CAPTIVE PROPAGATION AND HUSBANDRY OF REPTILES AND AMPHIBIANS

Edited by Randall L. Gray

Northern California Herpetological Society

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CAPTIVE PROPAGATION AND HUSBANDRY OF REPTILES AND AMPHIBIANS

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Acknowledgements: Few conferences are the product of one or even a few individuals, but the collective talents and energy of many. A few words of thanks published in the proceedings are not adequate payment to all who helped in the conference, but then their reward came with helping to organize the event.

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PREFACE

The following papers were presented at the Northern California Herpetological Society (NCHS) and Bay Area Amphibian and Reptile Society (BAARS) Conference held at the University of California, Davis on January 19 and 20, 1985. This was the second in a continuing series of conferences dealing with the captive propagation and husbandry of reptiles and amphibians.

As with the first conference, the purpose was to provide a forum for the exchange of information dealing with herpetoculture. The exchange of information is not only restricted to the delivery of papers but during the breaks. Seldom do several hundred herpetologists gather in one place and fill the air with technical (and not so technical) gossip. I have often wondered if I learned the most during the sessions or over lunch with enthusiastic herpetologists.

The value of publishing the proceedings of conferences of this type is often questioned by members of the scientific community. They feel that the validity of the information contained in the papers must be tested and/or critically analyzed before it is set in print and thus interpreted as a fact by the reader. We recognize this concern and caution the reader to examine the contents of the papers and use good judgement in distinguishing between theories and facts.

Proceedings are a valuable tool to those who are seeking information about a group of herptiles they know little or nothing about. Even when the information is considered "standard knowledge" by the more advanced herpetologist we must not forget that the beginners need a starting point.

To all those who are not listed above but provided assistance with the conference and/or proceedings, I give a special thanks. Your names were not listed only because of my oversight.

Randall L. Gray
Editor
June 1985

Cover photo: Flat faced rain frog (*Breviceps fuscus*). Coastal mountains of South Africa. Photo by S. Middleton.

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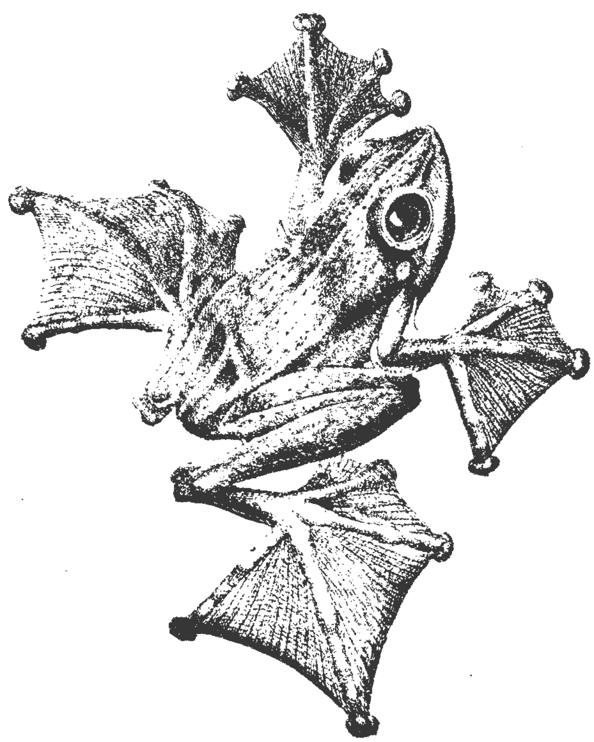

THE ECOSYSTEM APPROACH - NEW SURVIVAL STRATEGIES FOR MANAGING AND DISPLAYING REPTILES AND AMPHIBIANS IN ZOOS

Sean McKeown

Natural areas around the world are disappearing at an alarming rate. By the year 2000, many thousands of plant and animal species that exist today will be extinct. Reptiles and amphibians will not only lose their habitat, but in some cases, their temperature and humidity gradients as well (Pawley, 1983).

The American Association of Zoological Parks and Aquariums (AAZPA) is beginning to implement a Species Survival Plan (SSP) which will include reptiles and amphibians that meet certain key criteria. The SSP can be defined as an effort to "develop scientific and cooperative programs to propagate and preserve endangered species in captivity through population mamagement" (Foose, 1981).

In order to contribute more effectively to the long term captive survival of both endangered and potentially endangered herpetofauna, it may be time for zoos to consider <u>new strategies</u> for housing and displaying reptiles and amphibians.

Perhaps we should examine how reptile exhibits came to be what they generally are today. The first "modern" zoo to specifically build a structure to display reptiles and amphibians was the Zoological Society of London. The Society, founded in 1829, built its first reptile house in 1849 (Blunt, 1976). Although this structure was not created to house just snakes, they were the major group exhibited for the public (Ball, 1983).

Over the next 135 years the role of zoos developed from principally amusement through the "postage stamp" display approach of the 1950's and 1960's into today's multifaceted institutions.

During the past ten years major husbandry and captive breeding advances have occurred for reptiles and amphibians in both zoos and private collections. Many of these advances are a direct result of the exchange of information presented at captive breeding symposia. The use of natural spectrum fluorescent lighting, availability of a variety of insect food, routine use of vitamins and minerals, professionally trained zoo personnel and control techniques for ectoparasites have all contributed to better management. The end result has been a greater variety of display and breeding programs for most reptiles and amphibians, not just snakes.

Another positive trend within the past ten to fifteen years is the maintenance of some reptiles and amphibians outdoors in enclosures duplicating the natural environment as closely as possible. In the 1960's Chuck Shaw at the San Diego Zoo and Jack Throp at the Honolulu Zoo bred many Galapagos tortoises (Geochelone elephantopus) following this philosophy.

During the 1970's and early 1980's the Dallas Zoo had success maintaining and breeding American iguanid lizards in a large "naturalistic" outdoor area. The Atlanta Zoo did the same with bog turtles (Clemmys muhlenbergi). The Honolulu Zoo had modest success maintaining a variety of tortoises, turtles, lizards and crocodilians in "naturalistic" outdoor enclosures under specific environmental parameters in terms of sun, shade, humidity levels, types and amount of vegetation, use or non-use of soak pools and the selection of various natural substrates. In the private sector the remarkable accomplishments of Dick Bartlett (Reptilian Breeding and Research Institute) have encouraged others to develop "naturalistic" outdoor enclosures.

Still, indoor herpetariums (reptile houses) in zoos are not radically different from the first reptile house built in 1849.

Although the majority of reptile facilities now display a cross section of herpetofauna, virtually all are designed primarily for housing snakes. Snakes can be managed as solitary reptiles or in pairs on display and in small containers on reserve. Offspring can be managed successfully in a similar manner. Most reptiles and amphibians, however, are found in large social groupings during most of the year in their natural environments and can best be managed in a similar manner in captivity.

It may now be time for a reptile house "metamorphosis" - a new direction. Reptile facilities could better serve ectotherms and the viewing public if they were designed and built as a planned series of huge, temperature/humidity regulated "naturalistic" displays extending floor to ceiling with skylighting to allow in the natural sunlight. Such displays could serve to inform the public and give the zoo visitor an appreciation of herptiles while affording superior management of these animals.

Imagine a reptile and amphibian facility consisting entirely of ecosystems supporting large breeding groups of herptiles. Each display would afford viewing from at least two sides. The floor of each display would consist entirely of natural substrate and would be planted with native vegetation from that ecosystem. Skylighting would be utilized to allow natural sunlight to enter the exhibit, thus providing a natural photoperiod. Habitats and species of herptiles selected could be based on SSP and other conservation priorities as well as the proven husbandry skills of reptile staff members.

Wes Chun, a neotropical saurian expert, who earns his living as a graphic artist for Disney Studios in Burbank, California, has contributed a sketch of how one of these ecosystems might appear (Figure 1). This figure depicts a Madagascar Island Ecosystem display housing a colony of Madagascar angulated tortoises (Geochelone yniphora) on natural grasses. The palms and other plans support three species of lizards including two types of Phelsuma and one taxa of Uroplatus. The landscaping continues outside the exhibit into the public viewing area. The artificial rocks, made out of fiberglass, give the viewer a feeling of being drawn into the environment.

Each individual ecosystem would have a temperature control panel to regulate temperature highs, lows, and


The Endangered Ecosystem
Endangered Reptiles
Our Conservation Program

FIGURE 1

photoperiod. An automatic sprinkler system would reduce care time for trees, plants, and grasses. Food would be provided at specific locations or "feeding stations" making it accessible to each of the different species in the enclosure. Each display would be treated as a separate quarantine area with its own reptile handling and maintenance equipment to minimize the transfer of diseases between exhibits.

Modern graphic panels would tell about the uniqueness of each ecosystem and why major portions of the ecosystem are endangered. Panels would also illustrate and inform about which species are being displayed and what the zoo's conservation goals are for the species exhibited.

Techniques developed within the past ten years now make it possible to breed herptiles from all major groups. Display technology has not taken advantage of these advances. The ecosystem approach affords sound genetic management and would allow for the retention of a greater number of offspring. Behavioral data collection could be enhanced. Behavioral and reproductive studies could be applied to managing remaining wild populations or utilized for the reintroduction of species into portions of their former range.

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Desert collared lizard (Crotophytus insularis). Photo by R. Gray.

Timor monitor (Varanus timorensis). Photo by R. Gray.

CAPTIVE PROPAGATION AS A TOOL IN WILDLIFE MANAGEMENT

Randall L. Gray

INTRODUCTION

The earth is home to about 1.5 million species of living organisms. Flowering plants, fish, amphibians, reptiles, birds and mammals comprise about 290,000 of the species, of which plants account for 85 percent of this number (Ehrlich and Ehrlich, 1981). Predictions are that the total number will increase by another 10 to 50 million once all the species are described by science. The majority of the species yet to be discovered are insects.

The wealth of species is not equally distributed throughout the earth's various regions. Tropical rainforests only cover 7 percent of the earth's surface and yet they contain more than 40 percent of all living plants and animals. This fact makes the ongoing deforestation of the tropics a global concern. We are losing species at an unprecedented rate as these forests are cut for lumber or converted to range or cropland.

But we do not have to look to the tropics to see species on the edge of extinction. The federal government has listed 24 species of reptiles and amphibians in the United States as either threatened or endangered. Many states have gone beyond the federal lists and have added additional local herps. California has a total of 15 herps listed as rare, threatened, or endangered.

As I write these introductory paragraphs, I am reflecting back on my survey for the endangered blunt nosed leopard lizard (Gambelia silus) this morning in the southern San Joaquin Valley of California. This species exemplifies the problems facing most endangered species. Most of the San Joaquin desert has been converted to agriculture and leopard lizards do not fare well in cotton fields. Along with the blunt nosed leopard lizard other species such as the San

Joaquin kit fox (Vulpes macrotis), Fresno kangaroo rat (Dipodomys nitratoides), San Joaquin antelope ground squirrel (Ammospermophilus nelsoni) and a variety of plants find refuge on isolated blocks of remaining desert habitat. This remaining habitat is still being converted to other uses while concerned groups with limited funds try to buy some habitat for the survival of these species.

The loss of habitat for all the world's species is a result of the increasing human population. More people means that more land is needed for housing, farming and oil and mineral extraction. Although the human population is growing fastest in the Third World countries where tropical rain forests occur, we must keep in mind that one American child uses the same amount of resources as 16 children in Ethiopia.

With many species facing extinction the conservation community is looking with skepticism to captive propagation as one method for species protection. The herpetological community has joined this movement and many zoos and private individuals have established colonies of reptiles and amphibians that are declining in the wild.

Recent years have too often seen the banner of captive propagation waved as the ultimate species survival technique. Many individuals, primarily in the private sector, have justified illegally collecting as a means to establish captive populations for "the species' own good." The most radical statement I have heard (and totally unacceptable to me) was, "all species will soon be extinct in the wild so we must collect them now and get breeding colonies going." The idea of zoos or private collections being the only place that we can go to see a species of reptile and amphibians is both repugnant and frightening.

Captive propagation is a valuable tool to aid in the protection of a species but it must be used in conjunction with other techniques such as laws governing the harvesting of species. An example of a successful law is the U.S. Endangered Species Act in the United States which has proved very successful with the American alligator (Alligator mississippiensis). Too many of today's reptile and amphibian breeders ignore this fact and have discounted all wildlife laws because of a few poor ones.

