they are abundant in very few localities. The Castle Harbour Islands and the north shore in Devonshire are two localities where they are abundant. The lizards also occur on several of the smaller islands, but appear to have disappeared from most of Bermuda's mainland.

On the Castle Harbour Islands, which are relatively pristine compared to the mainland, several populations still occur. And recently, even the populations on Castle and Charles Islands (each of which are three square acres), have declined. This may be attributable to the litter left by picnickers. People stop in their boats, get out, and have a good time, leaving bottles and cans with enticing odors that attract skinks. These lizards are unusual, having an especially keen sense of small. This litter kills off many lizards. The cans and bottles are usually wet and slippery inside, and the inside temperatures reach high levels during the day, temperatures which are far too high for any vertebrate to survive for long. The lizards that crawl inside either drown or bake. Skeletons found in the sludge in each can and bottle attest to this.

Although these islands are national parks, litter is rampant due to the heavy usage. Also, the recently introduced kiskadee flycatcher (*Pitangus sulphuratus*) feeds on lizards (Samuel 1975).

The largest Castle Harbour Island, Nonsuch, is a 15-acre restricted access nature preserve, and is the only locality where *E. longirostris* is still abundant. The longterm goal of the Bermuda Government is to restore this National Park as a living museum of primeval Bermuda. David Wingate is trying to maintain and/or restore all the original plants and animals. This means keeping the island free of introduced species. He has been successful at keeping the kiskadees from becoming established by shooting the constant stream of immigrant birds coming from Bermuda's mainland. Wingate has also kept cats, rats, mice, toads, and whistling frogs (*Eleutherodactylus gossei* and *E. johnstonei*) off. And it seems the island is finally free of *Anolis* lizards.

The skinks are morphologically peculiar. As their name, *Eumeces longirostris*, states, they have an elongated rostrum or snout. Field and captive observations have shown that this alert lizard is not only an insect predator, but also a carrion eater, unlike most reptiles. I first learned this during lunch one day when a skink came out of a crevice and ate some of my ham and cheese sandwich.

It is not only olfactory stimulation that draws the lizards from the rock crevices; visual stimulation also brings them to potential food sources. Lizards were able to find cheese when it was placed in opaque containers open only at the top. They were also able to get inside the containers by climbing down a ridge and crawling or leaping into the container. This is how I collected the series I studied.

While Bermuda was rodent-free for hundreds of thousands of years, it appears that the skinks evolved almost mammal-like traits. Before humans arrived, much of Bermuda was a huge bird rookery, providing a virtually unlimited food supply for any enterprising animal that could lick out the insides of hatched eggs, eat fish remnants discarded by the birds, or prey on the large supplies of insects attracted to the abundant refuse. Apparently the Bermuda rock lizards became that opportunist.

In recent years an estimated 50 cats ran wild on the Frick property at Castle Point (on the mainland) and, during this time, Dr. Henry Clay Frick II observed the *E. longirostris* population there decline precipitously. Dr. Frick has since eliminated the cats, but careful observations there failed to reveal any lizards. This may be a reasonable locality for a reintroduction effort, one which the Fricks would welcome.

An effort like the success of the past four decades of work to protect the cahow by Childs Frick, Louis Mowbray, Robert Cushman Murphy, and David Wingate should be repeated with a new generation of dedicated individuals. This time the goal should be to insure a future for Bermuda's endemic lizard.

LITERATURE CITED

Cope, E D. 1861. On the Reptilia of Sombrero and Bermuda. Proc. Acad. Nat. Sci. Phil. 13:312-314.

Garman, S. 1884. The Reptiles of Bermuda. Bull. U. S. National Museum. 25:285-303.

Jones, J. M. 1859. The Naturalist In Bermuda. London Reeves and Turner.

Samuel, D. 1975. Feeding habits of the Kiskadee Flycatcher. Bermuda Biological Station Newsletter 4(5):3-4.

Simbotwe, M. P. and S. D. Garber. 1979. Feeding habits of lizards in the genera *Mabuya, Agama, Ichnotropis,* and *Lygodactylus* in Zambia, Africa. Transactions of the Kansas Academy of Sciences 82(1):55-59.

Smith, J. 1624. The Generall historie of Virginia, New England, and the Summer Isles, London.

Taylor, E. H. 1936. A taxonomic study of the cosmopolitan scincoid lizard genus *Eumeces* with an account of the distribution and relationships of its species. Kansas Univ. Sci. Bull. 23:1-643.

Verrill, A. E. 1902. The "Cahow" of the Bermudas, an extinct bird. Annual Magazine of Natural History 7(9):26-31.

Wingate, D. 1965. Terrestrial herpetofauna of Bermuda. Herpetologica 21(3):202-218.

Department of Biological Sciences

Nelson Hall

Busch Campus

P.O. Box 1059

Rutgers University

Piscataway, NJ 08854

COMBAT AND COURTSHIP OF THE EASTERN MASSASAUGA RATTLESNAKE -- COMPARISON OF FIELD AND CAPTIVE BEHAVIOUR

Bob Johnson

INTRODUCTION

The increased interest in the Eastern massasauga rattlesnake (*Sistrurus catenatus catenatus*) is, in part, a result of the recent efforts to protect fragmented populations of this species. Decline in habitat, as with most threatened species, remains the most serious threat to rattlesnake populations. In Ontario, the fear of venomous snakes results in the persecution of this species near cottages and farms. However, because of its shy, retiring nature, the massasauga rattlesnake does not suffer the bad press of many other crotalids and several populations are protected within provincial and federal parks.

The Eastern massasauga rattlesnake was kept at Metro Toronto Zoo for eight years and observations of four captive breedings into the second generation were recorded.

THE EASTERN MASSASAUGA RATTLESNAKE IN ONTARIO

Population strongholds in Ontario extend along the shores of Georgian Bay (Fig. 1) where remote areas serve as refuges from which local populations may be replenished. Increasingly, however, development is fragmenting populations and gene flow or movement from refugia is restricted. Small, disjunct populations remain in the Lake St. Clair and Wainfleet Marsh, Niagara areas.

Fortunately, self-sustaining populations are found within several reserves and national and provincial parks. Both provincial and federal governments have adopted an enlightened attitude towards the presence of this species and each has published information pamphlets explaining the need for preservation. In fact, Beausoleil Island, in Georgian Bay Islands National Park, is unique in that campers and rattlesnakes share the same habitat. Snakes on Beausoleil Island are found within 100 meters of campgrounds, regularly cross trails, and are occasionally captured in the middle of campgrounds (Parks Canada 1984).

A radio-telemetry study of massasauga rattlesnakes on Beausoleil Island found that the snakes did not hibernate communally. It was also confirmed (Wright and Wright 1957; Reinert 1982) that Eastern massasaugas prefer moist sites during hibernation and dry, upland sites in summer. Localized wet areas were often utilized instead of the larger wetlands (Parks Canada 1984). Although there was no evidence of homing behavior (Reinert also found that displaced specimens exhibited no homing behavior), at least one snake used the same route when moving to and from its winter hibernaculum. The need for habitat mosaics illustrates both the importance of such field studies and the difficulty of preserving habitat for this species.

Several late-August breedings were confirmed on Beausoleil (see also Klauber 1972 and Reinert 1981) and snakes were often encountered in close proximity to each other. Snakes were observed at rest with their bodies aligned and the head of one on the back of the other. Male snakes also exhibited this posture. In one area, three males followed a single female over a period of 35 days and males were found close to females after moving about 100 meters, independently of each other, after their release.

It seems males were using chemical cues to trail females and it is suggested that chemical attractants might keep males in close proximity to females and even confuse males as to the sex of individuals they encountered. These observations are significant in that they give us some insight into the behavior we observed during courtship and copulation in captivity.

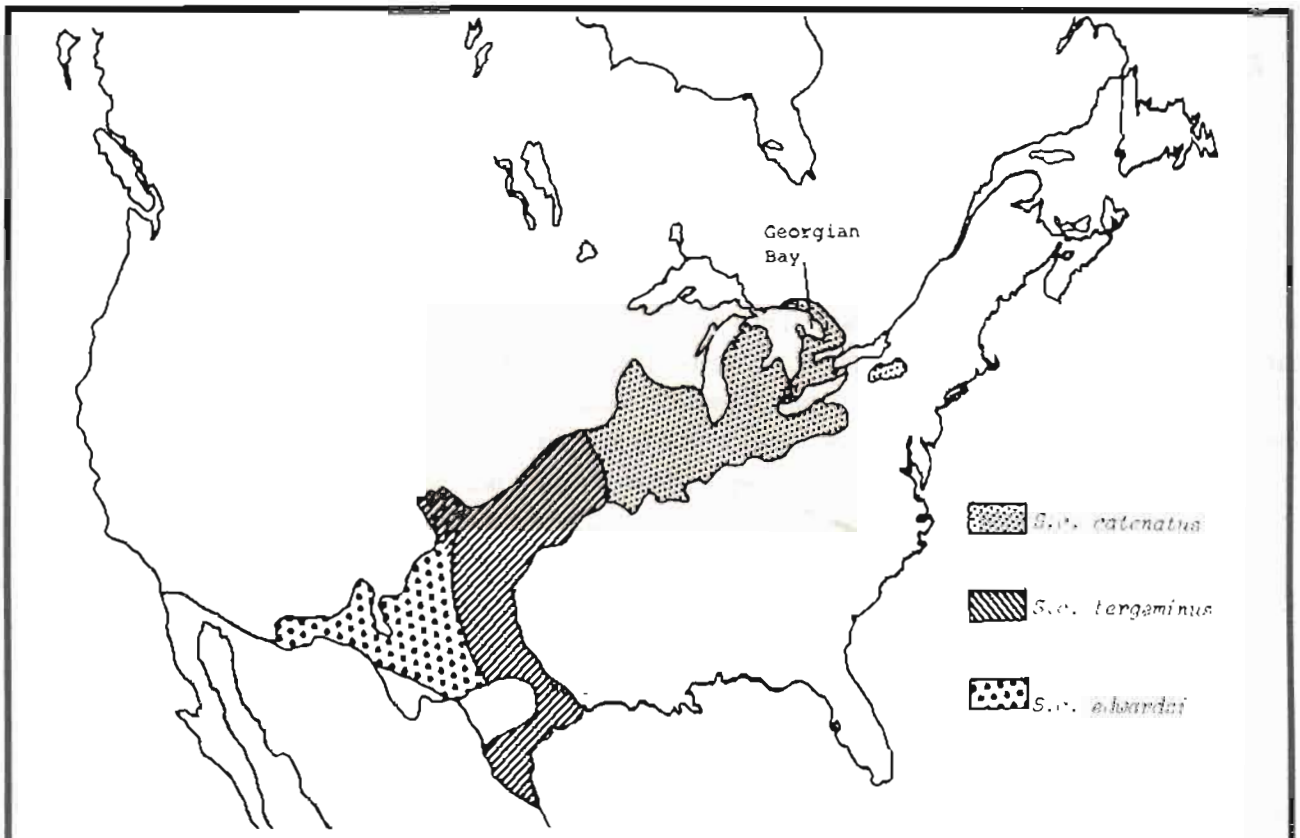


Figure 1. Range of *Sistrurus catenatus* (after Conant 1975).

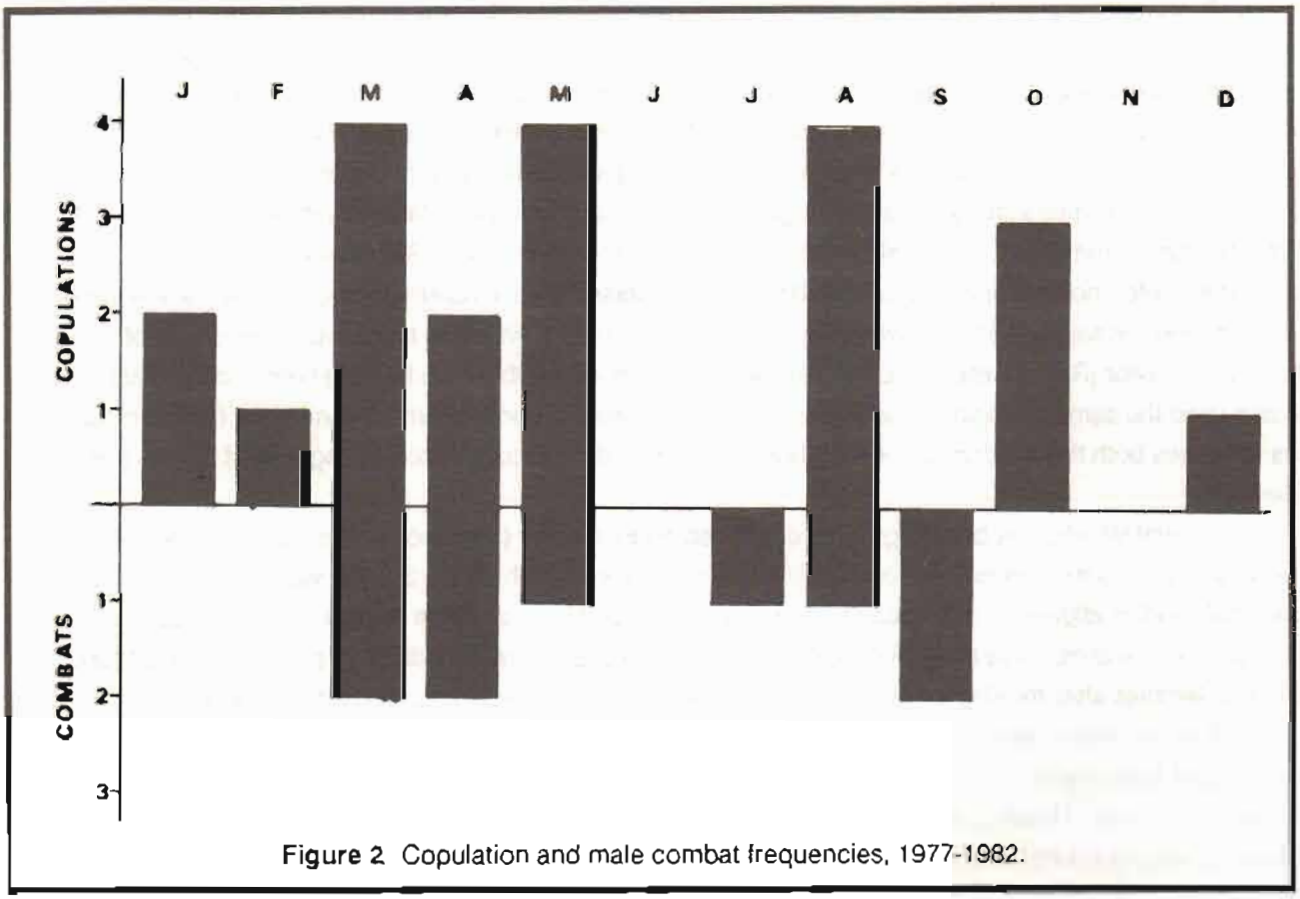


Figure 2. Copulation and male combat frequencies, 1977-1982.

MALE COMBAT AND REPRODUCTION AT METRO TORONTO ZOO

Three male and two female massasauga rattlesnakes were housed in an exhibit four meters long and one-and-a-half meters wide. Photoperiod varied between eight hours of light in winter and thirteen hours of light in summer. Temperatures fluctuated in summer from 30 to 25 degrees centigrade, and in winter from 27 to 22 degrees centigrade. These minor summer to winter temperature fluctuations would not be significant in stimulating reproduction. We suggest that reproduction was enhanced by a large exhibit and more than one pair of snakes.

Captive snakes were fed on dead mice once per week over the three months of spring and once every two weeks for the remainder of the year. In the wild, massasaugas prefer to feed on small mammals and snakes (Seigel 1986). Leopard frogs housed with massasaugas were not preyed upon (as suggested by Seigel 1986), although they occasionally received a bite (usually dry) during the excitement of feeding on dead mice.

We observed two seasons of active display and courtship and, as might be expected, combat was associated with an increase in copulatory behavior (Fig. 2). Male combat was recorded March, April, and May and again in July, August, and September. Copulations occurred most frequently in March, April and May, and in August and October. Copulation (Fig. 3) could be prolonged, lasting up to five hours, and on several occasions the female would move forward, dragging the male along with her.

Ritualized combat between males (Fig. 4) frequently occurred with a third snake, usually a female, present in the immediate area (Fig. 5). One male was observed in combat posture above a pair of copulating snakes, and a pair of snakes copulated while lying on top of a second male which had fought previously with the breeding male (Fig. 6).

Typically, the topping of one male over the other occurred when a male was able to rise above the other and fling its head and anterior part of the body down upon the second male. A great deal of head jerking preceded and followed ritualized combat. Head jerking also occurred without topping behavior. As suggested by observations of male snakes in the wild (Parks Canada 1984), the presence of a female may excite and confuse males (Akester 1983). In the presence of female chemical cues one male may inadvertently court another male (Fig. 7).

Litters of three, four, six, five, and six were born at Metro Toronto Zoo. Neonates from one litter had an average mass of 12.9 $\pm$ 1.6 g, and averaged 21.5 cm in length. Earliest postnatal sheds of three litters occurred after seven, eight, and four days.

As with other captive bred snakes, massasauga rattlesnakes became sexually mature in advance of their third year (Bielema 1973). The first litter of massasaugas, born at Metro Toronto Zoo, became reproductively active (combat and breeding) 25 months after birth (for a male) and two captive bred females gave birth 34 months after birth. A pair of second generation captive bred snakes born in April, 1980, bred and subsequently gave birth in June, 1982, at 27 months of age. One female gave birth to two litters within a six month period, on 10 April and again 3 October.

DISCUSSION

We observed that the initial stages of mating behavior and combat behavior are similar. It is not clear whether dominance behavior includes head jerking and tail looping or whether one male was confused as to the sex of the other snake. It has been suggested that male interactions without subsequent combat may be due to the breakdown of male aggression when each believes it is courting a female. Wild male snakes have also been found resting with other males without visible signs of combat or dominance. However, we also observed males in combat while resting with a female. It remains a possibility that female chemical cues would confuse males (as suggested by Akester 1983).