The single most important factor to insure a species survival is the protection of habitat. Preserving areas that provide the necessary interspersion of food, water and cover is the most important goal to insure the long term maintenance of a species. The beauty of habitat preservation is that, in reality, you are preserving an ecosystem. If you set aside habitat for the blunt nose leopard lizard you are also setting aside habitat for the San Joaquin kit fox, the San Joaquin antelope ground squirrel, the Fresno kangaroo rat, and many other species of plants and animals. Society (you and me) must accelerate its efforts to preserve large areas of the world's unique habitat for the benefit of wildlife and man.

ROLE OF CAPTIVE PROPAGATION

Reptiles and amphibians are collected in staggering numbers to supply the pet trade of North America, Europe and Japan. In Buenos Aires, Argentina, approximately 141,000 boas, 5,000,000 lizards, and 83,000 caimans were exported between 1976 and 1979 (Mares and Ojeda, 1984). This number does not include those individuals that died between collecting and exporting. Successful propagation programs can reduce the exportation of wildlife. Several species of reptiles and amphibians are seldom imported into the United States because they are being produced in large numbers by private breeders. Some of the species receiving this benefit are leopard geckos (Eublepharis macularius), plumed basilisks (Basiliscus plumifrons), Sinaloan milk snakes (Lampropeltis triangulum sinaloae), gray banded king snakes (Lampropeltis mexicana alterna), and ornate horned frogs (Ceratophrys ornata).

Captive propagated turtles have long been available to the pet trade. In 1970 a turtle farm in Mississippi produced 1.5 million captive hatched turtles (Honegger, 1980). Subsequent fears concerning the spread of the disease Salmonella restricted the sale of captive produced turtles in this country.

Sea turtles are exploited for their flesh as well as ornaments made from the shell. Attempts are being made in the Grand Cayman Islands of the Carribean to satisfy the turtle meat market through captive propagation. Many people believe that captive propagation of sea turtles will only increase the demand for turtle products. Increased demand could possibly lead to heavier black marketing of wild populations (Dodd, 1982).

Crocodiles are another species used for food and leather products. Crocodile farms in Australia, New Guinea and Thailand are trying to satisfy the demand for these products through captive propagation. In Thailand, over 6,000 salt water crocodiles (Crocodylus porosus) are in captivity. The number in the wild is believed to be around ten adults (Finkelstein, 1984). As with sea turtles, captive propagation could also be seen as a two edge sword. Presently captive colonies of crocodiles are supplemented by taking eggs from the wild. Should commercial crocodile farming prove successful, then the economic rationale for keeping wild populations and its habitat would diminish. This is not a pleasant outcome for successful propagation efforts.

The green iguana (Iguana iguana) has been reduced in numbers in Mexico and Central America because of habitat destruction and hunting for a food source. Scientists at the Smithsonian Tropical Research Institute in the Republic of Panama have developed a reliable method for incubating and hatching eggs of the green iguana. They are trying to develop methods for establishing "iguana ranches". These ranches would provide a food source for the local people and reduce hunting pressure on wild populations (Jacobs, 1985). But even these important goals must be balanced with habitat protection.

Captive propagation can also play a role in educating the public about endangered species as well as other species not presently threatened. Propagation projects can provide animals

San Esteban Chuckawalla (Sauromalus varius). A breeding program for this endangered lizard has been started at the Arizona-Sonora Desert museum in Tucson, Arizona. Photo by R. Gray.

Crocodile farm in Northern Territory, Australia. Photo by R. Gray.

for zoo exhibits which provides people an opportunity to get to know the species and develop an appreciation for its uniqueness. It is easier to motivate society to protect a well known species than an obscure salamander whom few have ever heard of, let alone seen.

Captive propagation can add to our understanding of the reproductive biology of a species. This information could prove useful in managing wild populations.

The successful propagation techniques developed for the more common species can help with the propagation of endangered species. It is preferable, if possible, to work with closely related species before taking significant numbers of an endangered species into captivity. Research with Andean condors (Vultur gryphus) provided valuable information to begin a captive propagation program for the California condor (Gymnogyps californianus).

The primary goal of most captive propagation programs for endangered species is eventual reintroduction back into the wild. This reintroduction can be used to supplement wild populations that have declined or have been eliminated due to natural or man-induced catastrophies. Another technique that should be discussed is "head starting". This differs from captive propagation in that wild eggs are collected, hatched in captivity, and the young released.

To understand the value of head starting you need to be aware of the survivability of individuals in the wild. In order for a species to survive it must reproduce itself. If none of its offspring survive, then the species declines; if several offspring survive to reproduce, then the species is increasing; to maintain a stable population each animal need only replace itself with one offspring that reaches sexual maturity.

Nature is a collection of environmental obstacles that makes survival difficult. Many populations of herps have a very high mortality, especially during the first year. Great Basin rattlesnakes (Crotalus viridis lutosus) only have a 40 percent chance of surviving the first year of life (Woodbury, et al., 1951). If the snake survives the first year its odds for survival in subsequent years are higher. Sea turtles are another example of a species with high mortality rates for the first year of life. Some estimates are that only 5 percent of hatched turtles survive to one year of age.

Predators are the primary cause for high mortality during the first year. As the animal increases in size and learns to be more adept at escaping predators it is less vulnerable to predation.

Head start programs are being used with sea turtles. Wild laid eggs are collected and incubated to hatching. The young are then raised to one year of age. Ninety-five percent survivability of hatchlings through the first year can thus be obtained. The larger sea turtles are then returned to the wild. Hopefully this means that there will be an increase in the number reaching sexual maturity.

The government of India appears to have brought the gavial (Gavialis gangeticus), a crocodilian, back from the brink of extinction with head starting. Eggs are collected from wild nests, hatched in captivity, and the young reared past the age of suspected maximum mortality and then released back into the wild in suitable habitat (Groombridge, 1982).

Captive colonies are used to provide stock for reintroduction into the wild. Some notable examples of captive bred wildlife being successfully reintroduced into the wild are the giant Canada goose (Branta canadensis), and the Peregrine falcon (Falco peregrini). The Peregrine falcon was eliminated from the eastern United States but now there are at least 27 wild established breeding pairs due to release of captive produced birds. This number is expected to double in the next year. An outstanding success story is exhibited by the giant Canadian goose (Branta canadensis maxima) which was eliminated from the wild in the north central United States. Captive bred animals were used to reestablish a population that now numbers over 20,000 birds (Lee et al., 1984).

The U.S. Fish and Wildlife Service and Houston Zoo have undertaken a captive propagation program for the endangered Houston toad (Bufo houstonensis). Habitat destruction has reduced the numbers to approximately 1500 animals. During 1979, 1980 and 1981 adult pairs of toads in amplexus were brought to the Houston zoo. From these founder animals captive raised individuals and newly metamorphosed toadlets, along with wild caught eggs, were introduced into the Attwater Prairie Chicken Refuge in Texas to establish a new population (Karl Peterson and Hugh Quinn, personal communication). The jury is still out concerning the success of this attempt. Only time will tell if the population becomes self perpetuating.

Galapagos tortoises (Geochelone elephantopus) have been bred at the Darwin Research Station in the Galapagos Island in outdoor enclosures. The eggs are collected and placed into incubators. The young tortoises are used to restock areas that were previously depleted of these huge tortoises (MacFarland and Reeder, 1975).

REINTRODUCTION INTO THE WILD

As I discussed above, the primary goal of captive propagation programs for endangered species is reintroduction into the wild. In order to restore animals to the wild you must have suitable habitat. Setting aside habitat for future reintroductions must take many parameters into account. The question facing wildlife conservationists today is how big a piece of habitat is needed. To answer this question, the scientific community has looked to the theory of island biogeography for ideas. Simply stated, this theory predicts that the smaller the island, and the further it is away from other large land masses, the fewer the species. Research in the West Indies has shown that the smaller the island, the fewer the number of reptiles (MacArthur and Wilson, 1967).

A wildlife refuge is analogous to an island. Instead of being surrounded by water, the refuge may be surrounded by cropland or urban development. How large the refuge needs to be depends somewhat on the species being managed. The theory of island biogeography indicates that if we only preserve 10 percent of the original ecosystem in parks, we will eventually lose 50 percent of the species in the preserve. Further, if the remaining 10 percent of the ecosystem is preserved in many small parks we will lose an even greater percentage of the species over time.

Carrying capacity is another ecological concept that should be well understood by anyone propagating species for reintroduction into the wild. This concept refers to the capability of a piece of habitat to support a given species. The blunt nose leopard lizard needs about two acres of desert to support one leopard lizard. If the area being considered for reintroduction of captive born leopard lizards is already at carrying capacity it will not support additional lizards. Animals should only be released into areas that are below the carrying capacity.

The success of captive propagation for reintroduction into the wild is measured by the survival of released animals. There is very little information about the survivability of reintroduced reptiles and amphibians. One ongoing study with the eastern indigo snake (Drymarchon corais couperi) has monitored wild populations in which captive born eastern indigo snakes and confiscated individuals were let loose. Several marked snakes were recaptured eight years later (Ken McCloud, personal communication). On the other hand, indications are that captive born desert tortoises (Gopherus agassizii) released into the wild are not afraid of coyotes and do not dig burrows for protection (John Brode, personal communication).

The nene goose (Branta sandvicensis) of Hawaii was reduced to only 35 birds by 1942. In 1960 20 captive raised birds were released into the wild, but these individuals had difficulty in adjusting. Better success has been achieved recently by rendering adult birds flightless and placing them in large, isolated outdoor enclosures. The captive born young have no contact with humans and can fly out of the enclosure when ready. But the birds are still having difficulty in maintaining self sustaining populations (Yates, 1984).

In another study captive bred mallard ducks (Anas platyrhynchos) were released just prior to the develoment of their flight feathers. Forty-three percent of the captive released birds returned to the release site the following year (Lee and Kruse, 1973). This is remarkable considering that waterfowl populations usually have a 60 to 70 percent turnover (mortality) each year.

Disease is another important factor needing consideration before releasing animals into the wild. In captivity viruses and gram-negative bacteria are a serious problem. A bacterial or viral epidemic can destroy years of captive breeding efforts and totally eliminate the founder colony. Diseases that are hosted in captive produced animals are introduced into wild populations along with the animal. If the disease is not already present in the wild population then it is likely that there is very little, if any, natural resistance to the pathogen.

Waterfowl biologists are concerned about duck virus entritus (DVE) which has a high mortality rate. This disease was first detected in captive raised birds. Over the last decade DVE has popped up in various areas.

A paramyxovirus is causing a severe problem to rattlesnake collections in many zoos. This virus has killed many of the founder individuals of the Aruba Island rattlesnake (Crotalus unicolor). This rattlesnake is a rare species occurring on Aruba Island off the coast of Venezuela. If this disease was introduced back to Aruba Island it could devastate the remaining populations.

Howard Lawler and James Jarchow at the Arizona Sonora Desert Museum are investigating pathological parameters in wild populations of selected herpetofauna in the Sonoran Region. This information will provide a profile of naturally occuring disease factors to compare against captive diseases. This information will be valuable in reintroducing the selected species back into the wild.

An analogy that brings the point home about exotic diseases is European man's first contact with native Americans. Diseases of little consequence to the early explorers, such as smallpox, helped to destroy the new world civilizations.

Another consideration in reintroduction of captive born animals is maintaining the integrity of the existing wild population's gene pool. A population is a group of interbreeding animals of the same species in a given area that share a common gene pool. The more isolated the population from other populations the more distinct the gene pool. A very conspicuous gene pool difference is exhibited by color or banding variations in different populations of kingsnakes (*Lampropeltis getulus*).

If a population were to stay isolated from others for a great length of time the population may evolve into a separate subspecies and eventually a new species. By introducing captive born animals from a different population into an established population you could alter the evolutionary future of that population. The geneticist Ian Franklin remarked, "Do we wish to conserve elephants, or insure the survival of its elephant-like descendants." When we have the luxury to reintroduce captive animals back into the wild population from which the founders were obtained we should do so. Therefore it is important to keep exact locality records for founder animals so that the genetic background of the animal can be determined.

The Arizona-Sonoma Desert Museum in Tucson is setting up breeding colonies for three species of montane rattlesnakes (i.e., *Crotalus willardi*, *C. pricei*, and *C. lepidus klauberi*). They are obtaining the founder animals from specific populations, in one case from the same watershed. Such progeny would be genetically suitable for release into the parent populations (Lawler and Belcher, 1983). This type of good planning should be incorporated in other captive breeding projects.

GENETIC CONSIDERATIONS

Over 100 years ago, Charles Darwin noted that inbreeding (breeding of closely related individuals) leads to a reduction in vigor and fertility. We now call this inbreeding depression, which refers to reduced fertility, smaller birth weights, and higher juvenile mortality.

By the mid 1970s many conservationists were worried about the effects of inbreeding in small populations of both captive and wild animals. A paper published in 1977 showed that highly inbred Prezwalski's horses (*Equus przewalskii*) were less productive than those that had not been as severely inbred. Another study looked at 44 species of mammals in captivity. The researchers found juvenile mortality of inbred animals was higher in 41 of the 44 species (Tangley, 1984).

There are few examples to draw from to demonstrate the deleterious effects of inbreeding in reptiles. The Houston Zoo has observed scalation abberation in highly inbred albino western diamondbacks (*Crotalus atrox*). The Telfar skink (*Leiolopisma telfairi*) of Round Island in the Indian Ocean may also be showing signs of inbreeding in wild populations. The insular population is small and often further reduced by hurricanes. The small population size encourages inbreeding (Cooper, 1983).

Early attempts to captive breed the endangered black-footed ferret (*Mustela nigripes*) were stymied. This was because the founder animals were already showing signs of inbreeding due to a very small wild population.

The Speke's gazelle (*Gazella spekei*) colony in the United States was started from one male and three females. Severe inbreeding depression was encountered in these animals which jeopardized the long-term maintenance of a captive colony. Several years ago, a breeding program was set up for the Speke's gazelle. This propagation program was predicated on the fact that inbreeding in itself creates selective conditions that will lead to the elimination of inbreeding depression. This program showed positive results within three generations (Templeton and Read, 1983).

If this breeding program proves feasible for other species we may have a way to avoid some of the adverse effects of inbreeding. Still a question raised by this type of breeding is whether the selected-for genotypes can survive in the wild.

Hugh Quinn and Karl Peterson of the Houston Zoo are setting up a similar breeding program for the Aruba Island rattlesnake (Crotalus unicolor). A small group of founder animals were brought to the United States. This number has been reduced in recent years by paromyxovirus, as discussed earlier. It will be several years before the results of this attempt to head off inbreeding depression will be available. Hopefully it will be successful and open up opportunities for other species of reptiles.

Another concept important to wildlife managers is genetic variability. The genetic diversity of populations has long been recognized as an important aspect of species survival. The variability of individuals in a population provides a better potential for some individuals to survive through periods of environmental stress. A well known example is insects and the pesticide DDT. Originally small doses of DDT would greatly reduce agricultural crop pests, but a few individuals would survive. The surviving individuals possessed a natural resistance to the given dosage of DDT. These resistent insects rapidly multiplied and once again became a pest. The next application of DDT had little impact since the majority of the population was resistant. Higher dosages of DDT were required and the cycle repeated itself again, and again.

There has been concern about the limited genetic diversity in elephant seals (Mirounga anqustirostris). This large mammal occurs off the California coast. Hunting reduced the population to about 20 animals but legal protection has allowed the population to climb back up to around 70,000 to 80,000. Electrophoretic studies show that there is very little genetic diversity in these animals. If a new virus were to enter the population it could create a catastrophe. Hopefully as the population continues to grow the genetic diversity of the population will diverge in various subpopulations.

Maintaining genetic variation in a small captive population is not easy. Small populations lose genetic diversity through both inbreeding and genetic drift. Population biologists have generally agreed that founder breeding group sizes of ten to 15 unrelated animals would represent 95 percent of the genetic diversity of the wild population. Why this is not the case with the elephant seal is a mystery.

In order to maintain at least 90 percent of the genetic diversity of this founder group over a 200 year period would require the captive population size to be raised quickly (Tangley, 1984). The target population size is dependent upon the generation time of the species. Species with a one year generation time such as side-blotch lizards (Uta stansburiana), would need a captive population of 1000 lizards. Species with longer generation times of 25 years, such as the Galapagos tortoise (Geochelone elephantopus), would only require that the captive population be maintained at 40 animals.

It is obvious that no one zoo or private breeder can maintain such large numbers of individuals in captivity. Breeding programs aimed at maintaining genetic diversity for successful reintroduction projects requires cooperation among the zoos and private breeders throughout the world.

A possible solution for not having to maintain large numbers of live animals is through cryopreservation (Rosenbaum, et al., 1985). Gametes can be kept in ultracold storage to maintain a reservoir for genetic diversity. There are some problems with decreased viability over time, but this technique is worthy of further investigation.

In the United States the American Association of Zoological Parks and Aquariums (AAZPA) has set up a program called the Species Survival Plan (SSP) to cooperatively manage rare or endangered zoo animals in North America. So far they have chosen to focus on 34 species and have developed at least preliminary breeding programs for seven of them.

The Puerto Rican crested toad (Bufo lemur) was the first amphibian selected for inclusion in the SSP. Approximately 75 toadlets were released in 1983 and another 800 in 1984 (Paine, 1985).

The AAZPA has set up general recommendations for the SSP program. A summary of these recommendations is listed below:

- Start captive population with ten to 15 individuals to insure 95 percent of the genetic diversity.
- Through breeding, the captive population should be expanded to the zoo's carrying capacity and then subdivide among different institutions. This will insure greater genetic diversity because different alleles randomly become lost or fixed in different populations. Dividing the animals also decreases the chance of losing the entire population to disease or disaster.
- Once the population is subdivided the sub-populations should be set up in a sex and age structure that can sustain reproduction and maintenance of maximum genetic variation. This usually includes equalizing sex ratios as well as the number of offspring per pair.

CONCLUSION

Captive propagation can play an important role in wildlife management. In some cases it may be the only way that we can prevent a species from becoming extinct. In extreme cases, it may be necessary to put all of the remaining animals in captivity, as with the California condor, but few species are at that crossroads yet. We must strive to maintain viable breeding populations in the wild. In their natural environment, with proper protection, breeding success is much more likely.

The single most important factor to insure a species survival is the preservation of habitat. Without habitat we can not maintain wild populations nor have areas to reintroduce captive bred animals. The prospect of only being left with zoo populations of many species of herps is distressing. We all need to work towards habitat protection to insure a biologically diverse world for our children.

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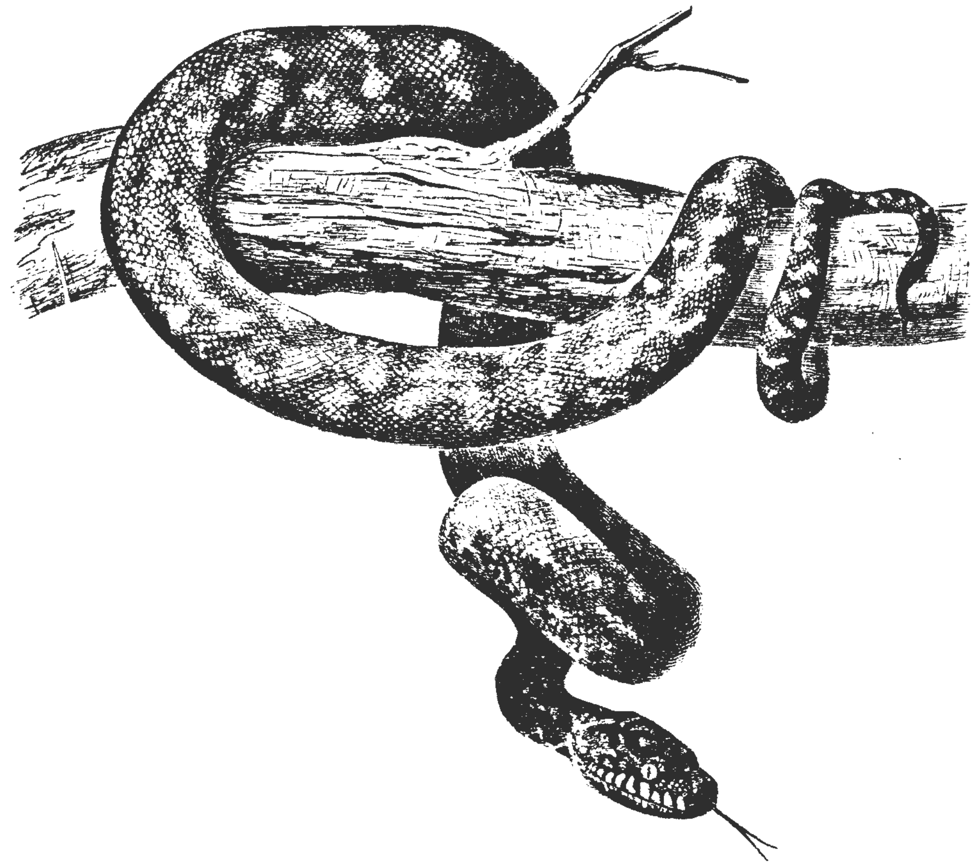

A HOLISTIC VIEW OF REPTILE PHYSIOLOGY

John A. Phillips

INTRODUCTION

The niche concept is a major tenet in biology. Defined as the "sum total of all the adaptations of an organismal unit" (Pianka, 1978), the niche of each individual, population, or species has been molded by the particulars of its environment. Although the niche has been described as being "n-dimensional" (Hutchison, 1957), the relationships between the organism and its environment can be divided into interactions with either the physical (abiotic) or biotic components. Thus, for each situation, there exists a triangular relationship between the organism, the physical environment, and the plants and other animals that organism encounters.

According to the theory or optimal choice, animals are efficient in all of their activities (Charnov, 1976; Cody, 1974; Emlen, 1966). Model systems have related optimization to diet, use of the environment, predator strategy, and reproduction. In all cases the organism attempts to maximize or minimize a particular quantity, goal, or in this case, niche dimension. The attainment of this goal is, however, constrained by all other dimensions -- the internal (physiological) and external (physical and biotic) limits particular to the system.

Zoological gardens (and others concerned with husbandry) often attempt to provide "optimum" conditions for captive animals. However, in doing so each species is denied the choice of optimizing any given situation in a natural manner. When animals are required to function in artificially "optimum" conditions, less than maximum physiological and behavioral responses often result (Phillips, 1984). As an example, the "preferred temperature" has been defined as the body temperature that reptiles select in a thermal gradient. Yet, most investigators would argue that few, if any, behavioral or physiological parameters are optimized at the preferred temperature. Instead, reptiles select environmental temperatures (the "normal activity range", Cowles and Bogert, 1944) that are most advantageous for the given situation and time.

Despite an increased awareness of environmental concerns, with relatively few exceptions, institutions concerned with captive propagation have only examined the niche dimensions that can be related to observable behavior, especially sexual behavior. These observations are usually restricted to interactions among conspecifics. Physical requirements, or rather the lack of those requirements, are only recognized and examined at necropsy. Even after necropsy, problems associated with environmental deficiencies are often not fully analyzed and are categorized as "stress-related" or "metabolic" death. Recently, Forthman-Quick (1984) examined the technologies now employed by zoos in the design of habitats for captive animals. Although this review stressed the role of niche elements as they related to "normal" behavior, little attention was given to the physical environment, especially as it relates to the physiology of the species. If successful reproduction is the bottom line, then examination of the environmental control of reproductive cycles is essential. Recently, my laboratory has been concerned with manipulating the physical and biotic environment for the benefit of reptilian species (Phillips, In press; Phillips, et al., In press), and several of these developments are reviewed in this paper.

FOCUS ON THE TOTAL ENVIRONMENT

The thrust of this presentation is to suggest that, along with naturalistic environments, we must give attention to the requirements for natural physiology and behavior. As with all organisms, reptiles evaluate the environment as a whole, not as a group of unrelated parameters. When compared with birds and mammals, reptiles appear to require a more simplistic environment, yet the subtle nuances of the simple environment often elude the investigator. There are numerous anecdotal accounts of reptiles (e.g., *Elaphe guttata*) breeding at will, even in "shoe boxes in closets", but these cases are exceptional.