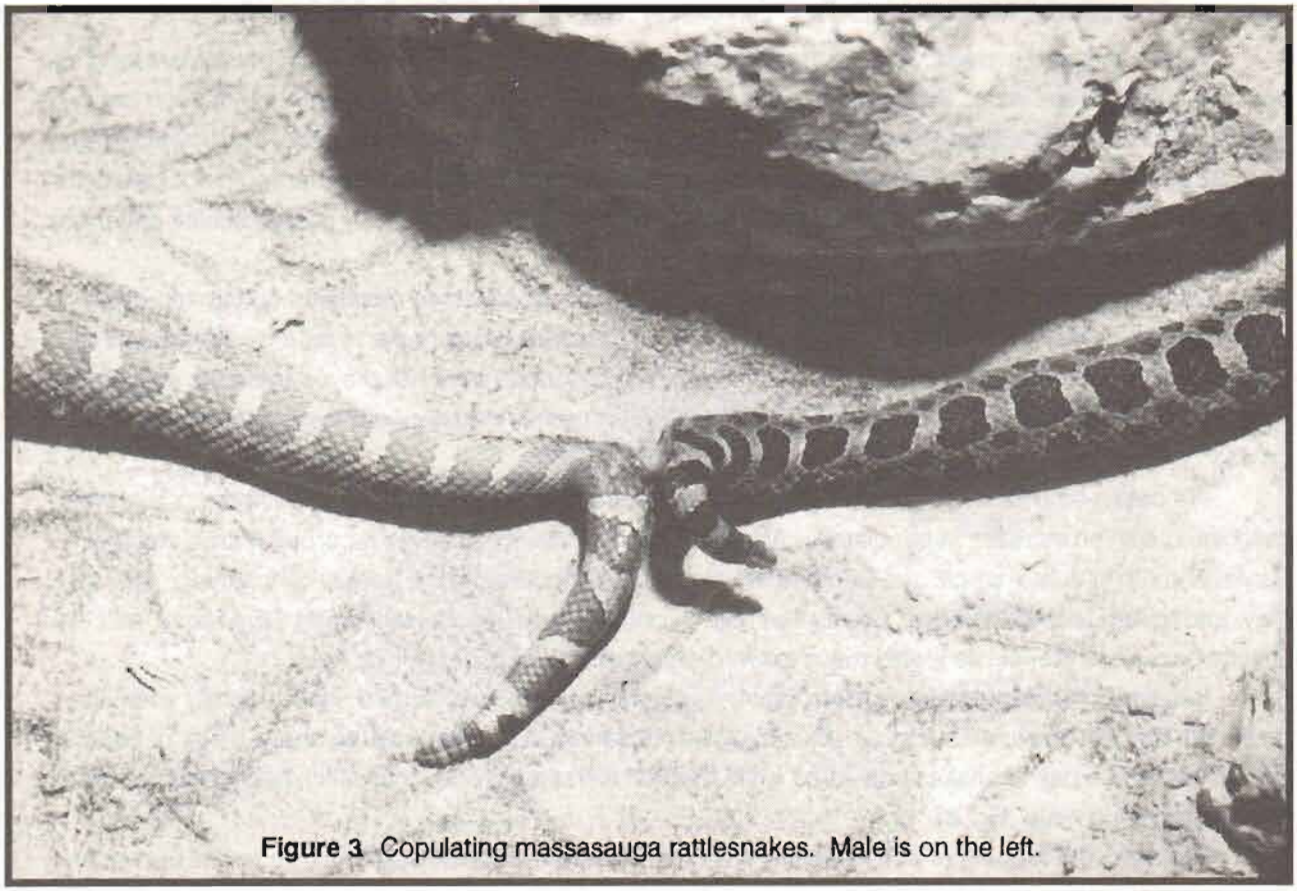


Figure 3. Copulating massasauga rattlesnakes. Male is on the left.

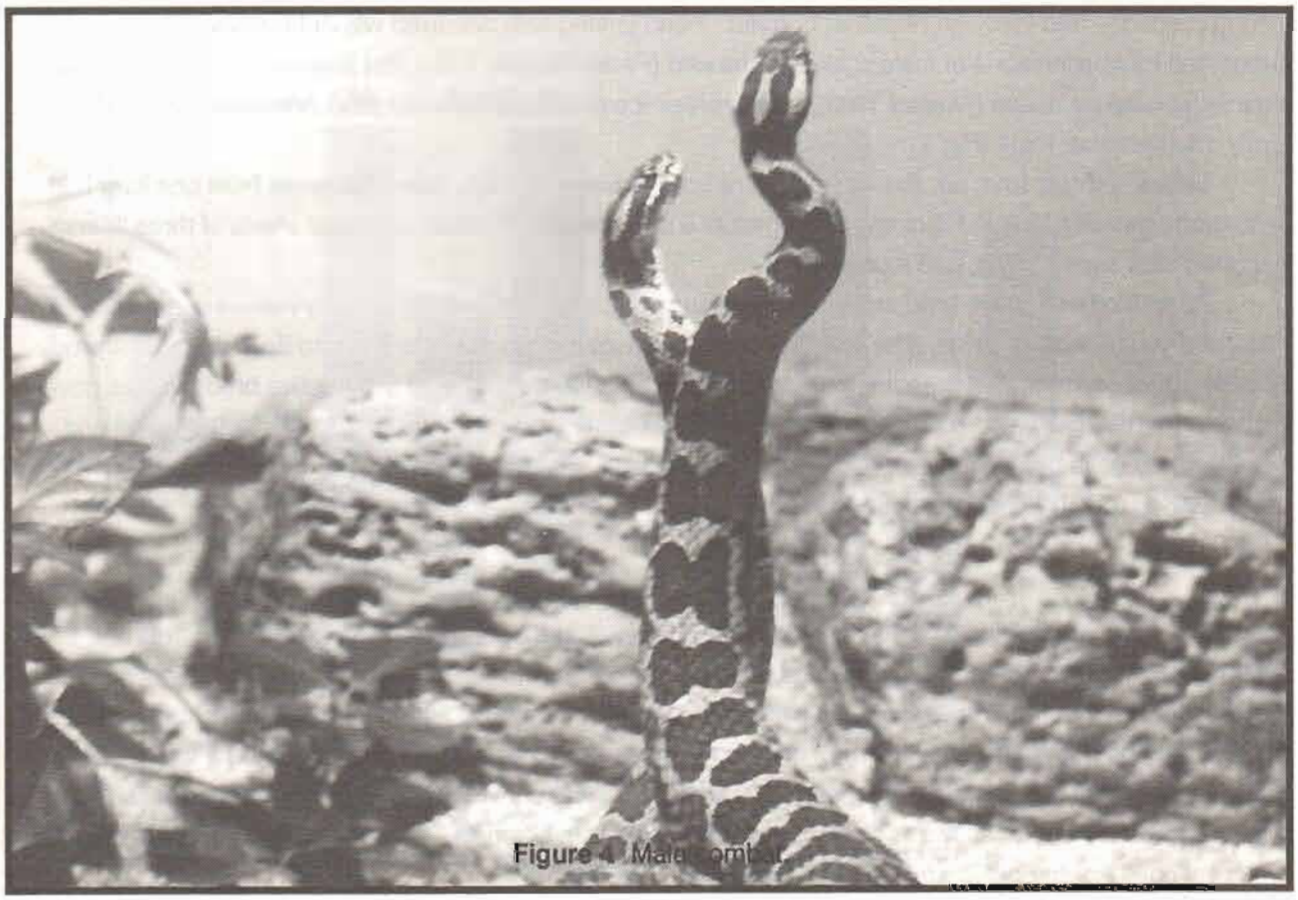


Figure 4. Male combat

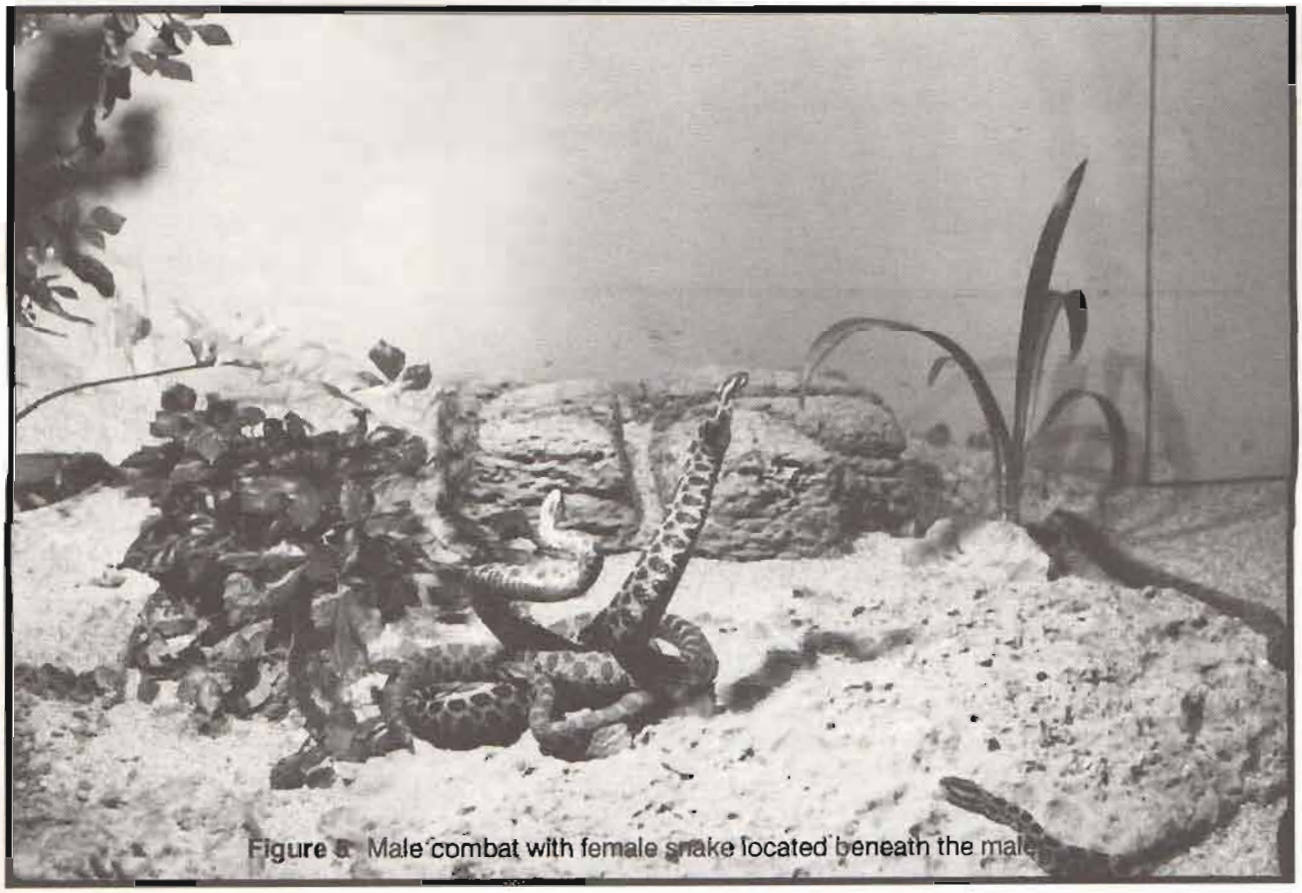


Figure 8. Male combat with female snake located beneath the male.

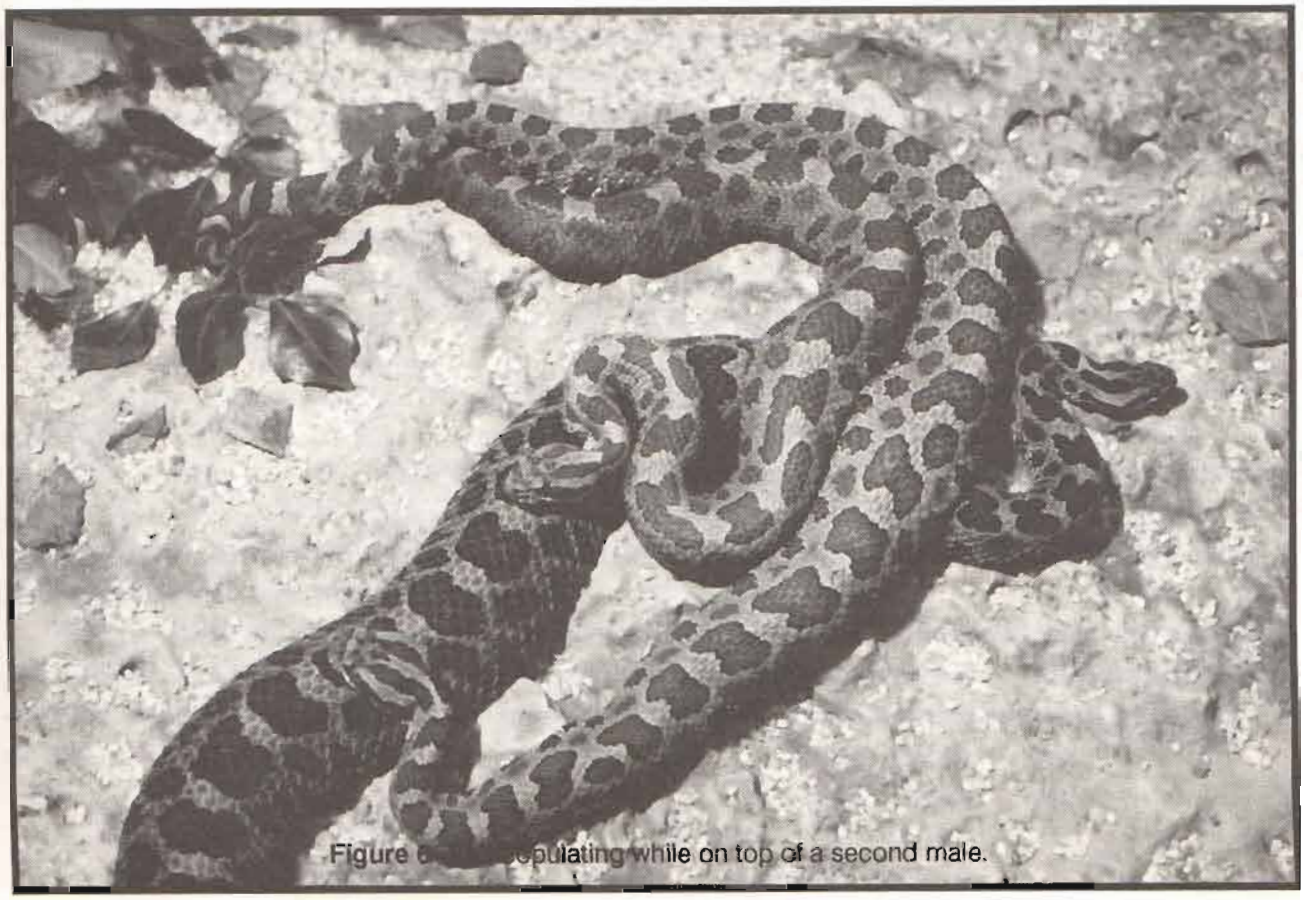


Figure 9. [illegible]opulating while on top of a second male.

Figure 7. Two males in copulatory alignment

Although it has been reported (Gillingham 1983) that subordinate male *Crotalus atrox* were refractive and would not copulate for up to an hour after losing a combat bout, we observed that subordinate male massasaugas copulated with females after losing combat bouts. However, neither snake bred immediately after combat, appearing too exhausted to do so. Several different males were observed to copulate with the same female over a three week period. The subordinate refractive period may provide an opportunity for the dominant male to breed, but does not preclude copulations by other males following the combat period.

A dominant male was observed in ritualized combat with two males on seven occasions over a four month period. This dominant male was not observed to copulate with a female during this period. However, one of his combatants was observed breeding for three hours. The "dominant" male displayed a combination of courtship and combat behavior while the other snakes copulated. This included chin rubbing, body jerks, tail wrapping, and topping behavior.

REPRODUCTIVE PLASTICITY

The factors which caused a female to give birth twice in a six month period may be the result of captive conditions, but nonetheless, it is significant that these snakes at least have the ability to produce two litters in one reproductive season. Based on a 1:1 ratio of gravid to non-gravid females, massasauga rattlesnakes may be biennial breeders (Reinert 1981). Although knowledge of the predominant reproductive cycling period is indeed significant in terms of the ecology of local populations (Reinert 1981), our understanding of reproductive potential and plasticity may tell us something of the environments in which this species evolved (see Aldridge 1979 and Schmidt 1938).

The cycling of lipid reserves may provide more understanding of reptile reproductive cycles (Johnson 1987). If massasaugas are capable of breeding in captivity twice a year because of high lipid reserves, we may speculate that when fat reserves are depleted the snake shifts into a biennial reproductive pattern. Reproductive potential is, then, less a result of periodicity and more a result of nutritional history -- shifts in reproduction match shifts in environmental opportunism. To be sure, limited opportunity for growth during short summers may result in a similar periodicity of reproduction, but the effects of optimal and sub-optimal habitat would account for annual, biennial, and triennial reproductive seasons in a single population. Snakes with long periods of winter dormancy, and those which frequent poor quality habitats, might be expected to breed biennially. This, of course, has implications for the management of wild populations (particularly those which make up the edge of range extensions) and may explain geographic variations in reproductive potential (Reinert 1981).

The reproductive plasticity of this species may account for its success after the historical retreat of its prairie habitat which had extended into northeastern North America in the post-glacial period (Schmidt 1938). Biennial reproduction could be the result of the historical loss of a habitat associated with a warmer period. The seasonal shift in habitat use to warmer, drier upland sites in the summer and for gravid females in particular (Reinert 1982) may be an example of habitat substitution for a species forced to adapt to a loss of preferred habitat as a result of climate change.

The plasticity of captive populations simply emphasizes the need for further field studies of local populations and comparative studies throughout the range of this threatened species. The management of wild populations will be facilitated by an understanding of adaptation to the decreasing number of isolated habitats.

ACKNOWLEDGMENTS

Parks Canada staff and, in particular, Michel Villeneuve of Georgian Bay Islands National Park, shared with me personal observations of the massasauga rattlesnake in the field and provided an unpublished radio-tracking study of massasauga rattlesnakes. Sandi Burden typed the manuscript and Miriam Richards prepared the figures.

LITERATURE CITED

Akester, J. 1983. Male combat and reproductive behavior in captive *Bitis caudalis*. Brit. J. Herpetol. 6:329.

Aldridge, R. D. 1979. Seasonal spermatogenesis in sympatric *Crotalus viridis* and *Arizona elegans* in New Mexico. J. Herpetol. 13(2):187.

Bielema, B. J. 1973. The eastern massasauga in west-central Illinois. M. Sc. Thesis, Western Illinois University.

Conant, R. 1975. A Field Guide to Reptiles and Amphibians. 2nd Ed. Houghton Mifflin Co., Boston.

Gillingham, J. C. 1983. Courtship, male combat and cominance in the Western diamondback rattlesnake. J. Herpetol. 17(3):265.

Johnson, B. 1987. Voluntary hypothermia in amphibians and reptiles. 11th International Symposium on Captive Propagation and Husbandry. Zoological Consortium, Thurmont, MD. Pp. 7-16.

Klauber, L. M. 1972. Rattlesnakes: Their Habits, Life Histories, and Influence on Mankind. 2nd Ed. Univ. Calif. Press.

Parks Canada, 1984. Resource management study of the massasauga in Georgian Bay Islands National Park. Unpublished.

Reinert, H. K. 1981. Reproduction by the massasauga. Amer. Midl. Nat. 105(2): 393.

_____ 1982. Movements and habitat utilization by the massasauga, *Sistrurus catenatus catenatus*. J. Herpetol. 16(2): 162.

Schmidt, K. P. 1938. Herpetological evidence for the postglacial extension of the steppe in North America. Ecology 19:396.

Seigel, R. A. 1986. Ecology and conservation of an endangered rattlesnake, *Sistrurus catenatus*, in Missouri. Biol. Conserv. 35:333.

Wright A. H. and A. A. Wright. 1957. Handbook of Snakes. Comstock, London.

Metro Toronto Zoo
P.O. Box 280
West Hill, Ontario M1E 4R5
Canada

STUDIO ELAPHE - COLOR AND PATTERN BREEDING PROJECT - A NEW OCCURRENCE OF TYROSINASE DEFICIENT ALBINISM IN Elaphe obsoleta

Lee Robbins, M.D.

ABSTRACT

Studio elaphe is a color and pattern-breeding project begun in 1978. Various pattern and color aberrancies have been explored through selective breeding over the last 10 years. In 1985, there was a new occurrence of tyrosinase deficient albinism in offspring of a greenish rat snake.