The environment can be divided into five components: heat, light, diet, space, and interactions with other organisms. Space requirements and social interactions are often dependent upon one another, species-specific, and seasonally variable. Because information on spacing has been more than adequately reviewed elsewhere (see Stamps, 1977), the remainder of this discussion will only examine space and biotic interactions as they pertain to the requirements of heat, light, and diet.

HEAT

The pivotal investigation by Cowles and Bogert (1944) showed that reptiles actively control their body temperature by utilizing the thermal variation in the environment. Subsequent field and laboratory studies have shown that reptiles regulate body temperature through a number of physiological and behavioral adjustments. The thermoregulatory repertoire of terrestrial species includes basking, burrowing, shuttling, orientational and postural changes, variation in activity patterns, and various types of blood shunting (Heath, 1964, 1965; Muth, 1977; Templeton, 1970; Tucker, 1967). These mechanisms of body temperature regulation were originally described in lizards, but are applicable to other orders of reptiles.

Most reptiles select a high body temperature during the photophase and a cooler temperature during the scotophase (Regal, 1967). This rhythm appears to be controlled by a circadian timer, with the same body temperature relationships being found when reptiles are experimentally exposed to constant light or dark (Cowgell and Underwood, 1979). Several investigators have argued that this rhythmic cooling at night is probably a mechanism for conserving energy. The selected high and low temperatures are species-specific and are dependent upon the adaptations a species has made to a particular microhabitat (Brattstrom, 1965). There is an inverse relationship between the high selected temperatures and the intensity of the heat source (Garrick, 1979).

Most species attain a signficantly higher body temperature after ingestion, with digestion being hindered when sufficient external temperature is not available (Harlow et al., 1976). Less than optimum body temperature is also known to limit feeding behavior (Wilhoft, 1958), territorial defense (Bartholomew, 1977; Schoener and Schoener, 1980), learning, and fight versus flight rection (Hertz et al., 1982).

While it is apparent that reptiles modify body temperature by adjusting their positions in the thermal environment, it is also important to note that the behavioral state of a reptile can also modify the core temperature. As an example, Sceloporus occidentalis shows a marked increase in body temperature during aggressive interactions (Engbretson and Livezey, 1972).

The thermal environment also influences reptiles on an annual basis. High environmental tempertures stimulate spermatogenesis and follicular growth (Licht, 1965). Even the parthenogenetic Cnemidophorus show increased follicular development with high external temperature. Gravid females maintain lower body temperatures than nongravid counterparts (Garrick, 1974). Many species require a period of lowered environmental temperature for gonadal recrudescence to be initiated. Interestingly, and probably a result of the internal circadian clock, alternating high/low 24 h thermoperiods provide greater stimulation in experimental situations than do constant high or low temperatures.

WAVELENGTH CHARACTERISTICS

Animals have developed a number of physiological responses to solar radiation. Vision, photoperiodism, melanogenesis and vitamin D synthesis are some of the processes influenced by sunlight. These and subtle biochemical actions are modulated by the duration, intensity and spectral quality of light.

Light can be divided into three wavelength groups -- infrared, visible, and ultraviolet. Infrared wavelengths are recognized as heat (and not light) in most vertebrates. Depending upon the wavelength and skin pigmentation, 60-100% of the solar infrared spectrum is absorbed at the surface of the skin. The heat associated with these wavelengths is perceived by cutaneous receptors; however, in many reptiles the receptor system is not infallible, and overheating of the skin (often to the point of burning; Frye, 1981) can occur.

Most terrestrial animals receive and process more information from vision than any other sense. As Schmidt-Neilsen (1983) notes, the transmission of light is so fast that even subtle changes in the environment can be recognized almost instantaneously by the visual system. Visible light also controls skin color changes, induces daily and annual rhythms, and rejuvenates microorganisms that are injured by ultraviolet light.

It has been known for nearly 50 years that the ultraviolet (UV) spectrum is responsible for the antirichetic activity of sunlight (Bunker and Harris, 1937; Knudson and Beuford, 1938). More recent investigations have shown that exposure to UV irradiation results in the cutaneous production of cholecalciferol (vitamin D_3) (Quarterman, et al., 1964; Holick, et al., 1979; McLaughlin, et al., 1982). Subsequent hydroxylation of D_3 to an active form (1,25-dihydroxycholecalciferol) occurs in other organs of the body. The active compound then acts to increase absorption of calcium from the gut and also affects the mineralization of bone (Kobayashi, et al., 1976). Maintenance of blood levels of calcium and phosphorus is dependent on the interaction of 1,25-OHCC, circulating hormones, various target organs and dietary clacium and phosphorus. A deficiency of vitamin D_3 will upset the balance of this complex homeostatic mechanism and may result in rickets in the growing animal or osteomalacia in the adult (Anderson and Capen, 1976).

At the earth's surface, solar radiation essentially provides an ultraviolet (UV) spectrum of 290-400 mm (Folk, 1974). Studies have demonstrated that optimum production of vitamin D_3 occurs upon exposure to light in the range of 295-305 nm (Takada et al., 1979; Kobayashi, et al., 1976). Captive environments in which an animal is not exposed to unaltered sunlight are almost always devoid of UV light. Indoors, artificial lighting does not (except for special lamps) provide enough photic energy in the range appropriate for cutaneous photosynthesis of vitamin D_3 (Wurtman, 1975). Light provided by skylights is also of poor spectral quality because the materials used are formulated to absorb UV radiation while allowing transmission of visible light (400-700 nm).

DIET

The nutritional requirements of domestic animals and certain laboratory species have been the subject of numerous investigations. By comparison only a few conclusive studies have examined the nutritional problems of captive exotic species, with reptiles receiving the least attention.

The manager of captive exotic species is always faced with the dilemma of feeding a "natural" diet or a nutritionally complete diet. If zoos (and other institutions) cannot provide a truly natural diet, physiologically complete rations should be the rule. With the exception of those species that eat live (or freshly killed) prey the reasons for inadequate natural diets are many. First, in the wild, animals eat "dirty" food items. Both vegetable and animal matter are constantly dusted with the minerals and micronutrients found in the soil. As an example, when a carnivorous lizard eats an insect or rodent, the prey item will inevitably collect and retain a moderate amount of substrate material during feeding. By comparison, captive reptiles are generally fed clean food items, be they insects, rodents, or vegetables. Dusting food items with vitamins and minerals for captive reptiles is a common practice. Unfortunately, while the dusting may provide adequate amounts of certain vitamins/minerals/micronutrients, these additives are not balanced for the food items that are commonly dusted. As an example, because the Ca/P ratio is different in each of the common insect food sources, dusting with an "average" powder will only modify the Ca/P ratio in each prey, and not necessarily balance that ratio. Another problem associated with captive reptiles is food preference. Although a balanced meal may be provided to the animal, many reptiles will preferentially choose one food item over all others, and again receive an incomplete diet. Finally, most diets are difficult to force feed to a reptile should that animal become ill. Most of these problems do not occur in captive snakes because the rodents fed to the snakes have been fed a complete diet and therefore provide a nutritionally balanced diet. Lizards and turtles are not as fortunate.

We have recently developed an artificial diet that has the potential for eliminating these problems (Phillips et al., In press). The diet has a beef heart base and all minerals and vitamins are added to provide a physiologically complete and balanced meal (see Table 1). Because the vitamin and mineral content of the diet can be manipulated at will, the specific needs of a given animal can be accommodated. For example, the Ca/P ratio can be increased to 1.2/1 (or higher) for juvenile reptiles. In addition, because the diet can be forced through a standard syringe, sick animals can be maintained on the diet for extended periods without complications.

THE TOTAL ENVIRONMENT IN CAPTIVITY

Because the physiology and ecology of each species has resulted from the particulars of specific microhabitats the manager of captive animals must attempt to provide as many of those conditions as possible. The preceding review of heat, light, and diet has attempted to show that there is a commonality of need in all species, even though any given species may thrive in conditions that would not be considered "average".

Heat is one of the most basic needs of all terrestrial vertebrates, yet heating systems in animal enclosures usually

TABLE 1

Beef Heart Based Diet for Lizards

Analysis: 22.1% Dry Matter; 5.40 kcal/g

Ingredient	Quantity	
Fresh, lean beef heart	500	g
$MnSO_4 \cdot H_2O$	19.1	mg
KIO_3	38	ug
Folic Acid	135	ug
L-ascorbic acid	50	mg
$Ca\ (H_2PO_4)_2 \cdot H_2O$	610	mg
$Ca\ CO_3$	8	g
Glucose monohydrate	1.12	g
Corn oil	2.25	g
Vitamin D_3	22.5	ICU
A-tocopherol acetate	4.5	IU
Vitamin A palmitate	135	IU
Ethoxyquin	13.5	mg

Recipe method can be found in Phillips et al., In press, Zoo Biology.

employ components that were originally designed for the physiological requirements of humans. Conventional methods for heating reptile enclosures often use either infrared heat lamps or several forms of central heating. Each method has its disadvantages. Heat lamps are not particularly energy efficient in that the heat generated dissipates into and is carried away by the circulating air. Heat lamps are also very hot at their surface, can cause burns, and at the very least will dry the skin of the animal. Heat lamps also provide visible light. If the lamps are the only source of heat in the enclosure and are therefore on during the cooler night hours, the natural light-dark cycle will be destroyed. Because light-dark cycles often contribute to the reproductive competency of terrestrial animals, this basic biological rhythm can be altered. Central heating, although superior to heat lamps in a biological sense, is usually complex and requires permanent installation. Central heating also does not offer the animal a choice of environmental temperatures.

Heat should be provided in two locations within each enclosure. Most terrestrial reptiles (with the exception of certain large snakes and tortoises) have a retreat within their home range. Because this retreat is dark, a thigmothermal (ground) heat source is most appropriate. (Any of a number of heat sources, hot rocks, etc., are adequate.) The temperature of this retreat should be far less than the temperature of the basking site. For most reptiles, a temperature of between 65-75^{o}F would be appropriate. The temperature provided in the basking area should be much higher (100-105^{o}F) If the species is strongly heliothermic (like many iguanids), then the basking heat source should be an incandescent lamp. Outdoor flood lamps (100-150w) are superior for basking areas. Not only does the lamp provide infrared heat, but the entire visible spectrum is represented (see Figure 1). A temperature of around 100^{o}F will allow the animal to adjust its daily temperature rhythm. To minimize disruption of the 24hr biological rhythms, the basking heat source should be coupled with the photoperiodic lights.

If reptiles are maintained in indoor enclosures throughout the year, one of two types of photoperiods should be be applied. If the species is from a tropical location (between $20^{o}N - 20^{o}S$ latitude), then the photoperiod should approximate that found in nature (about 11-13 hrs of light per day). Many tropical species have difficulty with the natural photoperiod of California (a photoperiod that fluctuates from 9 to 15 hrs of light per day). However, most of tropical species will eventually entrain to a novel photoperiod, especially if the animals were juveniles when first exposed to the new conditions. If the species is native to the northern hemisphere, then a natural photoperiod should be employed.

More important than the photoperiod per se, is the quality of light obtained during the photophase. As mentioned previously, infrared, visible and ultraviolet wavelenghts make up the solar spectrum. For the purpose of raising reptiles in captivity, strictly infrared sources can be eliminated from the discussion. Infrared sources have more problems than benefits, and for the most part, heat can be obtained from much safer and more biologically correct sources (as noted above).

If animals are maintained in outside enclosures, where the photoperiod is natural, ultraviolet light, and in some cases even visible light, will not be available when certain structural materials are used (see Figure 2). By far the most superior greenhouse material is that manufactured by Rohm and Haas, Plexiglas G-UVT.

Many investigators believe that their animals do fine without ultraviolet wavelenghts. If the proper amount of vitamin D_3 and calcium are in the diet then the animal will probably do well in a nutritional sense without additional UV light added. Yet, in many animals, the UV spectrum also influences the animal in other ways (see Miller, 1984). That being the case, providing as many environmental components as possible, is probably the best rule.

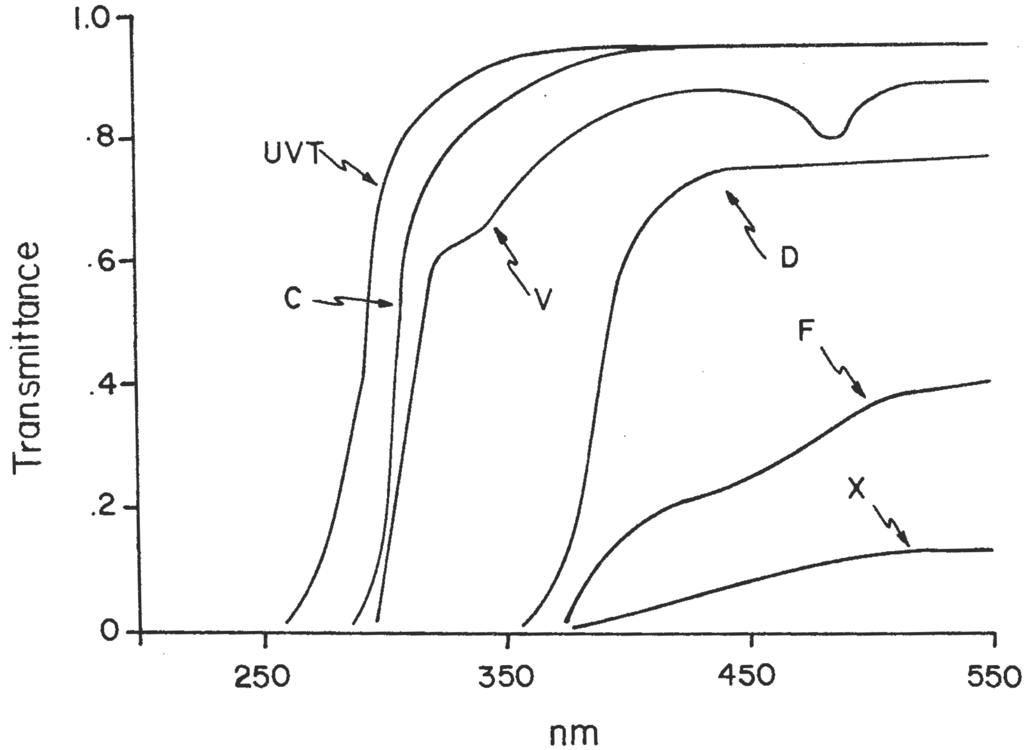

Figure 1. Spectral output (microwaves/cm) of lamps measured at a distance of 0.5m. Abbreviations: SS, summer sun; FS, Westinghouse FS-40 ultraviolet tube; FS/cta+, FS-40 tube with cellulose triacetate filter; 150, 150 watt Westinghouse outdoor flood lamp; BL, GE BL-40 blacklight tube; C50, GE C50 5000^{o}k florescent tube; IR, GE 250 watt infared heat lamp.

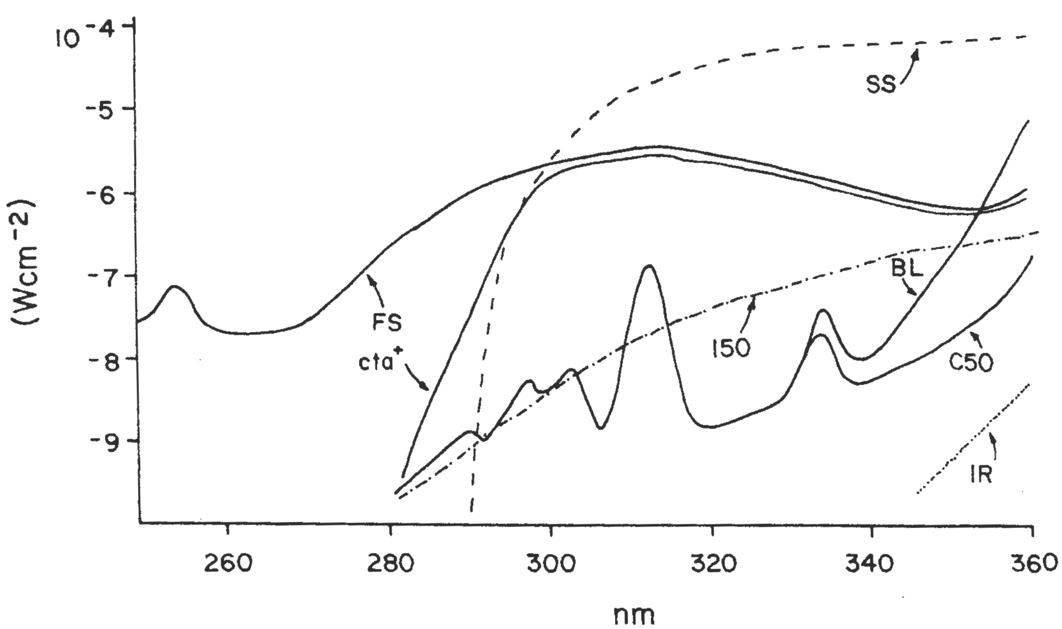

Figure 2. Percent transmission of natural sunlight through different plastics. Abbreviations: UVT, Plexiglass type G-UVT 0.25"; C, Cellulose triacetate 0.005 ; V, vinyl 0.01"; D, Tuffik acrylic double wall; F, greenhouse fiberglass roll; X, Xcelite double skin 0.25".

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THE INTERRELATIONSHIP BETWEEN AMBIENT TEMPERATURE AND REPTILE HEALTH MANAGEMENT

Douglas R. Mader

INTRODUCTION

It is common practice among reptile owners to increase the ambient temperature in the cage of an ailing animal. Unfortunately, most do not understand the physiological basis for this method of therapy and, consequently, do not know when it is adequate as the sole means of treatment. This manuscript will address the various physiological effects of increased ambient and, hence, body temperature as it pertains to reptile health management.

Hypothermia, or cooling the reptile below the temperature range of normal activity, as it is used to physiologically manipulate the animals for surgery or other invasive procedures will also be reviewed.

EFFECTS OF ELEVATED AMBIENT TEMPERATURE

An increase in body temperature, or fever as it is referred to in mammals, has been noted to be beneficial to the health of the individual as far back as 2500 years ago. Hippocrates claimed that sickness was due to imbalances in the bodily "humors," and that fever was able to "cook out" these offending humors thus helping the host overcome the disease (Kluger, 1979a). Patients were treated with "fever causing substances" as a method of treatment for such ailments as syphilis, tuberculosis, and cancer (Kiester, 1984).

Although reptiles, which are ectotherms - that is, animals whose body temperature depends directly upon the ambient temperature or environmental factors such as the sun - are incapable of developing a fever response as can mammals, they can develop a fever behaviorally when exposed to certain pyrogens. Vaughn et al. (1974) suggested that the development of this behavioral response dates back to the late Paleozoic, early Mesozoic period, antedating the development of fever in mammals as it is known today.

Fever in mammals, or its equivalent in reptiles, results when the thermoregulatory "set point" becomes elevated. Kiester (1984) notes that the elevated "set point" is due to a response to a triggering agent such as a bacteria or virus. After the arrival of the foreign substance in the body the macrophages release a hormone called an endogenous pyrogen (EP). The EP acts directly on a region in the brain called the hypothalamus, the portion of the brain generally thought to be responsible for thermal regulation (Kluger, 1979b).

In mammals this increase in "set point" is manifested by increasing metabolic heat production (shivering), surface vasoconstriction and behavioral means such as huddling or, in the case of humans, putting on extra clothes or drinking some warm liquid. Reptiles are generally incapable of increasing their own body temperature by physiological means. An exception to this are a few species of pythons which can increase their body temperature by rhythmically contracting their musculature while incubating their eggs. Reptiles otherwise depend entirely on behavioral means of thermoregulation.

This was demonstrated by Vaughn et al. (1974). Vaughn placed specimens of <u>Dipsosaurus dorsalis</u>, the desert iguana, in cages which had a temperature gradient ranging from 30 degrees to 50 degrees C. By means of thermocouples she could keep a running record of their body temperature. The lizards selected a place in the cage which yielded an average core body temperature of $38.5^{o}C$. The animals were then injected with either a solution containing <u>Aeromonas hydrophilia</u>, a common reptilian pathogen, or sterile saline as a control.

The saline injected control lizards failed to show an increase in body temperature. However, the lizards injected with a bacterial containing solution consistently chose a temperature which was $2^{o}C$ warmer than non-infected lizards. The lizards began to seek the warmer temperatures within four to six hours post-infection.

Vaughn also noted that lizards innoculated with the pathogen failed to demonstrate an increase in core temperature if the environmental temperature remained constant. This reinforced the hypothesis that reptiles cannot generate their own internal heat to raise their body temperature.

Kluger et al. (1975) furthered these studies by placing Dipsosaurus dorsali infected with A. hydrophilia at different temperatures and recording their survival. By three and one-half days all the lizards housed at 34°C were dead. Below is a table listing the results:

	Percent Mortality Rate	
Temp °C	Infected	Control
42	25%	4%
40	33	0
36 - 38	75	0
34	100	-

The increased survival at the higher temperatures supports the hypothesis that fever augments some physiological response and is beneficial to the host (Kluger et al., 1975). Since there were approximately an equal number of mortalities in both the infected and control group at 42°C Kluger believed that the deaths were not entirely the result of infection but rather an undetermined adverse effect of long term elevation in temperature.

From the work by Kluger, Vaughn and others it became apparent that increasing the body temperature was an asset in treating disease. The methods by which fever augments this process have yet to be fully elucidated.

Evans (1963) studied the antibody response of Dipsosaurus dorsalis at different temperatures. He infected the lizards with Salmonella typhosa. At various intervals post-immunization agglutination titers were run to determine the antibody response. He found that the antibody response was poor to non-existent at 25°C, good at 35°C, and moderately good at 40°C. He also reported that if the lizards were immunized at 35°C and then transferred to 25°C the antibody response was inhibited, but if moved from 35°C to 40°C after immunization the antibody response was enhanced. Similar effects of the enhancement of antibody response by elevated temperatures have been reported (Elkan, 1975; Marcus, 1981; Frye, 1982; Kiester, 1984; and Kollias, 1985).

EPs have facilitating effects on other aspects of the reptile's defense system aside from immunostimulation. Kluger (1979b) studied the effects of infection and EP release on serum iron. It is known that during infection in mammals serum iron falls due to a release of EP (Merriam et al., 1977; cited in Kluger, 1979b). The iron, which normally comes from red blood cell turnover, is sequestered in the liver and spleen until the infection is over.

Bacteria uses the iron as a growth factor. It does this by producing a chelating substance known as siderophore. Garibaldi (1972) has shown that the ability of the bacteria to produce these siderophores is diminished at elevated temperatures (which are species-specific for each bacteria). This fact, coupled with the drop in serum iron by the host, makes growth and continued propagation difficult for the bacteria.

Grieger et al. (1978) subjected uninfected D. dorsalis to febrile and afebrile high temperature environments. Neither group showed a decrease in serum iron. After infection with A. hydrophilia serum iron fell irrespective of the environmental temperatures at which they were housed. This finding suggested that the fall in serum iron was more a factor of the resonse to EP than an elevated ambient temperature. It was also noted that if supplemental iron was given to the infected iguanas there was a higher percent mortality compared to those not supplemented.

Leukocyte activation, including increasing phagocyte, bactericidal and viricidal activity, leukocyte mobilization, and augmented production of immunologically active T-cells, are all factors which are increased by the release of EP (Kiester, 1984). EP also stimulates interferon an antiviral agent elaborated from the host's cells.

Elevated temperature, in and of itself, has a direct effect on viral and bacterial kinetics. Studies done on viral activity in the pig, ferret and puppy show that the virulence of viruses is attenuated at elevated body temperatures (Kiester, 1984). Kluger (1979b) notes in his paper that studies done with the poliomyelitis virus show a 250 times greater yield when grown in culture at 37^{o}C as compared to 40^{o}C. Kluger (1975) measured growth rate of A. hydrophilia at five different temperatures: 34, 36, 38, 40, and 42^{o}C. The doubling time was approximately equal from 34 to 40^{o}C, but at 42^{o}C the bacteria grew markedly slower.

Combining the findings of the aforementioned experiments, it is apparent that a combination of factors due to an elevated body temperature work synergistically to help fight infection. The augmented antibody response, the increase in leukocyte activation, the stimulation of interferon and lysosomes, the decrease in serum iron and the decreased effectiveness of the bacterial siderophores combined with the actual decreased growth and replication of the bacteria and viruses represent a coordinated effort by the host to overcome disease.