INTRODUCTION

In 1966 I obtained four hatchling rat snakes from Gordon Johnston. These were red, yellow, grey, and black (*Elaphe gutatta gutatta, E. obsoleta quadrivittata, E. o. spiloides,* and *E. o. obsoleta*). The juveniles of *Elaphe* are disconcertingly similar in coloration (Bechtel and Mountain 1960) and give little indication of the brilliant hues of the adults. I was 14 at the time and all four escaped. In 1978, as artist and physician, conditions were ripe for a deliberate exploration of color and pattern.

The artistic premise has been that a snake is line and color. At the outset, the collection was ravaged by an amoeba epidemic and lengths of snake were embedded in polyester resin as lines in sculpture. Macrophotographic studies of pattern (1982) were used in preparation for a series of 51 oil paintings (1983). Paintings, sculptures, and live snakes were exhibited in an East Village art gallery in 1985.

The goals of this research are to unmask melanin in *Elaphe obsoleta* and to understand the genetics of pattern inheritance.

GENETICS

Albinism in *Elaphe obsoleta* has been widely documented (Dyrkacz 1981) and is inherited as an autosomal recessive trait. Bechtel (1981) described three forms of albinism in *Elaphe obsoleta*. These were differentiated on the basis of reaction of the enzyme tyrosinase upon tyrosine to produce DOPA(3,4-dihydroxyphenylanine) and upon DOPA to produce dopaquinone, which is converted to melanin without need for tyrosinase (Lerner and Fitzpatrick 1950).

The black rat snake juvenile is normally darkly blotched on a gray ground color with color becoming more uniformly black with adulthood. The juvenile pattern can be discerned in some individuals, especially when distended by a meal. Using the DOPA reaction, a histochemical test, Bechtel found three forms of albinism in *Elaphe obsoleta*. Tyrosinase deficient albinism is usually seen as pale reddish blotches on a white-to-yellow ground color. Tyrosinase positives occur as a hypomelanistic xanthic (yellow-tan color with slight melanin production) and a fully albino form which usually has more red pigmentation than the tyrosinase deficient form, and a pinkish ground color. Both amelanistic types have red eyes.

Corn snake coloration is dependent on four pigments: white (leucistic), yellow (xanthic), red (erythristic), and black (melanin) (Wagner). Two spontaneous mutations have occurred in *Elaphe gutatta gutatta*. The first described was an amelanistic partial albino, resulting from a single recessive gene mutation (Bechtel and Bechtel 1962). The next defect described was an inability to produce red pigment (Slemmer 1973). This resulted in a black corn snake. Snakes have since been produced that are unable to produce red or black pigments and these have been termed "snow corn" snakes.

A striped pattern mutation was found to be of autosomal recessive inheritance (Bechtel and Bechtel 1978). The corn snakes were of normal color with a dorsal stripe instead of the usual reddish-orange blotches on reddish-brown to gray ground color. These animals also had loss of the usual ventral checkerboard pattern. Interestingly, head pattern was unchanged.

The striping mutation was also associated with an unmarked ventral surface in *Pituophis* in 1958 by Riemer (1958). Striping in *Pituophis melanoleucus* was found to be dominant over the blotched pattern (Bechtel and Whitecar 1983). Zweifel (1981) also found association of light belly with striping in the California kingsnake (*Lampropeltis californiae*) and striping, a geographically isolated phenomenon, was also described as dominant over the banded form.

In the previous examples, a linkage of dorsal striping and ventral pallor was inferred. However, in a corn snake currently in the Studio Elaphe collection, a completely normal checkerboard pattern is seen. Pattern and color are independently inherited with head markings showing unique constancy.

MATERIALS AND METHODS

The collection numbers approximately 150 snakes housed in glass fish tanks, plastic sweater and shoe boxes. All animals in the collection are fed warm-blooded prey, primarily rodents.

The full adult population is hibernated from early December to March 1 at 55°F. Animals are segregated by sex. Photoperiod is natural in the Manhattan loft where this takes place.

In 1982 six hatchling greenish rat snakes (*Elaphe obsoleta obsoleta* X *rossalleni*) were obtained from Dr. Peter Weber, Professor of Biochemistry at Albany Medical College. These were produced by mating a wild-caught black rat snake (*E. o. obsoleta*) from Albany, NY and an Everglades rat snake (*E. o. rossalleni*).

The usual intergradation of black and yellow rat snakes (*E. o. o.* x *E. o. quadrivittata*) occurs in the Carolinas as four dark stripes on a ground of dark olive-gray. These intergrades showed dark brown blotches on an orange ground when they outgrew their usual juvenile coloration.

In early 1985, as the first stage of the plan to unmask melanin in *Elaphe obsoleta*, a mating took place with a known tyrosinase-deficient albino black rat snake. The bronze female laid seven eggs. After 60 days of incubation at 82°b F, all eggs hatched. Three were normally colored juvenile *Elaphe* (2.1) and four were albino with red eyes (2.2).

In normal yellow rat snakes, there is no yellow visible on hatchlings. With each shed skin, yellow gradually suffuses into the lighter ground color and eventually the blotches are lost entirely. In the yellow rat snake adult, coloration is four dark longitudinal stripes on a yellow to olive ground. In the Everglades rat snake the stripes fade and the ground is orange.

These albinos showed two distinct phenotypes. Initially one pair (1.1) was white with red eyes and the other two were white with blotches suffused with red. As they have grown, the yellow has filled in and two are essentially patternless pale yellow and two are pale yellow orange with red blotches.

As this is being written, eggs from the latter pair are incubating.

Currently there exists first generation of the cross between Baird's rat snake (*E. o. bairdi*) and a tyrosinase-positive albino black rat snake. Hopefully, this very red male will intensify the red of the natural Baird's.

Most other aberrancies have been intensified through inbreeding and outbreeding. Species with the most dramatic results thus far have included striping in the Sinaloan milk snake (*Lampropeltis triangulum sinaloae*), amelanistic corn snake, and parallel striping of the dorsum of an Eastern chain king snake (*L. getulus getulus*).

DISCUSSION

There are many questions raised by these findings. Why the association of patternless pale abdomen with dorsal striping was not seen in the example in the collection of Studio Elaphe is unclear. This animal has not bred, but shows the normal checkerboard. The difference may be due to a case of partial albinism. Bechtel's patternless striped corn was normally colored.

Patterning in the California king snake tends towards greater aberrancy in the amelanistic form. The striping, though dashed and dotted in the normally pigmented forms, behaves with an almost liquid brush-like fluidity. However, bizarre plaids have only been seen in normal phenotypes heterozygous for albinism.

The most significant development is the new mutation causing a previously unreported occurrence of tyrosinase-deficient albinism. Why did half the offspring have a pattern of red-orange blotches while the other sexual pair was essentially patternless? The hatching of the nine eggs of the former will likely clarify the issue.

In intergradation, which blocks of traits are inherited together? The breedings of the Baird's intergrades will also shed light on this. These intergrades show variable slurring of the blotches from the clearly blotched juvenile pattern towards the patternless adult of *E. o. bairdi*. Many questions remain.

LITERATURE CITED

Bechtel, H. B. and J. Mountain. 1960. Interspecific Hybridization Between Two Snakes of the Genus *Elaphe*. Copeia. 2:151-153.

Bechtel, H. B. and E. Bechtel 1962. Heredity of albinism in the corn snake *Elaphe gutatta gutatta*, demonstrated in captive breedings. Copeia. pp. 436-437.

Bechtel, H. B. and E. Bechtel. 1978. Heredity of pattern mutation in the corn snake, *Elaphe gutatta gutatta*, demonstrated in captive breedings. Copeia. pp. 719-721.

Bechtel, B. and E. Bechtel. 1981. Albinism in the snake *Elaphe obsoleta*. J. Herpetol. 15(4):397-402.

Bechtel, E. R. and T. Whitecar. 1983. Genetics of striping in the gopher snake, *Pituophis melanoleucus*. J. Herpetol. 17(4):362-370.

Dyrkacz, S. 1981. Recent Instances of Albinism in North American Amphibians and Reptiles. SSAR Herp. Circular #11., Oxford, OH.

Lerner, A. B. and T. B. Fitzpatrick. 1950. Biochemistry of melanin formation. Physiol. Review 30:91-126.

Riemer, W. J. 1958. Longitudinal striping as a pattern modification in the snake *Pituophis melanoleucus*. Chicago Acad. Sci. Publ. 165:1-9.

Slemmer, G. 1973. Breeding the white albino corn snake. Eastern Seaboard Herpetological League Newsletter.

Wagner, E. The Genetics of Pigmentation in the Corn Snake, *Elaphe gutatta*. Woodland Park Zoological Gardens, Seattle, WA. Also pers. comm.

Zweifel, R. G. 1981. Genetics of color pattern polymorphism in the California kingsnake. J. Heredity. 72:238-244.

Studio Elaphe
RD 1, Box 31
Hartwick, NY 13348

BASIC GENETICS FOR SNAKE BREEDERS

Mary Stafford

A basic knowledge of simple genetics is a useful tool in breeding the many color varieties of colubrid snakes currently available. Although almost everything I say here probably has some exceptions, I still hope to give you a basic grasp of how different genes are inherited and an understanding of how to apply this knowledge to get what you want from your breedings. Working with animals which are often not ready to breed until they are two or three years old, the herpetoculturist has a special need to choose the expeditious cross.

In 1865, Gregor Mendel, an Austrian monk, described experiments he had performed in the monastery garden with peas. He reported that some characteristics seemed to be "dominant" and others "recessive." When a plant with dominant characteristics was crossed with one with recessive ones, the resultant plants all exhibited the dominant characteristics. But when two of these second-generation plants were crossed, some of the resultant plants showed the recessive characteristics. Mendel called the three generations described here "parent," "F-1," and "F-2" generations (the "F" standing for filial), and he accurately described the 3:1 ratio in which dominant and recessive characteristics reappeared.

The hereditary transmission of such physical characteristics was understood by the 1860s, and Haeckel had identified the gametic cell nucleus as the likely source by 1868. Flemming described cell division in 1879. Mendel's important research, however, lay virtually unrecognized until the turn of the century, when deVries, Correns, and Tschermak all described similar experiments and acknowledged Mendel's contribution to genetics.

A view of genetic knowledge of the 1940s would have been this: the cells of all living things contain genes, which control the inheritance of the basic characteristics of the organism. Genes are carried on rod-like structures in the nucleus of the cell called chromosomes. In the somatic (or body) cells, chromosomes are present in pairs, one of each pair having been received from each parent. In gametic (or sexual) cells, chromosomes are present as single representatives of each pair, due to the reduction division of meiosis (Fig. 1).

The assortment of chromosomes in any individual gamete is random and contains chromosomes from both parents, but only one chromosome of each pair. The number of pairs of chromosomes varies from species to species. Fertilization is the union of two gametes, and their genetic material combines to produce the somatic cells of the new organism.

In 1953, the science of genetics took a quantum leap with the publication in "Nature" of a short paper by James Watson and Francis Crick, which was modestly called "Genetical Implications of the Structure of DNA." The deoxyribonucleic acid (DNA) molecule was known to be the material of which chromosomes are made. Watson and Crick described a double helix with a backbone of polysaccharides and rungs of joined nucleotides. Four nucleotides were present and they only joined with specific partners: adenine always linked to thymine, and guanine always linked to cytosine.

Watson and Crick's work described in biochemical detail an orderly system which had the means to replicate itself accurately. In the years since 1953, biochemical genetics has progressed so far as to be able to identify the exact nucleotide sequences of some simple genes.

You don't need to know the exact nucleotide sequence of a gene, though, to be a successful breeder. You only need to know what happens when two animals with different characteristics are bred together. Let's backtrack a little and set out some definitions.

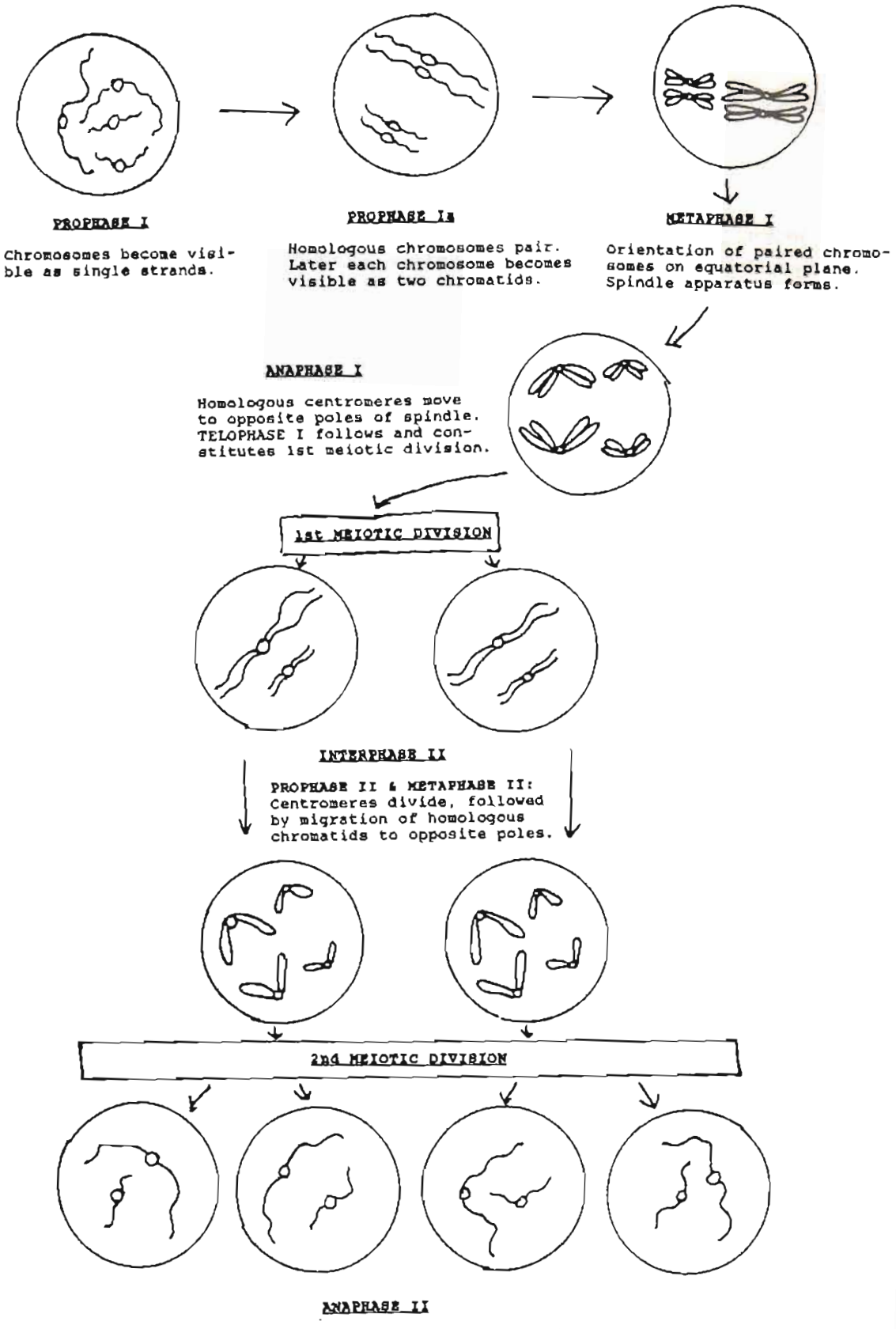


Figure 1: Meiosis

We've already talked about dominant and recessive genes. Actually they should be called alleles -- alleles are different forms of the same gene. Genes come in pairs, one inherited from each parent. In order for an animal to exhibit the recessive characteristic, it must have two copies of the recessive allele. An animal exhibiting the dominant characteristic can have either one or two copies of the dominant allele, for a recessive allele will be "covered" by a single dominant one.

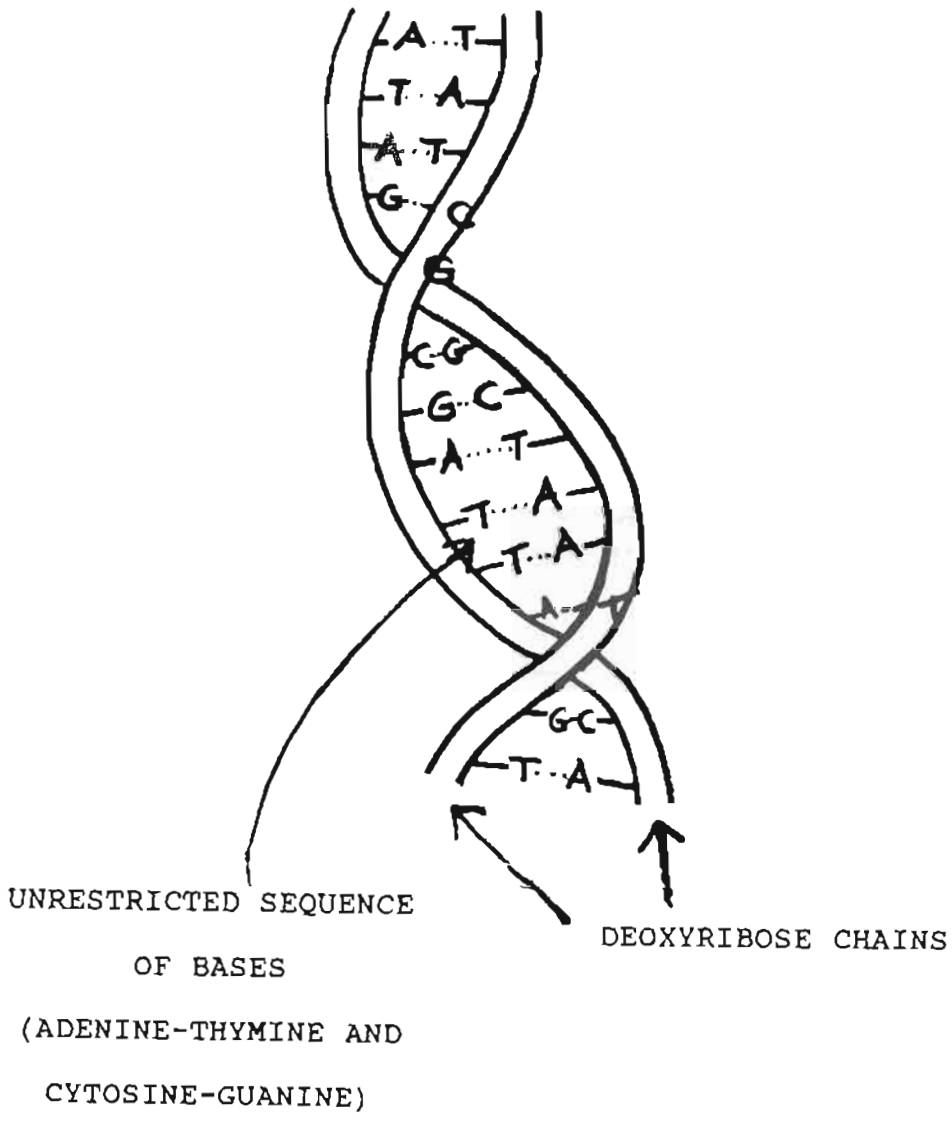


Figure 2. Diagrammatic Representation of a DNA Chain

When an animal has alleles of only one sort, either of the dominant or the recessive form of the gene, it is called "homozygous," or homozygote; if it has one dominant and one recessive allele, it is called "heterozygous," or a heterozygote. When the dominant allele is wholly dominant, as many are, there is no way to tell by visual inspection whether an animal is a "homozygous dominant" or a heterozygote. When the dominant allele is only partially dominant, a heterozygous organism may have a distinctive and different appearance from either dominant or recessive homozygotes.

What you can tell about an animal's genetic make-up by visual inspection is called its phenotype. If it exhibits the dominant allele's effect, you know it must have at least one dominant allele. If it exhibits the recessive allele's effect, you know it must have two recessive alleles. But phenotype hides a lot of information you need to make good breeding choices. What you really want is to know the genotype -- the actual genetic make-up -- of each of your animals. You want to know what nice little surprise packages of recessive genes your animal is hiding in its gametic cells.

How do you learn what an animal's genotype is? The answer is by keeping careful records and by making initial breedings that will reveal possible recessive genes. Once you have established the genotypes of your breeding stock, you should be able to plan breedings that will give you the types of animal you want, with no rude surprises. Let's look at some types of crosses and what the probability of the offspring would be.

A standard tool of the geneticist is the Punnett square. It is a quick way to look at all the possible genotypes likely to result from the recombination of the ganetic cells of two potential breeding animals and what the probable numbers of offspring of each type will be. A Punnett square provides you with an easy way to write down what you know about your stock and project the probability of getting the variants in offspring. The usual convention is to choose a single letter related to the characteristic being considered and use an uppercase letter for the dominant form and a lower case one for the recessive. In the first examples: D = a dominant gene, d = its recessive allele.

If you breed an animal that is a homozygous recessive to an animal that is a homozygous dominant, you have done the same experiment that Mendel did and your F-1 animals are all heterozygotes. They have inherited one allele from each parent.

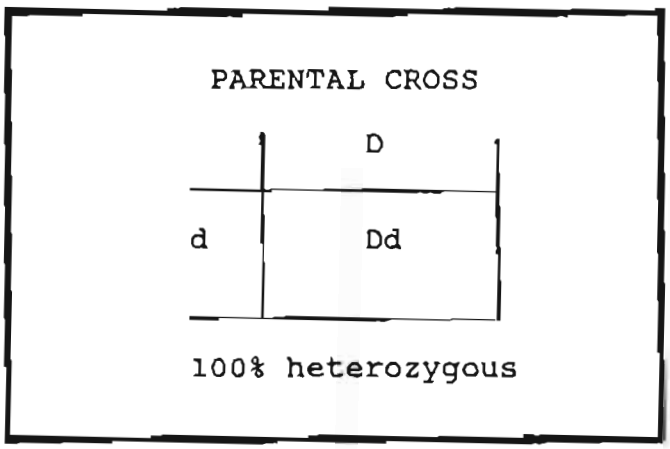


Figure 3. A 100% Heterozygous Parental Cross

If you breed a pair of these heterozygotes together, you will get (more or less) a ratio of three normal animals to one of the recessive type. Actually, the apparent 3:1 ratio is a 1:2:1 ratio -- one homozygous dominant, two heterozygotes, and one homozygous recessive. The difficulty with this type of breeding is that you will have to wait two or three years before you can breed the normal-colored animals to a recessive one

(assuming you've succeeded in getting one and it is of the opposite sex!) in order to find out which carry the recessive allele. Some breeders sell animals produced from such crosses as "2:1 probability heterozygotes" -- talk about a pig in a poke!

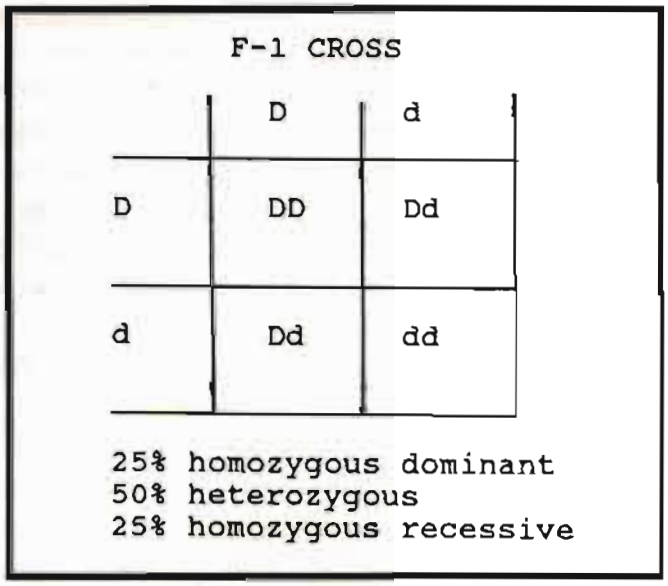


Figure 4. An F-1 Cross

If you don't have homozygous recessive stock, you may be forced to make such crosses until you produce some. This may especially be true if you are establishing stock of a new mutation. But if you have access to the "pure" recessive stock, a backcross is always more useful for a known heterozygote.

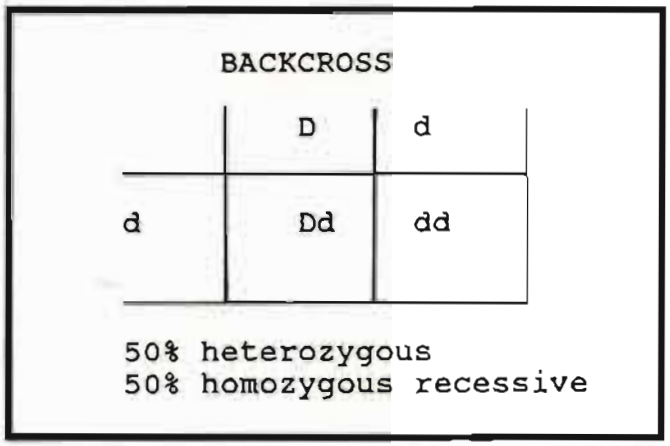


Figure 5. An F-2 Cross

A note of caution: the ratios given here apply best to large numbers. In the small numbers of a single clutch of eggs, you may not see such neat ratios, although a backcross, with its 1:1 ratio for heterozygote to homozygote, will usually give you proof of the hidden recessive allele. Nevertheless, it is possible to make a cross that shows nothing, because all the offspring are of the dominant phenotype.

Don't despair, but do the same cross again to get better numbers. And never forget that the luck of the draw can run in the opposite direction and give you a clutch of animals all of the recessive phenotype.

Color in reptiles is more complex than in mammals. There are at least four common types of chromatophores -- melanophores (black and brown pigment), erythrophores (red pigment), xanthophores (yellow pigment), and iridophores (don't produce pigment, but cause reflectivity and iridescence). In mammals, melanophores are the only common chromatophore.

Mutations can occur which affect the production of any of these chromatophores. A mutation is a change in the nucleotide chain of a gene which alters it, but still allows the animal to function. The most common pigment mutations are those called "albinos" – though in reptiles this is a misnomer, as usually only one color's production is being blocked. Thus, animals exist which cannot produce melanin -- the so-called "red albino" corn snake, and most of the other so-called albino snakes such as gophers, pythons, and black snakes. These animals produce levels of erythrin and xanthin normal for their individual coloration - but no melanin. "Amelanistic" is a more accurate term. Similarly, the so-called "black albino" corn snakes are really anerythristic. "Snow" corn snakes are NOT true albinos, but they represent a phenomenon you need to understand in breeding reptiles: they are "double recessive homozygotes," missing both melanin and erythrin. It should be noted here that more than one mutation may occur which affects the production of a pigment. It is possible to breed together two amelanistic animals and to produce only normally colored offspring. This has, in fact, occurred in some lines of amelanistic blacksnakes; so far as I know, amelanistic corn snakes seem to derive from a single common mutation.

Let's look at some crosses with animals carrying mutations in color production. In the following illustrations: M = melanin; m = no melanin; E = erythrin; e = no erythrin; X = xanthine; x = no xanthine, though, in fact, I know of no animals yet documented as axanthic.

An amelanistic animal backcrossed to an anerythristic/amelanistic animal will produce either animals uniformly like itself or animals like both parents, in about equal proportions. Similarly an anerythristic animal backcrossed to the same anerythristic/amelanistic animal will produce either all anerythristic animals or an even number of anerythristic and anerythristic/amelanistic ones.

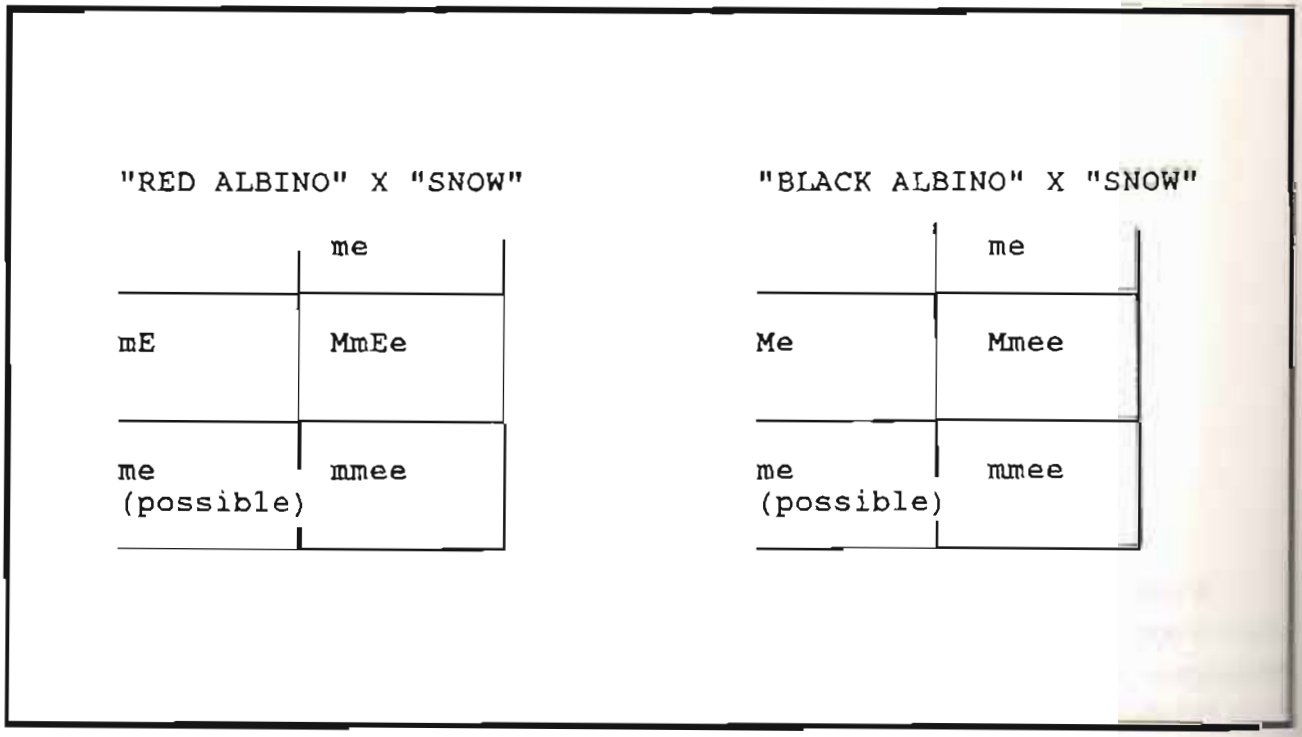


Figure 6. A Two-Gene Backcross.

The point of such backcrosses is simple. In choosing as one parent an animal whose recessive genotype is known, you have a fixed input against which to examine the genes of the other parent by observing the offspring.

To see why this is useful, look at the Punnett square necessary to plot a cross between F-1 hybrids of a two-gene cross such as the corn snakes we've been looking at (Fig. 7).

	ME	Me	mE	me
ME	$MMEE_1$	$MMEe_1$	$MmEE_1$	$MmEe_1$
Me	$MMEe_1$	$MMee_2$	$MmEe_1$	$Mmee_2$
mE	$MmEE_1$	$MmEe_1$	$mmEE_3$	$mmEe_3$
me	$MmEe_1$	$Mmee_2$	$mmEe_3$	$mmee_4$

1: phenotype "normal"
2: phenotype "red albino"
3: phenotype "black albino"
4: phenotype "snow"

Figure 7. A Two-Gene F-1 Cross.

Notice all the animals which will be phenotypically "normal," but are heterozygotes of one type or another. And if this isn't bad enough, let's propose a cross of F-1 hybrids of a 1-gene cross.

To sum it up, then: Once you understand where the genes are coming from and can draw a Punnett square to work with, you're in business. Keep careful records of your animals, including all you know about their parent's phenotype, plan your first breedings to give you as much genotype information as you can, and repeat crosses if necessary to gather sufficient information. What this sort of breeding program will give you is the ability to breed the colors you want and to sell your animals to others with good information on their genetic background.

	MEX	MEx	mEX	MeX	meX	Mex	mEx	mex
MEX	MMEEXX 1	MMEEXx 1	MmEEXX 1	MMEeXX 1	MmEeXX 1	MMEeXx 1	MmEEXx 1	MmEeXx 1
MEx	MMEEXx 1	MMEExx 4	MmEeXx 1	MMEeXx 1	MmEeXx 1	MMEexx 4	MmEExx 4	MmEexx 4
mEX	MmEEXX 1	MmEEXx 1	mmEEXX 2	MmEeXX 1	mmEeXX 2	MmEeXx 1	mmEEXx 2	mmEeXx 2
MeX	MMEeXX 1	MMEeXx 1	MmEeXX 1	MMeeXX 3	MmeeXX 3	MMeeXx 3	MmEeXx 1	MmeeXx 3
meX	MmEeXX 1	MmEeXx 1	mmEeXX 2	MmeeXX 3	mmeeXX 5	MmeeXx 3	mmEeXx 2	mmeeXx 5
Mex	MMEeXx 1	MMEexx 4	MmEeXx 1	MMeeXx 3	MmeeXx 3	MMeexx 6	MmEexx 4	Mmeexx 6
mEx	MmEEXx 1	MmEExx 4	mmEEXx 2	MmEeXx 1	mmEeXx 2	MmEexx 4	mmEExx 7	mmEexx 7
mex	MmEeXx 1	MmEexx 4	mmEeXx 2	MmeeXx 3	mmeeXx 5	Mmeexx 6	mmEexx 7	mmeexx 8

1: phenotype "normal"
2: phenotype "red albino" (has red and yellow)
3: phenotype "black albino" (has black and yellow)
4: phenotype "funny albino" (has red and black)
5: phenotype "snow" (has only yellow)
6: phenotype "unknown A" (has only black)
7: phenotype "unknown B" (has only red)
8: phenotype "colorless" (has none of three pigments)

Figure 8. A Three-Gene F-1 Cross.

24 Wadsworth Street
Allston, MA 02134

A CAPTIVE BREEDING PROGRAM OF THE CORN SNAKE, Elaphe guttata

Robert T. Zappalorti[1] and Otto Heck[2]

INTRODUCTION

In 1976, the Endangered and Nongame Species Program, Division of fish, Game, and Wildlife, New Jersey Department of Environmental Protection (NJDEP) classified the corn snake, *Elaphe guttata*, as a "threatened" species in New Jersey. In the spring of 1977, Herpetological Associates, Inc. (HA) was commissioned by NJDEP to study the ecology and distribution of *E. guttata* in southern New Jersey.

HA carried out distributional studies throughout the historic range in an attempt to reconfirm museum locations in Atlantic, Burlington, Cumberland, and Ocean Counties. Field surveys in these four counties throughout the duration of the study (1977 through 1987) failed to reveal corn snakes in Atlantic and Cumberland counties (Zappalorti and Johnson 1980; Zappalorti and Barber 1987). We did, however, confirm breeding populations in both Burlington and Ocean Counties. The historic location, in Burlington County, is the largest population, based on personal experience and available locality records (Zappalorti and Johnson 1982b). Although the land is relatively protected because it is within the State forest area, in recent years, the population has been reduced from its former numbers by illegal collecting. In contrast, the historic population in Ocean County has been reduced by serious loss of habitat from housing developments and illegal collecting by hobbyists and commercial animal dealers.

HA initiated life history and mark-recapture studies at both areas in the spring of 1977 and continued the investigation to the present date. Observations were made on mating, nesting, clutch size, incubation period, hatchling size, sex ratio, age class sizes, activity range, habitat utilization and behavior in the wild (Zappalorti and Johnson 1978; Zappalorti 1979; Zappalorti and Merli 1980; Zappalorti and Johnson 1982b; and Heck and Zappalorti 1985). However, recapture of marked individuals was rare because of their fossorial habits. Based on data gathered by HA between 1977 and 1982 (Zappalorti and Johnson 1982b), the NJDEP officially changed the status of the corn snake "threatened" to "endangered" on January 17, 1984. Subsequently, under contract from NJDEP, HA initiated a captive breeding program in 1982 to produce hatchlings, which were marked and released to help mitigate losses from the population (Dodd 1987).

PERSPECTIVE AND SCOPE

The corn snake is sometimes referred to as the "red rat snake" because of its bright red, orange or coral-red blotched pattern. This beautifully colored snake adapts well to captivity and has been known to live up to 21 years in zoos. Since they are able to adjust so well in the laboratory, and with proper care have been known to breed in captivity, HA recommended that an experimental captive breeding program be initiated in the spring of 1982 (Zappalorti and Johnson 1981).

The objectives of our two-part breeding program were:

1. To breed corn snakes in the laboratory and release the marked hatchlings on NJDEP wildlife sanctuaries that are managed and protected (Dodd 1987).
2. To capture gravid females in the wild, maintain them in the laboratory until their eggs are deposited, mark and release the females at their exact point of capture, hatch the eggs in the laboratory, sex, and mark the hatchlings, and release them at protected sanctuaries.

The success of a breeding program depends on the healthy condition of the snakes, and more important, the knowledge of the keeper implementing the program. One needs an understanding of the annual activity cycles in order to provide the conditions which will help with the inducement of courtship, mating, and deposition of fertile eggs. It is important to select healthy specimens for the breeding stock. Additionally, one must have the knowledge to accurately sex the adults. We used two techniques for sexing snakes: adults were sexed by probing (Schaffer 1934; Fitch 1960), and hatchlings were sexed by the hemipenis-popping method (Gregory 1983).

Specific Methods

In anticipation of the implementation of a captive breeding program of the New Jersey corn snake, the authors selected adult snakes and kept them in the laboratory (Riches 1976). HA staff captured 2.2 snakes in Burlington County (2 males and 2 females), and in Ocean County. A specimen was kept until it shed its skin, and if it did not eat after this, it was marked and released where it was originally captured (one male from Ocean County had to be released). The snakes that accepted food (laboratory mice) were housed in 20 gallon glass aquaria or sturdy, glass fronted wooden cages (91 cm in length by 30 cm in height and 30 cm in width). the tops of the aquaria were covered with screen and secured with clamps; the wooden cages were secured with locks to prevent escape. The males were kept separately from the females with one or two specimens to a cage. The snakes were usually placed in hibernation around November 15 (for a thorough description of snake husbandry, see Kauffeld 19679, Riches 1976, and Tryon 1985).

Hibernation in the Laboratory

Each snake was kept individually in a 20-30 gallon aquarium at HA headquarters in Ocean County, New Jersey and/or Trenton State College. The basement in HA's office has a "cold room" that is unheated. For the purpose of "photo-period," all cages were faced towards the four windows in the "cold room," which have a southerly exposure so that the snakes could experience natural daily light cycles. Since there was no heat source near the cages, the air temperature dropped gradually in the cages in correlation with the outside temperature (Tryon 1985). It should be noted that the temperature where the cages were (along the inside wall) was 10 to 15 degrees warmer than the inside temperature next to the windows. Each cage was set up as follows:

1. The substrate consisted of 10 to 20 cm of Pine Barrens sand
2. A hollow log was placed on the floor of the cage.
3. A medium of sphagnum moss, pine chips, and/or pine bark was placed over the log and sand at a depth up to 25 cm.
4. A water dish was provided in each cage so the snakes could drink if they chose to.