TEMPERATURE EFFECT ON ANTIBIOTIC THERAPY

Now that the effects of elevated temperature on the host's defense system have been reviewed, the effects of increased temperature on treatment will be discussed.

Amikacin is an aminoglycoside antibiotic in the same class as Gentocin, a drug commonly used to treat ill reptiles. Amikacin, however, possesses some attributes which in some cases make it a more appropriate drug of choice. Since it is likely to come into popular use, its value should be mentioned here.

Amikacin responses in gopher snakes (Pituophis melanoleucas) were noted at two different temperatures, 25^{o}C and 37^{o}

C (Mader et al., 1985). There were three major advantages of treating the snakes at the higher temperature.

First, the drug achieved a greater volume of distribution (i.e., greater quantities of drug gets into the tissues where it is needed to fight infection). Secondly, the rate at which Amikacin was cleared from the body was two times greater at the higher temperature, a factor which may be significant when dealing with potentially nephrotoxic drugs.

Thirteen reptile pathogens, which were isolated from clinical cases at the Veterinary Medical Teaching Hospital at the University of California, Davis, were used to test for the mean inhibitory concentration (MIC) of Amikacin at both $25^{\circ}C$ and $37^{\circ}C$ <u>in vitro</u>. This brings up the third reason to treat at the higher temperature. There was a significant difference (p 0.005) between the two tests, with the bacteria tested at the higher temperature being twice as sensitive (i.e., only needing half as much drug to inhibit growth).

This increased sensitivity, combined with the greater volume of distribution at the elevated temperature, suggests that raising the snake's body temperature during treatment should increase the efficacy of treatment.

METHODS OF HEATING THE ANIMALS

Various methods are currently employed to heat cages and artificial environments. Following is a survey list of some of these:

Heat tapes -

Examples would be roof de-icers and plumbers heat tape for pipes. These can be quite effective. They need to be connected to a rheostat to control their output. Always remember to only heat part of the cage to allow for a thermal gradient.

Hot rocks -

Various commercial brands are adequate for some of the smaller species. They must be checked to ensure that they don't get too hot.

Heat lamps -

Great for basking animals. They can be connected to a timer which will allow for a pre-determined daily exposure. Always check to make sure that the animals cannot come into contact or get too close to the bulb because serious burns and skin sloughing can result. (Note: Do NOT use "Sun Lamps", they are not the same thing!)

Aquarium heater in a gallon jar of water placed in cage -
These can be adjusted accurately to within a few degrees. You need to be careful that the heater goes all the way down to the bottom of the jar so that there is no dead space where the water will remain cool.

It is recommended that all heating elements be wired to a thermostat. This will ensure that the temperature never reaches the animal's critical high, a point where tissue injury or neural damage could result. Elaborate systems can be designed where timers and thermostats can be hooked up in the same circuit, thus allowing temperture fluctuations such as daytime highs and nighttime lows.

It is critical to remember that the ideal environment would be one with a built-in temperature gradient, i.e., cooler on one side gradually increasing in temperature to a warm spot on the opposite side. It is possible, especially for arboreal animals, for a vertical temperature gradient to be created, one cooler near the floor of the cage, and warmer near the top.

The question arises as to when is the best time to raise an animal's temperature when it is suspected to be ill. In properly maintained terraria with adequate temperature gradients no adjustments are needed. Vaughn (1974) demonstrated that his lizards consistently chose a warmer temperature by four to six hours post-infection with the bacteria. If an appropriate gradient is maintained, the animal should adjust its own temperature well before any symptoms are noted by the caretaker.

However, if adequate temperature gradient is not available and the reptile becomes ill, placing the animal into an environment with the appropriate temperature is recommended.

It will take anywhere from several hours to a week for the core temperature to equilibrate with the ambient temperature depending on the girth and weight of the animal.

It would be virtually impossible to list what the proper temperature is for all 5000 plus reptiles. It is recommended that reptile owners pay close attention to their animal(s) and learn all there is to know about them. This might entail doing library research, seeking professional veterinary or herpetological advice, or learning about the animal's natural environment. If these sources of information are unavailable then ensure that the animal has a broad temperature gradient, one which covers a large area, so that the animal can easily choose the preferred optimum.

HYPOTHERMIA AS A MEANS OF REPTILE ANESTHESIA

Hypothermia has been used as a means of inducing surgical anesthesia in reptiles (see Wallach, 1977). This is an extremely poor practice, however, and should be discontinued for two reasons. First, even though the animal's movements may cease, there is no evidence that there is adequate analgesia, i.e., that the patient <u>can still feel pain</u>. Secondly, the patient's immune or defense system becomes compromised due to the hypothermia. Since any invasive procedure carries a high risk of infection, this practice is contraindicated.

There are many alternatives to hypothermic anesthesia. Chemical anesthesia, as with Ketamine HCl, is one such example. This method has disadvantages as well in that it often take two to three days for the drug to be cleared from the system due to the slow reptilian metabolism.

The best and most precise method is inhalent anesthesia such as Metofane and Halothane, which can be administered via an induction chamber or by an anesthetic machine. The latter is preferred since exact quantities of the agent can be delivered to the patient via an endotracheal tube allowing accurate control over the depth of anesthesia.

It is extremely difficult to monitor the depth of anesthesia of the reptilian patient when it is left in the refrigerator. However, reptile medicine has advanced markedly over the past few years, and various methods of patient monitoring are now available for use during properly administered anesthesia. Dopplers and Electrocardiographs (E.K.G.) are two examples.

SUMMARY

It should be stressed that nobody should know an animal as well as the keeper. It is therefore imperative that caretakers spend time with their animals and get to know each one individually. What is normal for one may not be for another even with two animals of the same species.

To maintain a healthy colony and minimize health problems one must notice an animal when it exhibits the first signs of illness. This can't be determined unless what is normal for that particular animal is known to begin with. It is important to keep in mind that not all ailments will respond simply to warming the animal. It is highly recommended that caretakers seek professional advice freom a herpetologist or a veterinarian specializing in reptiles when they suspect a health problem.

Careful consideration of the reptile as an individual and knowing as much as possible about the species, its preferred temperature range, its diet, etc., and when to seek professional help will insure a healthy, productive collection, whether for personal enjoyment or commercial purposes.

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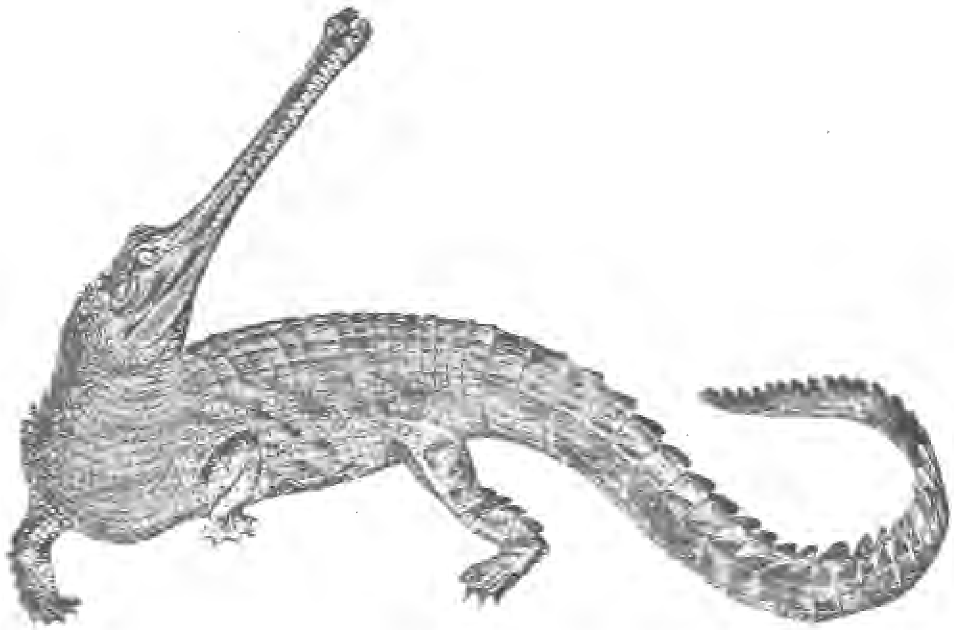
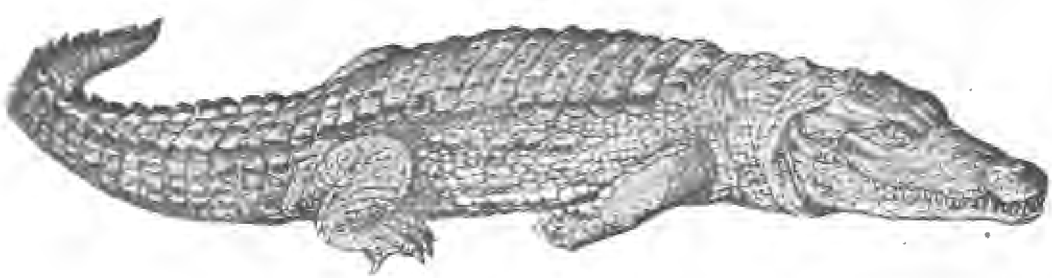
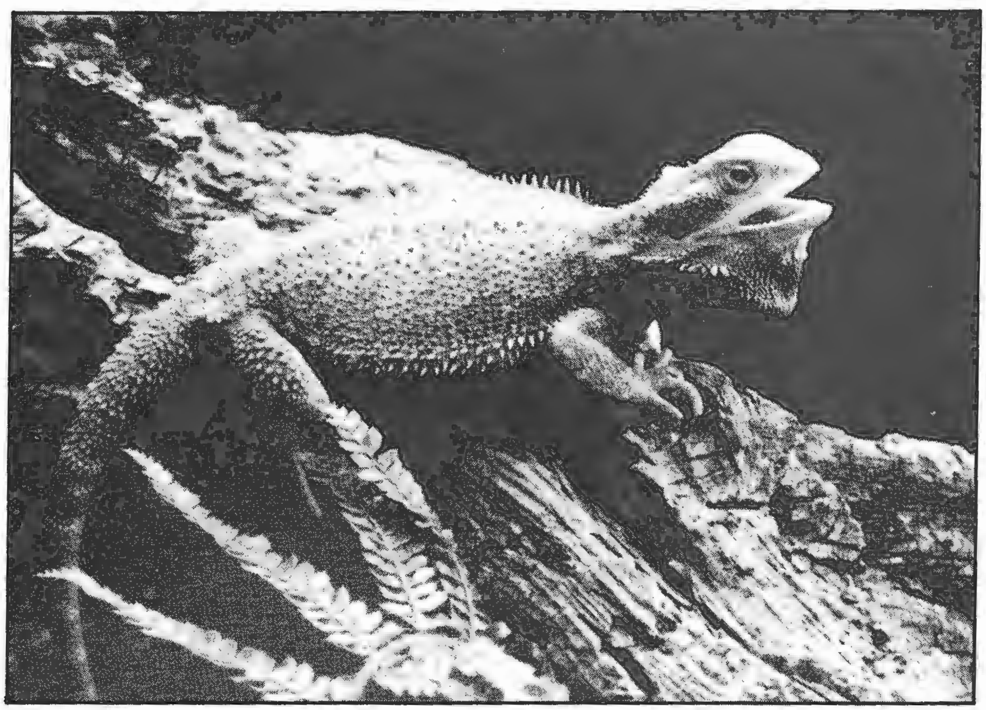




Shingleback skink (Trachydosaurus rugosus asper). Photo by D. McCampbell.



Bearded dragon (Amphibolurus vitticeps). Photo by D. McCampbell.

AN OVERVIEW OF LIZARD HUSBANDRY AND PROPAGATION WITH EMPHASIS ON THE WORK AT THE REPTILE BREEDING FOUNDATION

Thomas A. Huff

Although there is some disparity among taxonomists (Camp, 1971; Grizimek, 1971; Romer, 1956) on the placement of certain families within the suborder Sauria, most are in agreement that there are a total of 22 families (Table 1). Of these, only eleven, or one half of the currently recognized families, are commonly kept in captivity: the Gekkonidae, Iguanidae, Agamidae, Chamaeleontidae, Scincidae, Cordylidae, Teiidae, Lacertidae, Anguidae, Helodermatidae, and the Varanidae.