Each cage was checked once a week and kept damp, not wet. The air temperature in the cages ranged from 10 to 18°C in November and December; 7.5 to 12.5°C in January; 5.0 to 10.0°C in February; and 7.5 to 12.5°C in March. The snakes were removed from the "cold room" about 15 April and placed in the laboratory. During hibernation, the cages were provided with a constant supply of drinking water in order to prevent dehydration of the snakes.

Mating Snakes

Temperatures in the laboratory were kept between 20.0 and 28.5°C. After snakes were set up in their cage, they were offered food. Each snake was fed 2-3 mice per week. Once the snakes shed their skins, the pairs were placed together. The male was placed in the female's cage during May. The snakes were checked on an hourly basis in order to witness copulation. The following data were recorded for one of the pairs: May 10, 1983 1330 hrs: The male began courtship behavior; 1400 hrs: The female was receptive to the courtship behavior; 1415 hrs: Copulation was observed; 1435 hrs: The pair separated and the male was removed from the cage.

If there was no interest shown by the female initially, the male was left in her cage up to 3-4 days. When copulation was observed, the male was placed back in its cage for 24 hours, and then placed with another female in an attempt to have all females in the group fertilized. Gravid snakes were fed 2-3 mice a week for as long as they would accept food. Females usually go off food for a few weeks prior to egg laying (Riches 1976).

Care of Eggs

The gestation period is about 35-45 days. The female cited above was mated on 18 May and began probing behavior or "nest searching" on 20 June, at which time a "nest container" was placed in her cage (i.e., a plastic shoe box filled with damp sphagnum moss). Additionally, the water bowl was removed to prevent the female from depositing her eggs into it.

The eggs were deposited on 4 July at 2100 hrs into the "nesting container." The eggs were removed and placed in a one gallon, wide-mouthed glass jar, half filled with Pine Barrens sand and damp sphagnum moss. The eggs were then covered with moss, the jar being filled to about one inch from the top. A ventilation hole was drilled in the lid of the jar to allow air exchange. The lid also kept humidity high during the incubation period, thereby keeping the eggs damp, but not wet. The eggs were kept at cycling temperatures ranging from 20.0 to 30.0°C, never higher or lower (Tryon 1975 and Riches 1976).

RESULTS

Between 1982 and 1986, this program has resulted in the registration and release of 135 captive bred corn snakes in Burlington and Ocean Counties, New Jersey (Tables 1 and 2).

Results of Feeding Tests

Small mammals that were captured in pitfall and/or funnel traps were removed and taken to the laboratory. The adult corn snakes in our breeding colony were used to conduct tests to determine prey acceptance. Twenty-two corn snakes were used (12 adults, 5 juveniles, and 5 hatchlings) to test acceptance of eight small mammal species. Mammal identification was made by Dr. Richard Van Gelder of the American Museum of Natural History in New York City (pers. comm. 1985).

Each corn snake was kept separately in a glass container and/or wooden cage (80 cm long x 30.5 cm high x 30.5 cm wide). The floor of the cage was covered with 5-10 cm of sandy soil and pine needles and a hollow log was provided as a hiding place. We tried to provide semi-natural conditions in each cage.

A live and/or dead animal was quietly introduced into a cage. Prior to constriction, most snakes would demonstrate rapid tongue flicking and attack. Small rodents were always accepted by corn snakes that were not opaque. Opaque individuals would usually reject prey items. They would resume eating after casting their skin. Larger-sized rodents, such as small cottontails, gray squirrels, and red squirrels, were always rejected. Shrews were accepted by large snakes, but juveniles would only occasionally eat them if they were dead.

Table 1. Combined Number Hatchling Corn Snakes Produced and Released From Captive Breeding Program

YEAR BRED	NUMBER OF MALES	NUMBER OF FEMALES	TOTAL	SEX RATIO
1982	10	9	19	1.1:1
1983	13	12	25	1.1:1
1984	21	16	37	1.3:1
1985	24	24	48	1:1
1986	3	3	6	1:1
TOTALS: 5 Years	71	64	N=135	1.1:1

NOTE: Males represent 53% of captive bred corn snakes, whereas 47% were females (100%).

Adults from both Burlington and Ocean Counties were used as breeding stock during 1984-1985. We were successful in mating 10 of the 12 females held in captivity. Of these, a total of 75 eggs were deposited. Table 2 presents a breakdown of these data.

Since the initiation of this captive breeding program in the spring of 1982, our efforts have produced a total of 129 captive bred corn snakes that were released back into the wild. A breakdown of the sex ratio and number of snakes by year is presented above in Table 1.

Table 2. Number of Clutches Produced from Captive Bred Corn Snakes in 1985

CLUTCH NUMBER	NO. OF EGGS IN CLUTCH	NO. OF EGGS THAT HATCHED	NO. OF MALES	NO. OF FEMALES	SEX RATIO	AREA WHERE HATCHLINGS RELEASED
*85.01	8	6	3	3	1:1	Ocean County
*85.02	6	4	2	2	1:1	Ocean County
*85.03	11	11	6	5	1.2:1	Ocean County
*85.04	6	6	4	2	2:1	Ocean County
*85.05	10	0	0	0	--	-- None --
**85.06	10	5	1	4	.25:1	Burlington Co.
**85.07	4	0	0	0	--	-- None --
**85.08	9	6	4	2	2:1	Burlington Co.
**85.09	5	4	2	2	1:1	Burlington Co.
**85.10	6	6	2	4	.5:1	Burlington Co.
TOTALS	75	48	24	24	1:1	(50% x 50% Sex Ratio)

NOTE: * = Adults from Ocean County, New Jersey stock

** = Adults from Burlington County, New Jersey stock

Source: HA records from captive breeding program, unpublished data.

Table 3. Small Mammals Trapped at Drift Fence in Ocean County, New Jersey

Common Name	Scientific Name	May	Jun	Jul	Aug	Sep	Oct	Total	Percent By Species	Known in Corn Snakes Diet
**Virginia Oppossum	Didelphis virginiana	0	1	0	0	1	0	2	1.8%	NO
*Masked Shrew	Sorex cinereus	5	2	2	12	9	8	38	32.4%	YES
**Short-tailed Shrew	Blarina brevicauda	1	3	1	1	2	1	9	7.6%	NO
**Long-tailed Weasel	Mustela frenata	0	0	0	0	0	1	1	0.9%	?
*Eastern Mole	Scalopus aquatieus	1	0	1	0	1	0	3	2.6%	NO
*Eastern Cottontail	Sylovilagus floridanus	1	0	2	1	0	0	4	3.5%	NO
*Gray Squirrel	Sciurus carolinensis	0	1	0	0	1	0	2	1.8%	?
*Red Squirrel	Tamiasciurus hudsonieus	1	0	1	1	0	0	3	2.6%	?
*White-footed Mouse	Peromyscus leucopus	3	4	4	7	6	5	29	24.7%	YES
*Red-backed Vole	Clethrionomys gapperi	2	3	1	1	3	0	9	7.6%	YES
*House Mouse	Mus domesticus	0	2	0	2	1	1	6	5.2%	YES
*Jumping Mouse	Zapus hudsonius	0	1	1	1	1	0	3	2.6%	YES
*Woodland Vole	Pitymys pinetorum	1	1	3	2	1	0	8	6.8%	YES
TOTALS:	13 Species	15	17	15	28	26	16	117	100%	7 Confirmed
% BY MONTH:		12.9%	14.5%	12.9%	23.9%	22.2%	13.6%	=	100%	3 Unknown

NOTE: * = Varlous small mammals were offered, both live and dead, to cornsnakes in the laboratory during this investigation. A YES means the snakes ate the particular species on several occasions.

** = Potential predators to corn snake eggs, hatchlings, and/or adults under natural conditions

Northern fence lizards were always accepted in the tests, but there was a definite correlation between prey size and snake size. Hatchlings and juvenile corn snakes would attack and constrict small fence lizards and mice, but would retreat from large adult lizards or rodents. This was also true of adult corn snakes.

Large mammals would elicit defensive behavior (e.g., an "S" coil accompanied by a hiss and strike) and/or retreat behavior. Medium-sized rodents and shrews would elicit attack, constriction, and acceptance of the prey item. Our observations suggest that corn snakes selected prey on the basis of size and catchability. There have been other studies on feeding behavior of snakes, both in the laboratory and in the wild. Most notable are Reynolds and Scott (1982) and Reinert et al. (1984). See Table 3 for a breakdown of small mammals eaten during our experimental feeding tests.

SUMMARY

A total of 16 adult *Elaphe guttata* were kept in the laboratory as part of a captive breeding program initiated in 1982. Snakes were artificially hibernated in "cold rooms" (eight at Trenton State College and eight at Herpetological Associates' laboratory) at temperatures from 10 to 15°C between November, 1984 and March, 1985. Of the 12 females, 10 were successfully mated in the spring of 1986 and subsequently deposited 76 eggs. These were incubated and 48 successfully hatched (64%). All hatchlings were sexed and permanently marked. The sex ratio of the 48 snakes was exactly 1:1.

Marked hatchlings were released in Burlington and Ocean Counties (21 from Burlington and 27 from Ocean).[1] An intensive sampling regime was initiated in the Ocean County study area to recapture captive-bred *Elaphe guttata*. Information on movements and growth rate from 6 (7.4%) of the original 81 snakes was obtained (3 from 1983 and 3 from 1984). During 1985, 14 *Elaphe guttata* were found in Ocean County of which 6 (43% of the total caught) were captive-bred snakes. Population size was estimated at 189 snakes by use of the Lincoln Index. Range limits of the population size (95%) were 72-306 individuals.

Herptiles and mammals were also investigated by use of a 6 month drift fence trapping program near the release site. We captured 13 species of mammals, of which 7 were shown to be prey species of *Elaphe guttata* through laboratory tests. *Sorex cinereus* (32%) and *Peromyscus leucopus* (25%) were the most common mammalian resources at the study site. *Sceloporus undulatus hyacinthinus* were the most abundant reptile trapped in the drift fence (49%). There is a correlation between the known food items in the diet of *Elaphe guttata* and the prey species abundance in the drift fence. Feeding tests suggest prey items are consumed in similar proportion to their abundance at the Ocean County study site.

LITERATURE CITED

Dodd, C. K., Jr. 1987. Snakes: Ecology and Evolutionary Biology Chapter 15; Edited by R. Seigel, J. Collins, and S. Novak. Macmillan Publishing Co., New York, NY p. 478-521.

Fitch, H. S. 1960. Criteria for Determining Sex and Breeding Maturity in Snakes. Herpetologica, No. 16, pp. 49-51.

Gregory, P. T. 1983. Identification of Sex of Small Snakes in the Field. Herp. Review, Vol. 14, No. 2, pp. 42-43.

Heck, O. and R. T. Zappalorti. 1985. A Final Report on a Captive Breeding Program of the Corn Snake, *Elaphe guttata*, and Notes on a Sampling Program of Released Hatchlings. Unpublished report submitted to the NJDEP, HA File No. 85.21, pp. 1-41.

Kauffeld, C. F. 1957. Snakes and Snake Hunting. Hanover House, Garden City, New York, pp. 36-55.

Reinert, H. K., D. Cundall, and L.J. Bushar. 1984. Foraging behavior of the timber rattlesnake *Crotalus horridus*. Copeia 1984:976-981.

1 Experimental method and results of this phase of the study are available from Herpetological Associates, Inc.

[illegible], R. J. 1976. Breeding Snakes in Captivity. Pet Reference Series, #3. Arlo Publishing Co., NY

[illegible], R. J. 1980. "Double-Littering" in *Thamnophis sirtalis sirtalis*. Herptile 5(4):12-13.

[illegible], R. P. and N. J. Scott, Jr. 1982. Use of Mammalian Resource by a Chihuahuan Snake Community, in Herpetological Communities, Edited by N. J. Scott, Jr., Fish and Wildlife Service Report No. 13, p. 99-118.

[illegible], W. H. 1934. Diagnosis of Sex in Snakes. Copeia 1934: 181.

[illegible], B. W. 1975. How to Incubate Reptile Eggs: A Proven Technique. Bull. New York Herp. Soc. 11:33-37.

[illegible], B. W. 1985. Snake Hibernation and Breeding: In and Out of the Zoo. Dept. of Herpetology, Knoxville Zoological Park, Knoxville, TN.

[illegible], R. T. 1979. The Ecology of the Corn Snake, *Elaphe guttata guttata*, in Southern New Jersey, Part II. Unpublished Progress Report submitted to the NJDEP. Vol. 7, p. 1-32.

[illegible], R. T. and E. W. Johnson. 1978. The Ecology and Distribution of the Corn Snake, *Elaphe guttata guttata*, in New Jersey, Part I. Unpublished Progress Report submitted to the NJDEP, Vol. 4, p. 1-36.

[illegible], R. T. and J. Merli. 1980. The Ecology of the Corn Snake, *Elaphe guttata guttata*, Linnaeus, in Southern New Jersey, with Special Notes on Habitat Description, Part III. Unpublished report submitted to the NJDEP. Vol. 80.05, p. 1-11.

[illegible], R. T. and E. W. Johnson. 1982a. Critical Habitat Requirements of Two Threatened Species in the Davenport Basin, Ocean County, New Jersey. Unpublished report submitted to the Ocean County Planning Board, HA File No. 82.04, pp. 1-22.

[illegible], R. T. and E. W. Johnson. 1982b. Progress Report on Herpetological Research in 1982 on Endangered and Threatened Herptiles in New Jersey. Unpublished report submitted to the NJDEP, HA File No. 82.07, pp. 15-18.

[illegible], R. T. and R. D. Barber. 1987. Mammalogical and Herpetological Studies in the Manumuskin River Drainage Basin in Cumberland and Atlantic Counties, New Jersey between 1986 and 1987. Unpublished report submitted to the Department of Planning and Development, County of Cumberland, Bridgeton, New Jersey. HA File No. 87.01-C:1-58.

[1]Herpetological Associates, Inc.
1018 Berkeley Avenue
Beachwood, NJ 08722
and
[2]Department of Biology
Trenton State College
Trenton, NJ 08625

OROPHARYNGEAL FLORA IN BOIDS: LONGTERM FOLLOW-UP

Megan Hilf, M.S., Robert A. Wagner, V.M.D., Victor L. Yu, M.D.

Bacterial pneumonia causes considerable mortality and morbidity in captive reptile collections. At the Pittsburgh Zoo, although the numbers have declined due to improvement of housing conditions, approximately 3-5 snakes per year are confirmed on necropsy to have had pneumonia both by histological exam and by cultures taken from the lung.

In reptiles it is often difficult to determine with certainty whether bacterial pneumonia actually represents primary disease or whether it is secondary to other conditions such as stomatitis, viral infection, changes in the environment, or any other forms of stress. Nevertheless, more information concerning bacteriologic flora and its involvement in the disease process must be acquired in order to better facilitate treatment and management of pneumonia.

Studies have been done to examine the flora in association with various diseases. The organisms most often associated with pneumonia included enteric Gram negatives such as *Aeromonas, Pseudomonas, Klebsiella*, and *Salmonella*. Coagulase-negative *Staphylococcus* is often isolated although its pathogenicity in pneumonia is questionable.

METHODS

In order to assess the relationship of bacterial flora and the etiologies of pneumonia, we conducted a survey of laryngeal cultures during and following episodes of pneumonia in 1985-1986. The protocol for this study was simply to take cultures from the glottal slit of reptiles at the first sign of respiratory distress.

Respiratory distress was defined as oral/nasal bubbles, open mouth breathing, and pneumonic sounds in the lung. If the reptile survived, a one week post-therapy and one month post-therapy culture was taken. From this study we anticipated trends in bacterial flora in diseased reptiles as compared to their flora after recovery. Numerous Gram negative organisms were recovered from the initial cultures, making it difficult to know which, if any, were pathogenic and involved in the disease process. More information about normal flora prior to disease was needed. However, we did find that organisms recovered from the lung at post mortem had been isolated from the primary culture of the glottis before treatment.

From these preliminary results, we designed a prospective bacteriologic survey of the laryngeal flora of snakes from the Boidae family. Our objective was to determine the components of bacterial flora within the larynx of healthy snakes. In addition, we had a special objective: In the event that a snake developed disease during the course of the study, we would be able to compare the laryngeal flora in healthy snakes to that of snakes with pneumonia. Our hypothesis was that the bacterial pathogens involved in pneumonia in snakes will be those found in the "normal" laryngeal flora.

Culturing of the glottal slit was done bi-monthly. We wanted to examine the flora every two weeks when possible, but mating, recent consumption of food, and other factors interfered with this culturing schedule. The maximum duration between cultures was never more than four weeks. Fifty percent of the snakes were cultured at least 20 times during the one year period.

Eight boid snakes were chosen: three Burmese pythons, two reticulated pythons, and three boa constrictors. These snakes were chosen due to their predilection for pneumonia.

Cultures were plated on blood agar (non-differential media), Columbia CNA (Gram positive isolation), and MacConkey (for isolation of enteric Gram negatives). Plates were incubated under 5% CO_2 at 35°C and were read for growth at 24 and 72 hours. In addition to plating for aerobic organisms, the first 12 cultures taken were set up for recovery of anaerobic organisms. Final identification was done with the API 20E Systems (Analytab Products).

In addition to identifying the organisms we also quantified the amount of recovery of each organism. Quantification was done by the following: Very Rare = 10 colonies or less, Rare = 11 to 100 colonies in the primary streak, Light = greater than 100 colonies in primary streak, Moderate = greater than 100 colonies in secondary streak, and Heavy = greater than 100 colonies in the tertiary streak.

We used two methods for the actual culturing of the snake: laryngeal swab and tracheal wash. The reason for choosing a laryngeal swab rather than a lateral or medial swab of the oral cavity was based on the location of the glottis. Since the glottis is the passageway from the oral cavity to the lung, the flora here were thought to be more pertinent.

The second method was a tracheal wash. Even though we were taking precautions to avoid contacting the tissue in the oral cavity while swabbing the glottis, some contact was inevitable. In order to assess this problem, a tracheal wash was done for comparison. We found that bacteria isolated from the tracheal wash and the swabbing were essentially identical. Since swabbing was less traumatic, the tracheal wash was discontinued.

The definition of pneumonia used for this study was that which is well stated by numerous texts, in particular Frye: presence of oral/nasal bubbles, open mouth breathing, rhinitis with nasal discharge accompanied with prolonged starvation. The zoo veterinarian also auscultated the lungs for rales, wheezes, and gurgles.

RESULTS

We found that coagulase-negative *Staphylococcus* species was the most frequently isolated organism in the laryngeal flora. In addition, 16 different aerobic Gram negative bacilli were also isolated, *Providencia rettgeri* being the most common (Table 1).

Ten to fifty-five percent of the cultures taken from the eight snakes were "sterile," and in 20-85% of the cultures there was failure to recover aerobic Gram-negative bacilli (Table 2). No anaerobic organisms were isolated from the first 12 cultures taken; therefore, isolation for anerobes was discontinued.

A striking contrast was seen for the quantity of organisms isolated and the presence of pneumonia. Table 3 shows that healthy snakes had only very rare (10 colonies or less) Gram negative bacilli, while snakes with pneumonia showed light to heavy (greater than 100 colonies) Gram negative bacilli. The laryngeal flora in snakes remained fairly consistent throughout the year with low numbers of Gram negative organisms and low quantitation of organisms isolated (Table 4).

Cultures taken from the glottis at the time of pneumonia may be accurate indicators of pathogens in the lung, especially if the growth of the organism is light-to-heavy. Based on these preliminary results, it seems rational to institute antibiotic therapy based on known sensitivities of the organisms taken from the glottis. This remains to be confirmed in studies with larger numbers of snakes. Future investigators may find the methodology used here to have general applications in approaching this interesting clinical problem.

Table 1. Isolation Frequency of *Staphylococcus* and *Providencia*

Snake	Isolation Frequency of *Staphylococcus*	Isolation Frequency of *Providencia*
1	100% (14/14)	57% (8/14)
2	50% (1/2)	----
3	85% (17/20)	60% (12/20)
4	70% (14/20)	55% (11/20)
5	60% (3/5)	40% (2/5)
6	45% (9/20)	----
7	45% (9/20)	5% (1/20)
8	100% (4/4)	----

Frequency = # of times isolated/# of times sampled

Table 2. Culture Results Showing Absence of Gram Negative Rods or No Growth

Snake	No GNR	Sterile
1	----	----
2	----	----
3	25% (5/20)	10% (2/20)
4	30% (6/20)	15% (3/20)
5	20% (1/5)	20% (1/5)
6	50% (10/20)	25% (5/20)
7	85% (17/20)	55% (11/20)
8	75% (3/4)	----

GNR = Gram Negative Rods

Table 3. Relative Levels of Pathogenic Organisms in Healthy vs Sick Snakes

Organism	Healthy	Pneumonia
Providencia rettgeri	Very Rare	Heavy and Light
Pseudomonas aeruginosa	Very Rare	Light
Morganella morganii	Very Rare	Light
Alcaligenes orderans	Very Rare	Moderate
Salmonella	Very Rare	Heavy, Light

Table 4. Laryngeal Flora of Two Healthy Snakes, Over an 11-Month Period

Specimen Collection Date		Small Male Boa	Reticulated Python
October	9	NG	*Providencia*, *Staphylococcus*
	27	NG	*Providencia*, *Staphylococcus*
November	6	NG	*Providencia*, *Staphylococcus*
	20	*Salmonella* sp.	*Providencia*, *Staphylococcus*
December	12	*Staphylococcus*	*Providencia*, *Staphylococcus*
	27	*Staphylococcus*	*Providencia*, *Staphylococcus*
January	13	NG	*Staphylococcus*
February	6	*Staphylococcus*	*Providencia*, *Staphylococcus*
March	3	*Staphylococcus*	*Providencia*, *Staphylococcus*
	24	*Staphylococcus*	*Staphylococcus*
April	2	NG	NG
	10	NG	*Providencia*, *Staphylococcus*
May	7	NG	*Salmonella* sp., *Staphylococcus*
	19	NG	NG
June	9	NG	*Pseudomonas* sp., *Staphylococcus*
	18	NG	*Pseudomonas* sp., *Staphylococcus*
July	24	*Staphylococcus*	*Providencia*, *Staphylococcus*
August	6	NG	*Providencia*, *Staphylococcus*
	28	*Staphylococcus*	*Providencia*, *Staphylococcus*

Staphylococcus = Coagulase-negative *Staphylococcus*
Providencia = *Providencia rettgeri*
NG = No Growth

University of Pittsburgh*
VA Medical Center and the
Pittsburgh Zoo

*Correspondence:
Dr. Victor Yu
VA Medical Center
University Drive C
Pittsburgh, PA 15240

THE DIFFERENTIAL DIAGNOSIS OF MASSES IN REPTILES

Roger J. Klingenberg, DVM

"Lumps" and "bumps" are a relatively common finding in reptiles. Captive propagation of reptiles has served to increase the economic value of many of these reptiles, justifying the expense of veterinary services. Likewise, many owners value their reptiles simply as pets and seek quality care. This paper will elaborate on the highlights of the most commonly occurring masses in reptiles.

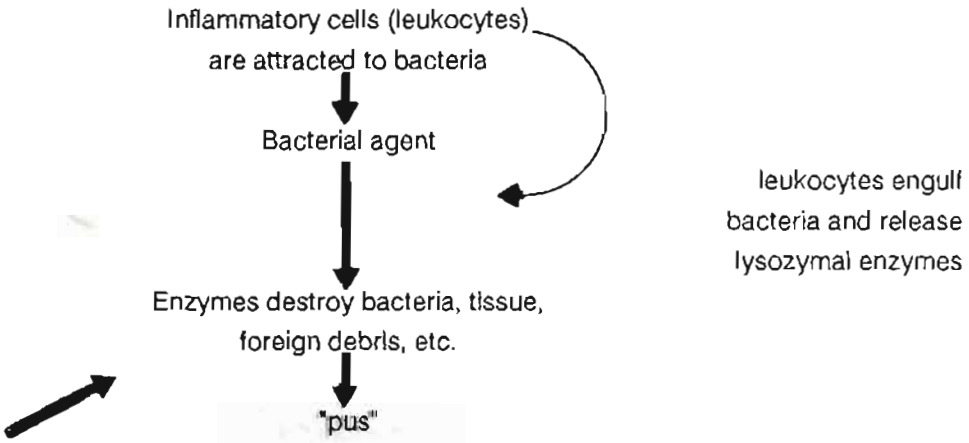
MAIN GROUPINGS OF REPTILE MASSES

A. Abscesses
B. Granulomatous masses
C. Cysts
D. Metabolic related masses
   1. Visceral gout
   2. Fibrous osteodystrophy
E. Tumors
F. Reproductive
   1. Egg binding
   2. Prolapses

A. Abscesses

An abscess can be defined as a well-circumscribed accumulation of pus, usually surrounded by a wall of fibrous tissue. As with mammals, reptiles respond to infectious bacteria by forming abscesses, which is the body's way of isolating and containing an infectious agent.

Mammalian abscesses are well liquified by the lysozymal enzymes released by the inflammatory cells attracted to the abscess. The general scheme is as follows:



Reptilian abscesses follow the same general scheme, except that due to the primitive nature of the leukocytes and the absolute or relative lack of lysozymal enzymes, the pus is caseous (cheese-like) and

inspissated (dry and hard). They may vary in size from tiny microabscesses to huge masses. Most abscesses are sub-cutaneous, but they can be deep and involve all major organs. As mentioned previously, rarely are reptile abscesses fluid-filled.

The main sources of abscesses are from direct transmission such as penetrating wounds, rodent bites, arthropod bites (mites & ticks), etc. As with most diseases, prevention is the best cure. Routine hygiene and proper caging will prevent most bacterial infections.

Another source of abscesses is metastatic spread from other sites of infection (mouth rot, pneumonia, scale rot, etc.) that spreads through the bloodstream before embedding in other tissues.

Unlike mammals, most reptile abscesses involve very tough, Gram negative bacteria. Mammalian abscesses tend to have primarily Gram positive bacteria that are less pathogenic and more amenable to treatment.

Treatment for abscesses is aimed at the direct removal of pus by excision, debridement, and drainage (flushing). Mammalian abscesses tend to have very liquid pus that is easily evacuated and drainage is easy to maintain. Reptile abscesses are characterized by caseated pus which must be forcibly scraped out or expelled. Drainage is difficult, but necessary to prevent reformation of the abscess. At my hospital, routine abscess treatment consists of:

1. Excision -- open the abscess
2. Debridement -- scrape or forcibly remove the pus
3. Pack – pack abscess with an ointment like polysporin or furacin.
   Use gauze if needed to keep the area from resealing prematurely.
4. Systemic Antibiotics – based on culture/sensitivity
5. Heat -- keeping at 85-88°F will:
   a. Increase immune response
   b. Decrease pathogenicity of some Gram negative bacteria
   c. Increase effectiveness of the antibiotics

B. **Granulomas**

Granulomas are similar to abscesses except for the following:

1. Less walling off by the host
2. Wider variety of causative agents
   a. Parasitic
   b. Fungal
   c. Bacterial
   d. Foreign bodies
3. Chronicity
   Due to chronic nature of these lesions, the associated lesions are characterized by chronic fibrotic tissues and inflammatory cells.

The treatment is similar to that of abscesses but complicated by 1) not being well located, and 2) difficulty in removal. The prognosis is dependent on the causative agent.

C. Cysts

The most common cysts are epidermal and sebaceous inclusion cysts in iguanas. The lizard tail is apparently well endowed with glandular structures similar to the sebaceous (oil) glands in mammals.

Iguana cysts are commonly misdiagnosed as abscesses, which is very understandable considering that cysts are often secondarily infected. The main difference is that most of the cysts are located on the tail, arise gradually, grow slowly, and seldom seem to affect the lizard.

The treatment of cysts involves surgical excision of the contents and inner lining of the cyst. If infected, an antibiotic ointment, and occasionally systemic antibiotics, are also required.

If a cyst is extremely large and infected, amputation of the tail is suggested. Any large mass on the tail of a lizard will cause ischemic necrosis (death due to circulatory impairment) eventually and leaving a dead tail in place seems to accelerate ischemia to other portions of the tail.

D. Metabolic

There are many metabolically induced processes that could create a mass. The two most common are fibrous osteodystrophy in lizards and visceral gout.

1. Visceral Gout

Protein breakdown is an important part of all living creatures' excretory systems. Nitrogen waste products are formed from this breakdown and must be excreted. Mammals accomplish this primarily through urea excretion in urine. Reptiles, for all practical purposes, are urecotelic (uric acid producers) with respect to their nitrogen waste removal. In a well hydrated reptile, uric acid, while poorly soluble, is cleared by the renal tubules. Uric acid is the white portion of the common feces/uric acid product passed through the common cloacal opening in reptiles.

Primary hyperuricemia is caused by abnormally high protein intake, as results from the feeding of excessive meat or organ meats. This is the most common cause of visceral gout in lizards (iguana and tegu), tortoises, and crocodiles.

Secondary hyperuricemia is the result of renal (kidney) failure or dehydration. In either case, there is a failure or inability of the kidneys to clear the poorly soluble uric acid from the plasma. This increased level of uric acid (hyperuricemia) results in the deposit of urate microcrystals into tissue, resulting in severe inflammation. Organ damage and extensive fibrosis results due to the chronic inflammation.

The treatment of visceral gout is prevention. By simply avoiding the high protein intake and balancing the diet, this disease can be prevented. Good husbandry will also help prevent the dehydration which impairs kidney clearance of uric acid. Nephrotoxic drugs which damage the kidneys' ability to clear uric acid should also be used carefully. Aminoglycoside antibiotics (Gentamycin, Tobramycin, etc.) should be used only if cultures indicate their use, and never in a severely dehydrated reptile.

Once visceral gout is established, surgical removal of affected tissues may alleviate symptoms if the involvement isn't too extensive. Some reptiles are best left alone if involvement is too extensive to lend itself to surgical correction.

2. Fibrous Osteodystrophy

Fibrous osteodystrophy results from the chronic imbalance of calcium (Ca) and phosphorus (P) in the diet of lizards and turtles. While this is a complex metabolic disease, the easiest way to

view it is to remember that calcium and phosphorus are maintained in inverse proportions in the body. If calcium is high, phosphorus is low and vice versa. Recommended levels would result in a Ca:P ratio of 1.5-1.0.

Common diets such as lean meats or lettuce may create a Ca:P ratio of 1:40 or worse. As P↑, then Ca↓. As Ca↓, this mineral is mobilized from bone to maintain blood levels. Boney tissues become so depleted that skeletal deformities such as "rubber jaw," fractures, muscle pain, and swelling result.

Treatment of fibrous osteodystrophy is to provide:

Change of Diet – It is essential to avoid all-meat diets and to eliminate non-nutritional items like lettuce from the diet. The ever-famous hamburger and lettuce diet is one of the worst diets imaginable. Hamburger is phosphorus rich and calcium poor, as well as lacking in vitamin/mineral content. Lettuce, while providing minimal Vitamin A, is worthless as a reptile nutrient.

For turtles, a corrective diet could include a mixture of protein sources (earthworms, grasshoppers, dog food, cat food, fish, etc.) with equal amounts of fruit/veggies (leafy spinach, green peas, banana, etc.). Lizards should be fed in a similar manner, providing an endless variety of fruits and veggies and as protein sources you could offer pinkies, limited crickets, waxworms, mealworms, dog food, or cat food.

Sub-adult and adult reptiles are somewhat resistant to dietary changes, so persistence is important. If a particular food group is preferred, then it can be mixed with others to facilitate the eating of those items. The dog food (or cat food) can be dry kernels that are soaked until soft, or canned products. The advantage of dog or cat foods is that they contain a lot of cereal and are fortified with essential minerals and vitamins.

Calcium Supplements – Injections of calcium gluconate twice a week for three weeks is recommended. In addition, the light sprinkling of a calcium/phosphorus/Vit. D_3 product over food is a good idea. A product such as D-Ca-Fos^R (Fort Dodge) or Pet Cal^R (Beecham) is useful in correcting deficiencies. I usually recommend an amount equivalent to the size of a match head per 8 oz. of body weight in each meal the first three weeks, and twice a week thereafter until a balanced diet has been reached.

Vitamin D -- As previously mentioned, a balanced regimen can be provided with a good supplement. Vitamin D is important in the uptake and utilization of both calcium and phosphorus. Exposure to unfiltered sunlight (for short periods to prevent overheating) or the use of Vita-lights helps the conversion of inactive Vitamin D to an active form.

Minimal Handling/Fracture Management -- Due to the fragile state of the skeletal system, a basic cage with minimal climbing materials should be provided. Avoid situations where the animal will be dropped, frightened into jumping, etc. If fractures exist already, have them checked as splinting can help provide a useful limb later.

E. Tumors

There is relatively little gathered information on reptilian tumors due to a lack of reporting. Very few people are willing to pay for histopathology even if a tumor is diagnosed.

The speculated causes for tumors are viruses primarily, with the other etiologies so poorly investigated that they cannot be considered.

The prognosis for tumors in a reptile depends on the size, location, and whether they are benign or malignant.

F. **Reproductive**

1. Egg Binding

Egg binding is a consequence of dystocia (difficulty in delivering) and is most commonly seen in turtles. It is an important entity because failure of an egg to pass can lead to "egg peritonitis" and death.

The signs of egg binding (which should be correlated with a breeding history) are straining, agitation, failure to eat, and a loss of condition. History of breeding and time interval thereafter are important to help differentiate this from common constipation.

Treatment includes housing the individual in separate confines to prevent stress and the copulatory advances of males. Warmth is important to provide a less stressful and more productive environment. Fifteen minute warm-water soaks often stimulate evacuation of fecal material as well as eggs that are only marginally caught. A dose of oxytocin (10 mg/kg) is controversial because contractions may be counterproductive in some cases. Surgical removal is recommended if an egg hasn't passed within 24-48 hours of the onset of dystocia.

2. Prolapses

Prolapses of the oviduct, hemipenes, and cloaca are common in reptiles. It is important to act promptly with these before circulatory compromise and dehydration lead to tissue necrosis (death).

Prolapses of the cloaca are most often due to parasitic infestations and constipation, both of which cause persistent straining. In early uncomplicated cases, the tissue should be gently pushed into position with a lubricated (K-Y jelly or Vaseline) Q-tip or thermometer. If the tissue prolapses again, then a retaining suture may be required. Once under control, the initiating cause must be determined and treated.

Prolapse of the hemipenes occurs with breeding, and breeding-induced trauma. Prolapse of the oviduct occurs with straining to pass eggs, constipation, infection, etc. Again, prompt attention is essential. Gentle cleansing with betadine solution, shrinking affected tissues with exposure to 50% glucose/mannitol, gentle replacement with a lubricated probe, and a retaining suture are usually required. If early, with minimal tissue damage, the prognosis is good. However, if extensive tissue damage and drying has occurred before discovery, amputation will likely be necessary.

SUMMARY

The purpose of this paper is two-fold: First, to introduce herpetologists to some of the ailments of reptiles and to a topical idea of how they are treated. This paper is very general and more detailed information can be obtained. Second, I hope to reinforce the point that good husbandry will prevent almost all of the diseases characterized by masses in reptiles. A little research into lifestyle and feeding habits will pay large dividends in the long run. Happy herping.

Sheep Draw Veterinary Hospital
6297 W. 10th Street
Greeley, CO 80634

ANOREXIA IN REPTILES

Roger J. Klingenberg, DVM

IF A HEALTHY REPTILE IN THE RIGHT ENVIRONMENT IS OFFERED THE RIGHT FOOD IN THE RIGHT MANNER AT THE RIGHT TIME, IT *WILL* EAT. RIGHT?

Now that I have your attention, let's take a look at this hypothesis. As a veterinarian who frequently treats reptiles, the question (problem) I'm most often faced with is "why won't my critter eat?" While a seven-set volume could be written on this topic, we'll limit this discussion to a brief overview of anorexia: causes and cures.

Back to our opening statement. Let's look at the variables in our hypothesis. Looking at it from another (more cynical) angle, the following requirements must be met in order for the reptile in question to eat. It must be:

1. Healthy
2. Maintained in a correct environment
3. Offered the correct food
4. Offered the food in the correct manner
5. Offered food at the proper times

Let's examine each of these requirements individually and look at all the variables.

1. **Healthy.** Poor health is the second-most-common reason, in my experience, why a reptile will refuse to eat. Many reptiles are acquired in a poor state of health and this is further complicated by the stress of a new environment. The most common states are:
   A. *Parasitism.* This includes both internal (worms, protozoans) and external (mites and ticks) parasites. This condition is especially prevalent in the "banana boat" variety of reptiles that are captured, kept together to further contaminate others, stressed, and shipped to us.

      Fecal exams should be performed on *all* new captives, not only for their own sake, but to prevent contamination of other reptiles. This can be performed by your veterinarian, or you can invest as little as $150 in a microscope (you may recoup your money on preventing one lost reptile) and learn to recognize parasite eggs. Pure recognition of eggs, not the type, is the important thing; this will be discussed further in the treatment section.

      Protozoans are more difficult to diagnose and it requires more microscope practice.
   B. *Infections.* Infectious conditions such as mouth rot, scale rot, pneumonia, etc., are commonly encountered and are often accelerated by poor husbandry techniques (to be elaborated on later).
   C. *Malnutrition/Hypovitaminosis.* Malnutrition is usually the result of being fed the wrong foods, in inadequate amounts, in a poor environment.
2. **Environment.** Proper husbandry in the form of environmental control is the *single most important* factor in getting reptiles to eat.
   A. *Heat.* Inadequate heat is a difficult problem to correct, but it is a common reason for food refusal by reptiles. The assumption made by many reptile owners is that if they keep the room temperate at 70-75°F, then the reptile should be warm enough. To test this hypothesis yourself, pick up a reptile at this temperature range and feel how warm it is (isn't, to be more exact).

      Through the years I have tried all manner of heating, and I'm convinced that for most reptiles, sub-terrarium heat as provided by heating pads or cables under the cage is the best. I prefer the

sub-terrarium heat because the reptile can cuddle up to the heat source when desired and move away if overheated. An example of a well-designed cage is the Sta-In-Pet reptile cages that provide all components in one unit (see address in back).

Heat lamps may cause burns and are difficult to regulate, but a well-designed cage for tree-dwelling reptiles could use one if properly linked to a thermostat. Plain light bulbs tied into a rheostat work well, but must be well insulated or in an area out of the reptile's reach. Use of any type lamp/bulb which produces visible light 24 hours a day is discouraged because this is a very abnormal and stressful photoperiod. Red bulbs may be used during the "dark" phase, since reptiles apparently do not see red.