A search of the International Zoo Yearbooks for the past 24 years shows that up until 1971, the only species bred with anything close to regularity in zoos were: the common basilisk, tokay gecko, and the leopard gecko. The rhinocerous iguana and the Madagascar day gecko were also bred by several institutions before 1972. In 1971, eight institutions reproduced Eublepharis macularius; the first year for which there were multiple successes with a single species. Not to downplay the excellent success based on good husbandry by some institutions, from 1959 until 1975, most of the lizards bred in captivity were either a fluke, were gravid upon arrival (and therefore not a captive propagation), or they were bred in semi-captive environments. This last group includes many of the agamids and gekkonids bred at Tel Aviv University in Israel, reproduction of some desert sceloporids in Arizona, breeding of lacertids in European gardens and zoos, and the reproduction of numerous iguanid species in Mexican zoos. These reproductive successes were achieved by housing large colonies of the respective species in outdoor enclosures, where they had access to natural sunlight and wild insects.

It was my intention to show the notable progress which had been made in captive lizard propagation after 1971; however, the results have not been all that notable. It is true

Table 1: Lizard Taxonomy

Order: Squamata
- Suborder: Sauria
  - Infraorder: Gekkota
    - Family: Gekkonidae
    - Family: Pygopodidae
    - Family: Dibamidae*
    - Family: Iguanidae
      - Subfamily: Sceloporinae
      - Subfamily: Tropidurinae
      - Subfamily: Iguaninae
      - Subfamily: Basiliscinae
      - Subfamily: Anolinae
    - Family: Agamidae
    - Family: Chamaeleontidae
  - Infraorder: Scincomorpha
    - Family: Scincidae
      - Subfamily: Tiliquinae
      - Subfamily: Scincinae
      - Subfamily: Lygosominae
    - Family: Feyliniidae
    - Family: Anelytropsidae
    - Family: Cordylidae
      - Subfamily: Cordylinae
      - Subfamily: Gerrhosaurinae
    - Family: Xantusiidae
    - Family: Teiidae
    - Family: Lacertidae
  - Infraorder: Anguimorpha
    - Family: Anguidae
      - Subfamily: Diploglossinae
      - Subfamily: Gerrhonotinae
      - Subfamily: Anguinae
    - Family: Anniellidae
    - Family: Xenosauridae
  - Infraorder: Varanomorpha
    - Family: Helodermatidae
    - Family: Varanidae
    - Family: Lanthanotidae
  - Infraorder: Amphisbaenia
    - Family: Bipedidae
    - Family: Amphisbaenidae
    - Family: Trogonophidae

*There is perhaps more disagreement about the placement of this family than any other. Some taxonomists believe that dibamids belong with the skinks and others have placed them with the cordylids.

that more species of more famlies were captively reproduced in the next ten years, but there were more institutions reporting, more specimens in captivity, and a greater variety of lizard families imported. The basic reproduction rate has really remained quite low. Although difficult to calculate, I would estimate that no more than one or two percent of the lizards maintained in captivity have actually reproduced. This is unfair to the few active private herpetoculturists who have done much better, and who have brought the overall average even that high. At the Reptile Breeding Foundation, we have successfully reproduced approximately 65 percent of our lizard specimens. I am pleased with this figure when based on the overall global picture, but I know it should be higher.

The literature on captive reproduction of lizards has been scarce and is noticeably minimal when compared to the articles on the other reptile suborders. A search of the papers submitted to the International Zoo Yearbook and those presented at Symposia in North America, Europe, Australia, and Japan reveals only a few papers on captive reproduction of lizards. It is interesting to note that there are some very definite trends in popularity in the saurian families kept in different countries. In North America, the gekkonids far outrank any of the other families in popularity as captives. The iguanids, and scincids probably rank second and third. In Europe, Great Britain, and Western Asia, the lacertids hold the top position with the agamids second, and the gekkonids third. In Australia the scincids rank first, with the varanids and gekkonids next in popularity.

The First Annual Reptile Symposium on Captive Propagation and Husbandry was held in July 1976. At that symposium there were seventeen papers presented; only two of which dealt specifically with lizards. That trend has continued for the last six meetings. Of the seven symposia held in this series, there have been a total of 147 papers presented only 27 of which dealt primarily with lizards. This has been the case with most of the meetings in North America dealing with captive reptile propagation and husbandry (one notable exception was the Northern California Herpetological Society Conference last year, where out of a total of twelve papers presented, five dealt expressly with lizards). In 1978 the Society for the Study of Amphibians and Reptiles (SSAR) held a symposium on reproductive biology and diseases of captive reptiles in Tempe, Arizona. There were a total of 30 papers, with only two dealing specifically with lizards. It should be pointed out that in all of these symposia there were many papers presented which dealt with reptiles in general, and they may have included information which would be of use in the husbandry and propagation of lizards, but they most often dealt with information relative to the keeping of snakes in captivity.

In North America, this trend has been the norm for articles in newsletters and regional journals, as well as other symposia and conferences. The percentage is not as low in Europe and other parts of the world, where lizards seem to be on an almost even par with snakes in their popularity with herpetoculturists. This trend is also reflected in the public reptile collections in North America. When one looks at those forms maintained in captivity, there are relatively few species for which we have accumulated data. It would appear that zoos in particular, and private herpetoculturists in general prefer snakes, turtles, and crocodilians to the lizards. It is really only the larger, more impressive or unique species of lizards which have been commonly kept in captivity, such as the helodermatids, the chamaeleontids, the more colorful gekkonids and the larger iguanids and varanids. Small unimpressive forms, many of the nocturnal forms and the fossorial species are seldom kept by herpetoculturists. But, it is disturbing that most of the lizards kept in public collections have shown very poor reproductive success.

Most major public collections display one or both species of Heloderma, and usually in large numbers. The Inventory of Reptiles & Amphibians in Captivity, 1984 (Slavens, 1984), records 30 institutions or private collectors housing a total of 80 specimens of Heloderma horridum, and 48 collections housing 202 specimens of Heloderma suspectum. Of these collections reporting, only two clutches totaling five neonates of Heloderma suspectum were reported as hatching during that reporting period. This is a deplorably poor reproductive performance. Likewise the chamaeleons, many of the agamids and the varanids have been commonly imported, but have a very poor record of both reproduction and longevity in captivity, only about 50 percent of the lizard species maintained have reproduced. This percentage plummets if we count the number of specimens kept of these species and compare it with the number of specimens reproduced.

The reason for the lack of popularity of lizards by private individuals, as compared to snakes, is not known. Perhaps it is just that as with the general zoo visitor, these herpetoculturists are more interested in the large and more impressive forms. It may be somewhat more a financial and maintenance consideration with the private individual than it is with the public institution, since lizards, generally speaking, require more time and greater expenditures for food than do snakes. To properly provide insectivorous lizards with a varied diet, a number of different insect species must be provided. They are expensive to purchase, and both time consuming and messy to raise. Snake keepers can often find a free or relatively inexpensive source of rodents. Carnivorous and herbivorous lizards do not present the same problems, although certain genera such as the Phelsuma, demand a fair bit of work and expense in preparing the artificial nectar preparations which they require.

REPTILE BREEDING FOUNDATION

Although the Reptile Breeding Foundation established itself in the herpetocultural community by propagating many of the insular forms of Epicrates in captivity (Huff, 76, 77b, 78a, 78b, 79a, 79b, 80), in recent years we have placed more of a focus on various lizard species. There were several reasons for this. First, we felt that very few institutions were working with lizards, and many were working with boids. Second, after many years of working with live-bearing reptiles, the opportunity to work with some egg-laying forms, and the challenges that accompanies them was very appealing. Third, it offered new challenges such as rearing a variety of insects, designing new caging, and creating a familiarity with a group of reptiles of which we were relatively ignorant. Fourth, and of lesser importance, was the fact that we felt we could house greater numbers of small lizards than we could boas and pythons. This held true until we established large groups of Varanus bengalensis and Cyclura cornuta. I would like to add that we have not terminated our work with the Epicrates, but we certainly lessened our involvement with that group. We have also increased our collection of turtles and tortoises and colubrid snakes.

We started with what are normally considered the more common geckos in an effort to learn something about this family in general. We established colonies of Ptychozoon lionatum, Eublepharus macularius, Platyurus platyurus, Gekko monarchus, and others, and immediately had great success in reproducing, hatching and raising the young. Admittedly, most of these reproduced with very little assistance from us, but we did learn things that we were later able to apply to other species and other families. Once we felt confident in our knowledge of these lizards, we obtained some of the rarer and more unusual forms. Here too we achieved success in our reproduction programs and we learned that the information acquired from the commoner forms was applicable to these other more obscure species.

In recent years the gekkonidae have been popular with a small group of herpetoculturists. The most popular genera appear to be Eublepharis and Phelsuma. We too are involved with both of these genera, and are pleased with our success with some of the rarer Phelsuma. Our reproductive success with Phelsuma guentheri, the Round Island Day Gecko, is second only to that of the Jersey Wildlife Preservation Trust, from whom we received our initial animals (2.2) in 1982. We have also had fair success with the dwarf form of Phelsuma sundbergi from LaDigue Island, and with Phelsuma longisulae pulchra and Phelsuma laticauda. We have also established a very successful colony of Teratoscincus scincus and are in the process of establishing colonies of Teratoscincus przewalskii and Chondrodactylus angulifer. Additionally, we continue to maintain breeding colonies of some of the other gekkonids. We have just established new groups of Cyclura cornuta and Varanus bengalensis which will be our first endeavors with the larger iguanids and varanids, and which we are hopeful of breeding this year. Other lizards which we are working with include the skinks, with groups of Scincus scincus and Egernia frerei, the anguids with Diploglossus warreni and Ophisaurus apodus, and the cordylids with Cordylus warreni depressus, Cordylus warreni breyeri, Cordylus cataphractus, Cordylus jonesi, Cordylus giganteus and Pseudocordylus microlepidotis. We also have a group of male Gerrhosaurus validus which we hope to pair with females, and then breed these impressive animals.

In establishing these large numbers of insectiverous lizards, it has been necessary to also establish a sufficient and reliable source of food for them. We have always raised rodents, but we needed to start reproducing a variety of insects in addition to this. At the present time, we maintain breeding colonies of mealworms, crickets and wax worms. There have been many other changes in our overall operation with the addition of such a large lizard inventory. Many of these species are basking forms, so heat lamps, black lights, vita-lites, hot rocks, etc. needed to be provided, with a substantial increase in our electrical rates. We also hired one additional staff member just to look after the planting and design of the Phelsuma terraria and the care of these enchanting lizards.

GENERAL INFORMATION

A bibliography follows this paper which includes some of the major papers on lizard husbandry and captive propagation, for those families for which we have information.

Undoubtedly the iguanids have been studied more in the field than any of the other suarian families; the gekkonids place a distant second, and yet because of their size and diversity, there are many members of these families which we know very little about.

Because of the diversity of species, and the unique characteristics of many of these species, it is often necessary to speak in generalities. Please accept that many of the statements I am about to make cannot be held accountable for every species, but that they are important comments to be considered in the keeping of saurians in captivity, especially if your goal is the reproduction of those animals and the production of healthy and viable neonates.

Some of the critical requirements in lizard husbandry include proper diet, environment, temperature, and for the diurnal species, a good source of ultra-violet light. Although all of these things are important for all species of reptiles, in the case of the lizards, these requirements must meet a narrower spectrum of exactitude.

As has been seen in the snakes, crocodiles and testudinata, diets can consist of fairly basic and easily obtained items: rodents, vegetables and meat. With these reptiles and their food items, it is relatively easy to supplement with vitamins and minerals.

The insectivorous lizards must be fed a fairly wide variety of insects if we are to insure their good health in captivity. Insects and the other invertebrates inhabit a wide variety of environments and ecological niches. As a result they feed on a tremendous variety of foods and their chemical make-up is undoubtedly diverse. The techniques for raising insects in captivity as a food source are poorly known except for a very few species. Crickets, mealworms, wax worms, cockroaches, flies and locusts are basically the only groups of insects which we can produce in the quantities needed to feed a captive insectivorous lizard. This does not take into consideration the numerous and varied invertebrate forms that these lizards may consume in the wild.