Hot rocks, one of the original heating ideas for reptiles, are not used much anymore. Snakes seldom seem to the use them, probably because they have to leave their privacy for the heat. Lizards will occasionally use hot rocks. Sub-terrarium heat requires no space inside the cage, can be coordinated with privacy (housing), and does not require you to clean this heat source after a major fecal assault by the reptile.

Cable systems have allowed for a lot of creativity with heating. There are many systems, from shelf systems to heating perches, for arboreal reptiles. An address for info on one such shelf system is included at the end of this article.

Spotlights for turtles and lizards function both as heat and light sources. These lights should be considered as a separate system from other forms of heating since they are employed for only a short time period each day.

I would highly recommend that all reptile owners make the $2 investment in a thermometer to remove all guesswork from temperature control.

B. *Light.* Lighting can be a problem if there is too much or too little of it or, if it is too erratic. The advantages of a quality, limited-UV light source (like Vita-lights®) are well documented. These are:
   - o to provide for the production of active Vitamin D
   - o to stimulate appetite/activity
   - o to mimic a normal photoperiod, a necessity for normal physiological functioning and for normal changes in reproductive hormones.

   Excessive light is also well-documented as being stressful and counterproductive. Many authors have spoken of calcification of soft tissues in iguanas resulting from overproduction of Vitamin D and the resultant mobilization of calcium. I prefer the use of a Vita-light attached to a timer providing for 8 hours of light in winter, 10 hours in spring/summer, and 12-14 hours in summer.

C. *Humidity.* Reptiles from jungle environments need a higher humidity level. One way to tell if you're providing adequate humidity is to watch the shedding process. If shedding is difficult and results in a fragmented shed, increase your humidity. A light misting every other day (daily while shedding) with a plant mister will work nicely. Another method of providing humidity is to provide a Tupperware® nesting box with slightly moistened peat moss as a hiding spot.

   It is also important to avoid excess humidity as skin problems (scale rot, fungal infections, etc.) may result. If condensation in the cage is readily apparent without misting, or if the substrate is completely moist, then re-evaluate your methods.

D. *Privacy.* Hiding places are recommended for *all* reptiles, but especially for extremely shy reptiles like ball pythons. Good hygiene requires that these hiding areas be easily cleaned. But they must also be large enough to overlap part (but not all) of the heated portion. It won't help to have the hiding place at one end of the cage and the heat source at the other.

E. *Size.* The larger the cage, the better, but heat must be adequate not matter what the size is.

F. *Decoration.* Arboreal reptiles need branches or other items to climb while burrowers require a substrate that facilitates active burrowing (sand, corn cob). Keep the cage simple, which will encourage more frequent cleaning and eliminate cage related injuries.

G. *Manner of Cage Construction.* Again, try to keep things simple. Use a construction material that lends itself to easy cleaning, and try to eliminate sharp edges and excessive screen. Cage-related wounds can result in disastrous bacterial infections.

H. *Roommates.* This should be obvious. Don't solve your anorexia problem by letting your reptiles dine on each other. Do research to see if your reptiles are compatible.

I. *Substrate.* Let the controversy begin! Everyone has their own opinions, and there is no absolute right or wrong.

In my opinion, the *type* and *age* of the reptile determine the type of substrate required. For instance, juvenile kingsnakes are best kept on paper so they don't accidentally ingest any substrate and to observe stools easier. Mature kingsnakes do better in sand, corn cob, or wood chips where they can burrow and live more normally. My substrate preference, from best to worst, is:

	Snakes			**Turtles**		**Lizards**
1)	Sand	5)	Astroturf	1) Dirt	1)	Astroturf
2)	Corn cob	6)	Rocks/Gravel	2) Ground oyster shell	2)	Paper
3)	Paper	7)	Peat moss	3) Pellets	3)	Sand
4)	Wood chips	8)	Leaf litter	4) Paper	4)	Corn cob

The pros and cons of these substrates are:

1) Sand. While I always expected sand would be hard to clean, it is actually easily cleaned if "scooped" frequently. I have heard from a number of persons worried about possible ingestion problems but I have seen no problems due to the small grain size. Sand is a nice substrate for burrowing reptiles. Disadvantages include dust (minimized by using "washed sand"), weight, and price. Sand has also been implicated as a source of fatal intestinal impactions in both turtles and lizards. While small amounts will obviously have minimal effect, some of these reptiles ingest enough to cause problems.

2) Corn Cob. This is an excellent bedding. It is dust free, good for burrowing, and able to soak up and dry feces (good odor control). It is easily cleaned by scooping soiled areas with a spoon or similar utensil as done with sand. Disadvantages include cost and potential ingestion by small reptiles. I don't use this bedding with snakes less than 12 inches. Sloppy reptiles, such as turtles, can cause a tremendous mold problem by supplying continuous moisture to the corn cob bedding.

3) Paper. I admit it, paper looks like hell! However, it is cheap, easy to clean, bacteriostatic, and won't be ingested. This is my substrate of choice for juvenile reptiles or sick reptiles being treated.

4) Wood Chips. Cedar chips are an absolute no, because of toxic fumes (stick your head in a bucket of turpentine and you'll see what I mean), but pine chips are very acceptable. Pine chips are cheap, easy to clean, and lend nicely to burrowing. I again avoid chips in small reptiles or sloppy eaters as ingestion can be a problem.

5) Astroturf. Looks nice, isn't ingested, and is very versatile. However, I find astroturf hard to clean and my reptiles always rearrange it by pushing it all over the cage. Some persons have stated that the highest quality astroturf (up to $20/sq. yd.) will not be rearranged and holds up to numerous cleanings.

6) Rocks/Gravel. Looks nice, but unacceptable due to the difficulty in cleaning, tendency for waste products to accumulate, breeding successes of parasites, and the tendency of lizards and turtles to purposely and accidentally ingest them.

7) Peat Moss. Moist peat moss is acceptable in hiding cages for gravid or shedding, or for high humidity purposes, but not as a whole-cage substrate. Peat moss tends to retain too much moisture and its high acidity can lead to skin scalds and infection. Fecal debris, parasites, fungus, etc. thrive in this medium.

8) Ground Oyster Shell. I have personally never used this substrate, but it sounds like an excellent idea for turtles, as consumption would be advantageous instead of dangerous. Apparently this substrate is available through many pet dealers and is quite economical.

9) Alfalfa Pellets. Again, I have not used these either, but it seems like a good idea for turtles. My reservations would be cost and dust production, but again ingestion would not be a problem.

10) Dirt. Turtle people will appreciate the difference between dirt (soil) and sand. As previously stated, sand has been blamed for fatal intestinal impactions in turtles. Dirt, or a good quality soil, hasn't caused similar problems. Advantages of dirt are that it is: a. economical, b. good for burrowing, and c. not a problem with ingestion. Disadvantages are a. difficulty in cleaning, b. possible soil contaminants, and c. dust production. Overall, properly managed, this is the best all around turtle substrate.

11) Leaf Litter. I have never used this and won't, because of possible contaminants such as insects, molds, bacteria, etc.

I'm sorry if I left your favorite substrate out or knocked what you're using now. Use what works best for you but remember to consider cleaning, odor control, dust, cost, potential ingestion, and waste build-up.

J. *Cleanliness.* All substrates need to be changed eventually, preferably before they walk away by themselves. If daily scooping of sand, corn cob, etc. is provided, a good general cleaning need only be done once every 2-3 weeks or longer.

The cleanser/disinfectant I use and prefer is a bucket of sudsy water with Clorox^{R} added (1 tsp. dish soap plus 1/2 cup Clorox^{R} per 2 gallons of water). Clean the water dishes weekly or they will become a bacterial cesspool.

3. **Feeding the Right Food.** Each species has its own preference for food and if you're not willing to research this, then you may be out of luck. Some common variables:

A. *Color.* As an example, many herpetologists report that ball pythons will each black or brown mice, but not white.

B. *Type.* Similarly, many pythons/boas will eat mice but not rats, and vice versa.

C. *Alive vs. Dead.* People are surprised to learn how many reptiles prefer dead prey. I believe that most reptiles can be trained to eat dead prey, which solves two major problems: 1) maintaining smelly mice/rats in huge numbers; and 2) preventing rodent attacks on the reptiles.

D. *Pattern.* Reptiles are creatures of habit and "patterns" of eating develop early. Box turtles are an excellent example. If offered a broad variety of foods at an early age, when preferences are developing, they will sample a broad range of food. Similarly, if a hognose snake eats pinkies, lizards, and toads at an early age, it will continue to do so as an adult. If a narrow range of food is provided, then strict preferences (like toads only for hognoses, or crickets/worms only for box turtles) will develop.

While these patterns of feeding can be broken, it can be very difficult and frustrating. Scenting, head-splitting, and mixing foods are just a few techniques required to "trick" the reptile into eating the desired diet. You *must determine* your *reptiles' preferences* before a switch can be made.

E. *Size.* I'm always amazed that some people expect an 18-inch python to eat a 2 lb. rabbit. The general rule of thumb is not to offer a food item any larger than twice the size of the head of the reptile you're attempting to feed.

Many herpetologists have been amazed at how turtles and lizards (especially iguanas) will eat after their food has been chopped into small pieces. Don't you prefer to eat small pieces of watermelon rather than shove the whole thing in?

Try a pinkie or fuzzie for your stubborn mouse/rat eater and you may be surprised.

F. *Variety.* Baby chicks, gerbils, hamsters, or other foods item often have to be switched with rats or mice, especially in boas and pythons.

Some lizards (such as iguanas) and land turtles should *always* be offered a wide variety of food items, not only because they need a varied, balanced diet, but also to prevent pattern eating.

4. **Feeding in the Correct Manner.**

A. *Where Fed.* I have owned snakes that would not pursue their food - it had to be placed in the exact location (usually in the hiding spot) at the right time.

Juvenile snakes often eat better when placed in a small container with their intended prey.

Where to feed your reptile is a trial and error process and requires observation of your pet.

B. *Time Allowed to Feed.* Most experienced herpetologists recommend removing food items if not consumed within a specific amount of time (15-30 min.). This is generally true as a reptile will become apathetic toward an ever-present prey item. *However,* some reptiles require longer exposure times and this is certainly worth a try, especially in private and nocturnal reptiles.

Turtles and lizards, such as iguanas, require longer exposure (hours) as they often lounge through their meal.

C. *Frequency of Feeding.* Excessive exposure to food will "turn off" some reptiles, so don't harass them if they are not interested

5. **Feeding at the Right Time.** While this may seem obvious, there can be some subtle hints on feeding times:

A. *Shedding.* Opaque and shedding reptiles seldom eat and should be left alone. Some non-opaque reptiles which are enduring shedding difficulties will also stop eating.

I routinely lightly mist my cages once or twice daily when I notice the reptiles are opaque. This increase in humidity dramatically decreases shedding problems. If they are having shedding difficulties, I place small snakes in a peanut butter jar (with holes in the lid) with warm, moist wash cloths in a warm (80-85°F) room for 8-12 hours. The same thing can be accomplished for larger snakes by using an aquarium. The Tupperware® nesting boxes with slightly dampened peat moss are excellent egg laying containers, but work equally well for shedding chambers.

B. *Breeding.* Many gravid reptiles won't eat for many weeks prior to birthing. Most reptile breeders agree it is good to offer food, but not to be surprised by a refusal to eat.

Males often stop eating during breeding but tend to resume after days (or weeks) of "cooling off."

Juvenile reptiles often won't eat until after the post-hatch shed, or the very specific presentation of food.

C. *Nocturnal.* Some nocturnal reptiles will only feed at their natural time of feeding (night). Also, some of these guys desire privacy and won't eat while three guys in lawn chairs watch the cage like a vintage episode of "The Dating Game."

D. *Seasonal.* Younger reptiles tend to eat well year-round, but as they mature they may become "seasonally picky." Winter is the most common time for these reptiles to become picky, perhaps indicating an environmental problem (inadequate heat, light, etc.).

Hibernation is both practical and physiologically desirable for many temperate species, *if done correctly.* I have only hibernated snakes, but they do well maintained at 50-55°F for 3-4 months in shoeboxes with their normal substrate. They should be fed their last meal at *least* two weeks prior to hibernation, and allowed to cool down gradually over the course of 1-2 weeks before being placed at 50-55°F. As hibernation is ending, they should also be allowed to warm gradually for 1-2 weeks. I maintain small water dishes (for drinking only) in the hibernation boxes, although many people feel this is not required or desirable, and offer water 2-3 times during the hibernation period. Most reptiles feed voraciously after hibernation and many experts recommend that poorly feeding hatchlings be hibernated for 1-2 months.

Never attempt to hibernate a sick or failing reptile without being willing to take some risk.

E. *Handling.* Some reptiles won't eat after a recent handling, so let them settle in for 24 hours before attempting to feed.

F. *Activity.* Most reptiles become active, restless, or agitated when hungry and this is an excellent time to offer food.

G. *Moisture.* Many reptiles can be induced to eat by using a mild warm water mist before offering food.

How are you doing so far? If your reptile can pass the following quiz, thus ensuring that it fulfills all the postulates of our hypothesis, then proceed. If not, return to go, reform your husbandry practices, and quit looking for a quick fix.

REPTILE EVALUATION FORM

1. **Health**

A. Skin	Are there any lumps or bumps? Are there areas of damaged, roughened, or discolored scales? Are there any open sores or wounds? Is the reptile experiencing shedding problems? Is the shell roughened, pitted, or eroded? Answer: _____
B. Mouth	Is there any evidence of infection (missing teeth, gaping, crusty discharge, discolored tissue)? Is the reptile reluctant to open its mouth? Any bleeding? Answer: _____
C. Nostrils/Respiratory	Are the nostrils occluded? Is a nasal discharge noted? Is open-mouthed breathing observed? Is a foamy discharge seen in the open mouth when breathing? Is there wheezing? Any bleeding? Answer: _____
D. Eyes	Are the eyes swollen or non-visual? Are the eyecaps having difficulty being shed? Answer: _____
E. Parasites	Are any mites or ticks present? Have you noted diarrhea (watery, mucusy, or bloody stools)? Answer: _____
F. Gastrointestinal	Has your reptile regurgitated? Has diarrhea occurred (see previous category)? Answer: _____

G. Skeletal/Neurological — Have you noticed a lack of activity or a reluctance to move? When your reptile moves, does it have difficulty? Does your reptile seem disoriented or complacent? Does your reptile seem to be in pain?
Answer: _____

H. Overall Condition — Is your reptile underweight? Is your reptile all skin and bones? Have you noticed any abdominal lumps or bumps?
Answer: _____

If you answered yes to any of the above questions, you need to seek medical help for your reptile. If you answered no, then your reptile is probably healthy and you may proceed.

2. **Environment**

Have you provided for the following? Give yourself one point for each category you have *adequately* provided.

A. *Adequate* heat	_____	F. Adequate size cage	_____
B. Light	_____	G. Safely constructed cage	_____
C. Humidity	_____	H. Proper substrate	_____
D. Privacy (hiding area)	_____	I. Proper cleanliness	_____
E. Appropriate cage decoration	_____	J. Compatible roommates (if not alone)	_____

If you scored less than a seven (out of 10), stop and re-evaluate the environment.

3. **Correct Feeding** (The Proper Food)

Have you properly researched your reptiles' food preferences? Have you avoided feeding during shedding periods and after excessive handling? Have you taken the season into account?
If you answered yes to the above questions, proceed.

4. **Feeding Methods** (The Right Manner at the Right Time)

Have you tried the following? Score 1 point for each category you have *adequately* tried.

A. Different color food	_____	F. Feeding at night	_____
B. Different size food	_____	G. Light misting before feeding	_____
C. Different type food	_____	H. Strict non-handling before feeding (privacy)	_____
D. Alive and dead food	_____	I. Leaving food items in cage longer or shorter times	_____
E. Feeding in different locations (Where you feed or place items)	_____		

If you scored 6 or more, proceed. If you scored 6 or less, try different feeding techniques.

If your reptile has passed the quiz, then you have a legitimate anorexic reptile which has *not* responded to good husbandry practices and needs help.

INSTANT CURES FOR ANOREXIA

Got your attention, I hope. There are *no* quick fixes or easy cures. Ninety percent of anorexic reptiles can be helped by correcting the previously described husbandry problems. For the 10% that do not respond, I hope the following guidelines will prove to be helpful.

The ideal anorexic treatment program is a balance between stimulating appetite and providing supplemental nutrition until appetite and thus feeding is regained.

Stimulating Appetite

1. **Heat Therapy.** Most people believe that they provide adequate heat for their reptile, but the aforementioned $2 investment in that highly scientific instrument, the thermometer, usually proves otherwise. Heating a reptile to 80-85°F *in conjunction with* proper husbandry techniques will often stimulate reptiles to eat.

   Heat therapy in the range of 85-90°F stimulates the immune response of reptiles. However, temperatures in this range are not advised on a regular basis.

   If research suggests your reptile is from a high elevation (over 4,000 ft.), then cooling at night is appropriate. For example, an Arizona mountain kingsnake might be heated to 80-85°F during the day and dropped to 70-75° at night.

2. **Sunlight/Photoperiod.** Photoperiod is of great importance with turtles and lizards, but plays a role with snakes as well. Basking is important to all reptiles, but mainly turtles and lizards, and these reptiles should have a spotlight provided. Short periods of exposure to outdoor sunlight is a powerful appetite stimulant and should be tried. Remember that your reptile may become quite aggressive after (or during) these excursions, so be careful.

3. **Hydration.** Turtle experts have stated that providing a tepid water bath for ill or stressed turtles will allow for proper hydration and bathing. This physical and psychological boost will often result in feeding soon thereafter.

4. **Vitamin B.** Vitamin B_1 (thiamine) given by IM injection has been proven to both increase metabolism and appetite in many species. *Coupled with heat*, Vitamin B_1 injections may help *borderline* cases. Dosage is unknown but since Vitamin B is a water-soluble vitamin, toxicity is not possible. I use commercially available injectable thiamine or Vitamin B complex preparations and give an IM injection every other day for up to two weeks at a dose of 1/2 cc per 10 lbs. As stated, Vitamin B is useful in only marginal cases.

5. **Anabolic Steroids.** Anabolic steroids such as Winstrol-V® are also proven appetite stimulants in humans and mammals. Anabolic steroids function by a) stimulating metabolism, b) slowing the protein breakdown cycle, and c) providing a vague "euphoric" sensory perception.

   The adequate dose for reptiles is unknown, but I use 1 cc per 25 lbs., injected IM once a week for no more than 3-4 weeks. Unlike Vitamin B, anabolic steroids are potentially harmful. Side effects to be considered are immune suppression, sterility, and liver damage. The conservative doses given above have not been detrimental, in limited clinical experience, but more research is needed.

PROVIDING FOR SUPPLEMENTAL NUTRITION

1. **Force Feeding.** There are no exact guidelines to dictate when to force-feed a reptile. A mature python may be able to tolerate weeks/months of fasting while a juvenile reptile may be able to survive only a couple of weeks.

   I force-feed when I have exhausted *all* the aforementioned husbandry techniques and the reptile is showing signs of weight loss or failure to grow. What do we feed?

   I prefer feeding natural foodstuffs because they are a balanced meal without any guesswork. They also represent food items the reptile might ordinarily obtain in the wild. Examples of natural foodstuffs are:

   A. *Snakes*: Pinkies, fuzzies, mice or rats, lizards, goldfish, etc., depending on the size and type of snake.
   B. *Lizards*: Pinkies, waxworms, dog or cat food, insects, etc., depending on the size and type of lizard.
   C. *Turtles*: Earthworms, goldfish (minnows), insects, dog or cat food, etc., depending on the size and type of turtle.

Figure 1. Preparing to force-feed a snake. Apply a blunt, cylindrical object to force the jaw open. Note position of the open glottis in the middle of the floor of the mouth. Care should be taken to avoid inserting food or water into the glottis, which leads to the trachea (windpipe).

Figure 2. Placement of the food item in the mouth. Once the food item is in place, gently remove the gag and guide and food item into the back of the throat. Use proper sized food items (see text)!

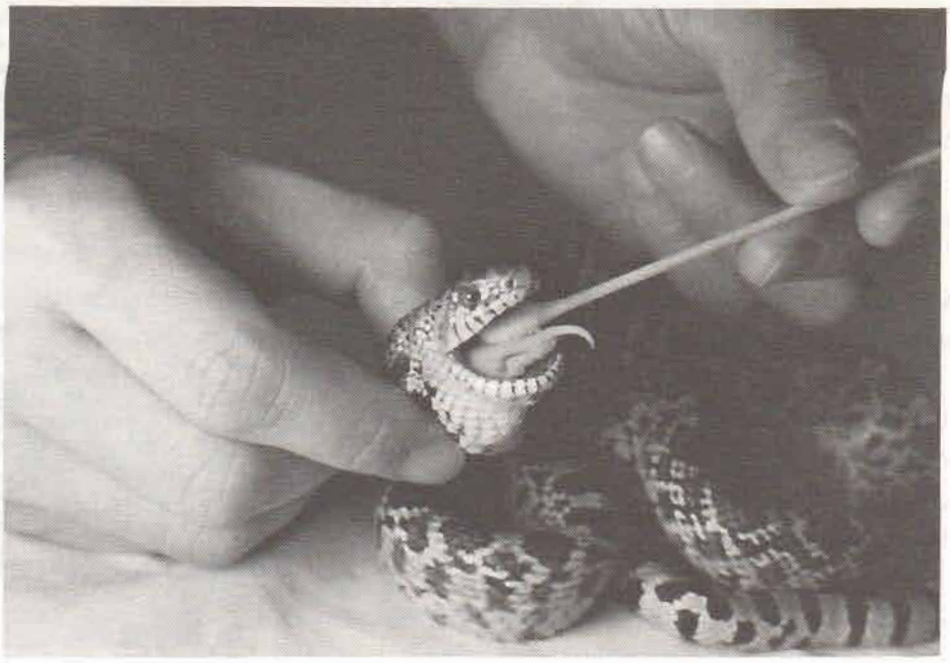


Figure 3. Encouraging swallowing of the food item. Use the blunt, smooth end of a probe to gently advance the food item until the glottis can be seen. At this point the rear teeth will usually hook the food item. If excessive resistance is met, do not force swallowing – use a smaller food item. (NOTE: The Q-Tip shown in this photograph is not recommended for advancing food items, as the cotton tip may hook and injure the teeth.)

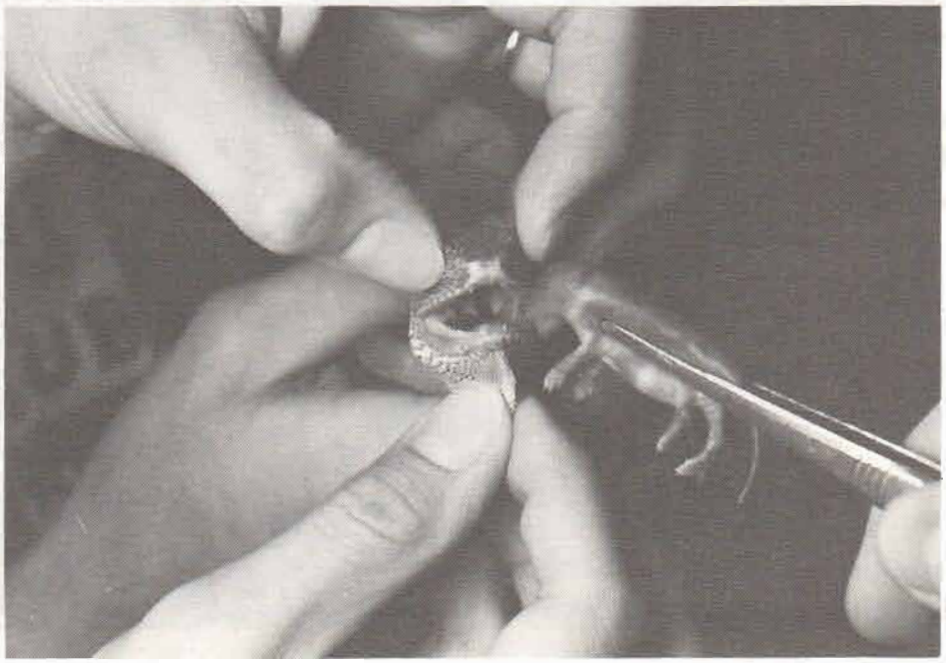


Figure 4. Force-feeding the lizard. Using gentle traction, the mouth is pried open by pulling downward on the dewlap and upward on the upper jaw. Natural foodstuffs can be fed easier to a lizard than a snake, since the former's throat is much larger once the mouth is open. The pinkie pictured here is too large and a smaller food item would be more suitable.

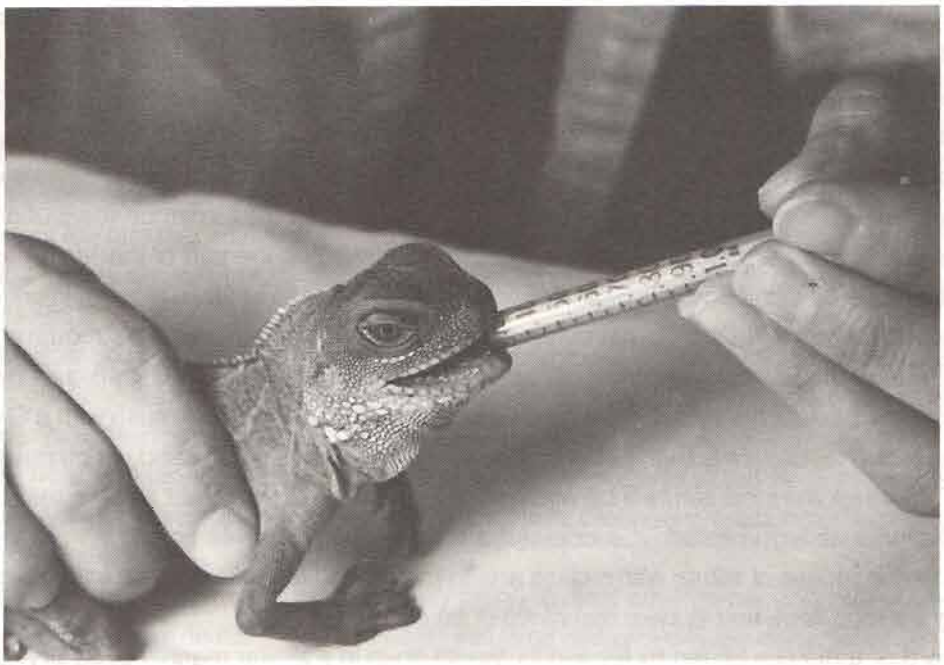


Figure 5. Force-feeding liquids to a lizard. Lizards that bite and chew at a syringe can be fed small amounts of nutrient rations as they chew on the syringe. This must be done very slowly and the lizard allowed to swallow. If a large volume is to be administered, a tube should be passed to the stomach.

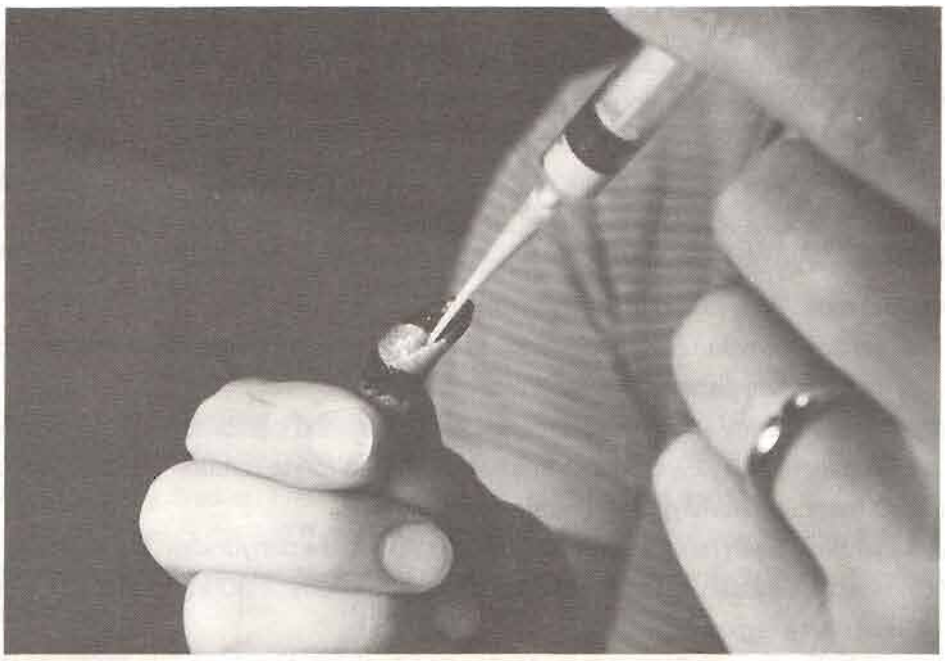


Figure 6. Force-feeding fluids to a snake. Administration of liquids such as Flagyl^{R}, Panacur^{R}, etc., can best be accomplished with soft rubber tubing, such as a "tom-cat" catheter.

You may have noticed an emphasis on protein sources. This is appropriate in an anorexic reptile. However, the foodstuffs mentioned, for the most part, are a complete and balanced meal.

Size is extremely important in determining what to force-feed. Juvenile snakes should be offered items as small as possible -- better that they are too small than too large. Lizards and turtles can be offered chopped pieces of these foodstuffs, so size can be better controlled. For juvenile snakes I prefer using pinkies that are fresh thawed because they hold together better (dismemberment of a force-fed item is a main reason for failure) and you aren't causing suffering on the part of the "victim."

In all force-feed situations, it is desirable to have assistance in order to prevent injuries to the reptile if it thrashes. *Gently* slide a *small* diameter, *smooth* instrument into the snake's mouth and lock it in place at the rear of the jaw forcing the mouth open. Quickly introduce the food item, and *gently* guide it *head* first into the back of the throat. Placing mild pressure on the food item, push with a Q-tip or similar "tool." If excessive resistance is met, let up and place the reptile into a cage and watch quietly without startling it. With luck the food item will be well placed in the back of the throat and the teeth will hook such that it cannot be regurgitated. If successful, avoid handling for 24-48 hours.

The major causes of failure with snakes are: 1) forcing too-large items, 2) startling the reptile after introducing the food item, and 3) poor technique (it takes practice!).

A lizard or turtle can be fed by forcing its mouth open in a similar manner or by prying the mouth open slowly, using one set of fingers on the lower jaw/dewlap and another on the upper jaw/nose region. Lizards and turtles tend to have larger throats than snakes and introduction of food items and advancement tends to be a little easier. It is almost impossible to force-feed a turtle that is underweight to the point that it can withdraw completely into its shell, and doing so is an extreme stress to the animal.

Please let me reinforce some important points about force-feeding:

A. *When*? Only when all other methods have failed and there is an obvious loss of weight or failure to grow.

B. *Why*? Only as an extreme measure to preserve life. Force-feeding is an extremely controversial topic and some expert herpetologists feel it is detrimental in all cases. My thinking is that it should *never* be used for convenience or an easy way out, but simply in selected cases (see When).

C. *How Often*? If force-fed excessively, a reptile will have no desire to eat. Force-feed only to sustain, not maintain. The idea is to sustain the reptile until the inclination to eat is realized.

Proper husbandry techniques and attempts to feed normally *must be maintained*, or you will be force-feeding and most likely failing in the long run.

There are many formulas and medications force-fed for various medical conditions, but it is not the purpose or intent of this paper to cover all of them.

2. **Fluid Therapy**. Fluid therapy should be attempted only by trained persons, because overhydration can be disastrous. In general, 2-1/2% dextrose in a balanced electrolyte solution (lactated ringers solution) can be given SQ (subcutaneously) or IP (intraperitoneally).

Turtles that have been refractory to force-feeding or where dehydration is a problem can be given an IP injection of approximately 2-3 cc's daily as needed. Similarly, a SQ injection can be given in the loose skin of the axillary region.

Lizards are best given IP fluids if dehydration is suspected, but due to ease of force-feeding, I seldom elect to administer fluids.

Snakes can be tubefed a mixture of good electrolyte solutions (Gator-Aid, Pedialite, etc.) orally, so again, IP fluids are indicated only in extreme cases or if nephrotoxic drugs are concurrently in use. I have administered up to 5 cc's per lb./IP without problems.

OTHER DRUGS USED TO TREAT ANOREXIA

1. **Flagyl^{R} (metronidazole)**. Flagyl^{R} is a current favorite "miracle" drug among herpetologists. It is an antibiotic which affects a) anaerobic bacteria, b) protozoans (trichomonis and glardia), and c) amoebiasis.

   Flagyl^{R} is used by some herpetologists for every regurgitation episode, diarrhea, and failure to eat. It is *vastly* overused and *has never been proven to stimulate appetite* in any way. If Flagyl^{R} works it is because of the fact that with luck you've cleared up a protozoal/bacterial overgrowth of the gut.

   Side effects are rare but sterility, liver damage, and blood dyscrasia can occur with excessive doses/amounts.

   The dosage I use is 50 mg/kg repeated once (if needed) 48 hours later. This dose is much lower than most published doses but seems to work every bit as well. I tube this with either the liquid form (available only in Mexico) or tablets ground and mixed with water.

2. **Panacur^{R} (fenbendazole)**. This is a member of the group of drugs called benzimidazoles, which are used exclusively for parasite control (worming). Unlike its precedent, thiabendazole, it is safer and has a wider range of action.

   Panacur^{R} is my wormer of choice, rarely failing to clear any internal worm diagnosed. Given by the tube orally at 10 mg/lb daily for three days, this drug has proven very efficacious. No adverse effects have appeared in hundreds of reptiles treated with Panacur^{R}.

3. **Ivermectin^{R}**. Ivermectin^{R} without a doubt will revolutionize parasite control (both internal and external). However, inadequate research and experience with dosage prevents further discussion until a later date.

4. **Levamisole^{R}**. Levamisole^{R} is an outdated wormer as the safety range is too narrow compared with Panacur^{R} and other drugs. However, this may be a potent immunostimulant that could prove useful in reptiles.

SUMMARY AND SHOTGUN FLOW CHART

If I haven't driven home the importance of proper husbandry by now, then I guess it is hopeless. I strongly advocate consulting a professional or experienced herpetologist, reviewing your husbandry techniques, and the use of good judgment before employing chemical means of anorexic treatment. The following flow chart is by no means intended to encourage the use of drugs for anorexia. the flow chart is meant to *illustrate* the potential use of these drugs and to hopefully stop their use as a first-step procedure. Every reptile and every situation is a little different. An extremely sage portion of the Hippocratic oath of physicians states, "Above all, (let your treatment) do no harm."

RECOMMENDED READING

While there are *many* excellent sources of information that should be taken advantage of, several articles and books I have found to be quite good are:

Applegate, R. Feeding Baby Snakes. 1762 Pepper Villa Dr., El Cajon, CA 92021.

Fogel, D. 1988. Captive Care of the Ball Python, NOTES FROM NOAH, Volume XV, No. 8, May 24, 1988.

Frye, F. 1981. Biomedical and Surgical Aspects of Captive Reptile Husbandry. V. M. Publications, Inc., Banner Springs, KS.

Lewis, L. 1977. Nutritional Diseases and Disturbances in Mineral Metabolism Characterized by Changes in Blood Levels. Colorado State University Veterinary Notes.

Lewis, L. 1978. Nutrition and Feeding Notes. Colorado State University Veterinary Notes.

Marcus, L. 1981. Veterinary Biology and Medicine of Captive Amphibians and Reptiles, Philadelphia, Lea, & Febiger.

Wallach, J. 1969. Medical Care of Reptiles. J. Am. Vet. Med. Assoc. 155:1017-34.

Wallach, J. 1977. Management and Nutritional Problems in Captive Reptiles. Current Veterinary Therapy, Vol. VI.

ITEMS MENTIONED IN TEXT

Cages: STA-IN-PETS, P.O. Box 2932, Santa Fe Springs, CA 90670 (213) 946-0804

Shelf System Using Heating Cables: For an excellent article on how to build such a system, send a SASE to Brian Backner, M.D., 17 Margaret Rd., Sharon, MA 02067

FLOW CHART TO ILLUSTRATE MEDICAL INTERVENTION OF ANOREXIA

All husbandry techniques have been reviewed and reinforced

↓

failure to eat → Significant Medical Problems -- seek professional help

No discernible medical problems are noted. Your reptile has passed the previous reptile health quiz.

A. Increase heat to 80-85°F night and day
   1. Use a thermometer
   2. Cool @ night if indicated
B. Try sunlight exposure
C. Hydration is adequate

Relatively healthy, active, and non-emaciated reptile

↓

Provide specific appetite stimulant

A.Vit B_1
B.Winstrol-V (?)

↓

Still refuses to eat

↓

Force-feed and continue good husbandry techniques

Emaciated, listless, inactive reptile

↓

Provide appetite stimulant & nutritional supplements

A.Vit B_1
B.Winstrol-V (?)

↓

Still refuses to eat

↙ *"Shotgun" Approach*
Panacur + Flagyl + continue force-feeding. Fluid therapy if required.

↘ *Conservative Approach*
Continued force-feeding

Success!
Resume and practice good husbandry

Still Anorexic?
"See a pro or start digging the hole."

Sheep Draw Veterinary Hospital
6297 W. 10th St.
Greeley, CO 80634

BANDED TREE VIPER (*Trimeresurus wiroti*).
Photo by William B. Love.

YELLOW BLOTCHED MAP TURTLE (*Graptemys flavimaculata*).
Photo by Gary Meslaros.

PAINTED FROG (*Atelopus varius*).
Photo by David M. Dennis.

THREE STRIPED ASIAN BOX TURTLE (*Cuora trifaciata*).
Photo by David M. Dennis.

PUERTO RICAN CRESTED TOAD (*Peltophryne lemur*).
Photo by David M. Dennis.

INDOCHINESE BOX TURTLE (*Cistoclemmys galbinifrons*).
Photo by David M. Dennis.

HUAMANTLAN RATTLESNAKE (*Crotalus scutulatus salvini*).
Photo by David M. Dennis.

AUSTRALIAN DEATH ADDER (*Acantophis antarticus*).
Photo by William B. Love.

GIANT HILL TURTLE PLASTRON (*Heosemys grandis*).
Photo by Alan Foust.

GIANT HILL TURTLE CARAPACE (*Heosemys grandis*).
Photo by Alan Foust.

BORNMUELLER'S VIPER (*Vipera bornmuelleri*).
Photo by William B. Love.

WEST AFRICAN BURROWING PYTHON (*Calabaria reinwardti*).
Photo by William B. Love.