A rodent is a rodent is a rodent. This, of course, is an oversimplification, but all rodents eat basically the same type of diet: grasses, seeds, bark, and the occasional invertebrate. Commercial laboratory chows have been perfected over a number of years (thanks to the fact that rodents make such deisrable experimental specimens) and chemically contain a close approximately of what the rodent would eat in the wild; and certainly provide what is required by the rodents for proper nutrition in captivity. The same can be said for chicks and commercial poultry chows. It is regrettable that this type of study has not been done for the lizards (or for any other reptile), but they have never been of any commercial importance. In fact the answers to many of our problems would come about if we could somehow figure out a way to make reptiles commercially or economically important.

As much as I hate to admit it, the study of herpetoculture, in general, would be more advanced, and our knowledge of our specific captive's general requirements better documented, if we were rearing these reptiles for the skin trade. Reptile skin is fashionable, and if we were to exploit the market (and the animals) and establish 'skin farms' to rear these animals, there would be a great deal more research into the health and well-being of these animals. Create a multi-million dollar market, and researchers would be knocking our doors down to study the husbandry, reproduction, and health requirements of reptiles. The same would hold true if it were discovered that some part of a reptile held the answer to a cure for cancer or some other human disease. That reptile would then become extremely important scientifically, and commercially, and researchers would produce data at a fantastic rate; much of which would be of inestimable value to the herpetoculturist.

In the Medical Post of January 8, 1985 (Henahan, 1985) there is an article entitled, "Lizards give a morphine-like decapeptide." The author states, "a decapteptide isolated from the skin of an Australian lizard [the species is not named] produces morphine-like resistance to pain in human volunteers." If this proves to be a valuable drug for human use, and if it cannot be artifically synthesized in the laboratory, then the captive propagation of these lizards, for the manufacture of the drug, seems a likely progression. And, that would generate research and study into the propagation and husbandry of this species (and, perhaps, related species) in captivity.

I believe that, without exception, we must supplement the diet of insectivorous and omnivorous lizards with vitamin and minerals. A complicated problem is how to properly administer those supplements. The common practice of dusting insects before they are fed is only partially effective. If the lizards eat the dusted insects immediately upon presentation it undoubtedly is more beneficial than if the insects are not eaten for hours or days afterwards (time gives the insects the opportunity to rid themselves of this supplemental coating). Adding vitamins and minerals to the drinking water or the water which is sprayed on plants or the cage surface is of limited value because many of the necessary nutrients are not water soluble and it is extremely difficult to calculate how much an individual lizard is receiving. Actually this holds true for dusting...one never knows if an individual lizard has received an adequate amount. Many breeders have experienced spinal malformation, congenital deformities, and numerous other problems with captive born lizards. And although we don't have definite proof, it would appear that these animals are not receiving all that they require for proper nutrition.

Calcium should be offered to all egg laying lizards, because many of the insects which we feed them and almost all of the mineral preparations are somewhat deficient in this mineral. We provide crushed cuttlebone <u>ad libatum</u> to all of our lizards, and we have found that the majority relish it and readily consume it from a dish. It would appear that these individuals recognize the deficiency and seek this supplemental source. Miller (1984) has pointed out the complicated interaction of calcium, phosphorus, vitamin D3 and ultra-violet light. This information should be read and taken to heart by all herpetoculturists.

Live bearing lizards must be provided with adequate cover and concealment for any expectant young. This is another problem which we don't encounter in any but the ophiophagus snakes. Most snakes just do not eat their young; however, probably the majority of lizards will consume their young or eggs if given the opportunity. We provide as much cover as possible in the form of brush piles, rock piles, flower pots, and living and artificial plants.

Lizards are frequently housed in large communal cages, and with almost all species there are great problems of territoriality and competition. In many forms this is readily apparent. The varanids have a very definite hierarchy; Phelsuma, Eublepharis, and many others exhibit strong male dominance and though they can be kept in large groups, it is rare to find two males that will tolerate each other in the same environment. We have even encoutered problems where one male Phelsuma can see another in an adjoining cage. We have corrected this problem by designing our cages with opaque glass on the sides, so that if two cages are placed next to each other, the occupants of one will not be able to see those of the other. It is quite possible that many of the species which we have been unsuccessful in our attempts to propagate, but which appear to be quite communal, may in fact not be. No one has yet arrived at a standard formula for the number of animals (based on their size) that can be housed in a particular size cage. Additionally, we may not recognize the territorial displays of certain species, or the symptoms of stress in those species.

We have been providing all of our diurnal lizards with ultra-violet light in the form of black lights or vita-lites.

In the case of our Cyclura and Iguana we have a large outdoor enclosure where they can spend their days during the warmer weather. Even some of those forms which are primarily nocturnal seem to benefit from ultra-violet light. We have been providing it for Teratoscincus, and most of the animals will come out for short periods of time during the day and receive some exposure to this light source. The Gerrhosaurus validus are provided with a sun lamp, and they spend almost the entire day basking under this lamp.

Heat is provided in a number of ways. Some animals are housed in terraria which have no supplemental heat, but are governed by the room temperature. Others are provided with hot rocks, incandescent reflected light from above, heat lamps and specific basking sites, and through the use of substrate heat tapes. The Teratoscincus are housed in specially designed ten and fifteen gallon aquaria. These aquaria were constructed with opaque glass on all but one end where there is standard clear glass. In place of a glass bottom, we have installed a sheet metal bottom. These aquaria are lined up in rows on standard metal shelving with a heat tape running under the cages. This very effectively heats the substrate (in this case sand) and provides a very adequate environment for the geckos.

There are some very general guidelines on egg incubation temperatures and methods for certain species, but with others it remains an unknown. At the Reptile Breeding Foundation we allow eggs which are adhered to the cage surface or furnishings to incubate in the cage. The only interference from us is the installation of clear plastic cups affixed with silicone sealant surrounding the eggs, so that the hatchlings will not escape or be eaten before we can retrieve them. Most other eggs which are laid under rocks or buried are removed and incubated in plastic shoe boxes with moist peat moss. These are simply placed on a shelf in one of the animal rooms where the temperature fluctuates between 22 and 30°C. The one exception to this is with the eggs of Teratoscincus scincus. These we have had the greatest success hatching in a human incubator (Isolette) at a constant temperature of 32°C, and at a rather high relative humidity (about 80 percent).

One of the areas which has helped us immensely with our husbandry of certain species has been the establishment of naturally planted terraria (Huff, 1982a) Based on the realization that it is impossible to duplicate nature in captivity, but that one can do a lot toward approximating a naturalistic environment, we have planted most of our lizard terraria with live plants. Where possible we have planted cages with either the same or similar species found in the lizard's natural environment. When we don't know the lizard's natural requirements, we have used the premise, "if the plants thrive and do well with the given temperature, light, and moisture, then we are at least approaching what the lizard may require."

PROBLEMS

One of my complaints with a good number of herpetoculturists is that they often (conveniently) fail to mention their failures, or the problems which they are having with their captives. If these problems are not brought to light, how are they to be rectified? For that reason, I have decided to include a section on some of the problems which we have experienced.

We have been very successful in our reproduction of Phelsuma guentheri, but we are producing an inordinately high percentage of female neonates. Why? The first thing that comes to mind is that sex in this species may be temperature dependent. Fine, but that is just one of the problems. These lizards lay their eggs on the glass sides of the terarium. Removal of the eggs is impossible without jeopardizing their survivability. It is not practical to incubate the entire terrarium. We are in the process of setting up some new terraria that have removable, double-pane, sides so that if the eggs are laid on the glass, we can remove a pane of glass and place it in an incubator at a controlled temperature. However, if sex is temperature-dependent, why have some males been produced when incubated at the same temperature that produces mainly females?

We have also been quite successful in our breeding program with Teratoscincus scincus, but to date we have not been able to reproduce or to even get viable eggs from Teratoscincus przewalskii, a very similar species. Why? What needs does this latter species require that the former does not, and which we are therefore not providing.

Some breeders have had difficulty with spinal deformation in their captively reproduced offspring. Why have they had problems and we or someone else has not, and yet we are maintaining them under very similar conditions? What are these minute but critical differences in the husbandry of our collections?

We have successfully reproduced Cordylus warreni depressus, Leiocephalus carinatus hodsoni and Phrynosoma douglassii but to date have not been successful in rearing the young. What reuqirements do the young have that the adults apparently do not need? We are attempting to correct this situation by providing higher levels of ultra-violet light, greater vitamin and mineral supplements, and a broader diet. However, in the latter we are limited. At times I think we are nothing more than blind men groping our way through a strange house. We don't know enough about their activities or dietary habits in the wild. Do the young feed on a particular type of invertebrate? Do they require a specific trace element which we are not providing? These problems are frustrating. However we can take pride in the fact that we have learned how to maintain the adults, and how to induce mating...In that regard we know more than we did a few years ago, but we need to know more. These are the challenges of the herpetoculturist.

A possible solution to the above problem was mentioned at the conclusion of this paper by Dr. Richard A. Ross. He mentioned to me that in a paper on iguanas (Miller, T., in press) reference was made to the fact that young hatchling green iguanas, Iguana iguana, had been observed eating the faeces of adult iguanas, and it was postulated that this allowed the habitation of the young lizard's gut with beneficial flora for proper digestion and good health. This may not be the answer to our problem, but it is a potential solution and one well worth investigation. (This experience also serves to illustrate the value of sharing your problems publicly, and of the immense value of meetings and gatherings of herpetoculturists).

We successfully maintained and achieved viable copulation with Ophisaurus apodus. One female laid a large clutch of eggs in a burrow which she excavated under a large pile of rocks in a communal cage containing six animals. She guarded them for over a week and then, without warning, one of the males chased her out of the way and consumed the eggs. Regrettably, we have not had another clutch of eggs to remove and incubate away from the males. But would it even be necessary, or was this just a fluke? We have also continually had problems with Diploglossus warreni eating their young, and with the adults and juveniles viciously attacking one another.

Perhaps the greatest frustration for me has been that we have specimens of several species which we have been unable to pair or group. Several years ago I presented a paper (Huff, 1982b) on the need for cooperation among zoos and herpetoculturists in assisting one another to pair or group potential breeding animals. There are still many individuals and institutions which would sooner maintain a single example of a rare or unusual species than place it on loan to another collection where there is the potential for reproduction. As an example, a look at some of the less common cordylids in Slaven's 1984 inventory shows many institutions holding a single specimen of a species, and only a few collections with so much as a pair. Sometimes the problem is an international border. This is something we face in Canada all the time. There may be a zoo in the United States which is willing to send us an animal on breeding loan or a dealer willing to sell one, but because of paperwork and hassles with applying for permits, etc., the deal falls through. Let's get together, let's initiate breeding loans, let's fight regulations which make it difficult to obtain a single captive animal from a collection in another country.....LET'S COOPERATE!

CONCLUSION

Generally speaking, more work needs to be done with captive saurians. We need to learn the parameters necessary to properly maintain these reptiles in captivity. We need to learn the enviromental factors (both artificial and real) which induce reproduction. We need to learn more about the proper nutrition, health, and growth rates of neonates. And, above all, we need to share this information; we need to disseminate data, ideas, observations, and thoughts...We need to work more closely together.

We need to establish large gene pools of reproductively active species to supply zoos and individuals and eliminate the need to remove these animals from the wild.

The Convention on International Trade in Endangered Species (C.I.T.E.S.) has listed those species which it believes to be endangered or threatened as a result of commercial activity. It is not surprising to see chameleons, iguanas, monitors, tegu lizards, the cordylids, the Phelsuma and the Helodermatidae on this list. Of all these groups, only with some species of Phelsuma are we even close to reaching a point of self-sufficiency...a sad commentary on our profession (or hobby, as the case may be).

Because of the C.I.T.E.S. regulations and other legislation protecting some of these species, animal importers are seeking other lizard species to supply the pet trade. Many of the poorly known Southeast Asian agamids, the lesser known South American iguanids, and the Asian skinks and geckoes are being imported by the hundreds of thousands. How long can that drain continue before those species are also depleted in the wild, and reclassified as threatened or endangered. The only species for which there is a captive self- sustaining population and which has the potential to meet the demand of collectors and the pet trade is the leopard gecko, Eublepharis macularius. If we can do it for that species, we can do it for others. Sure varanids, teids, and the larger iguanids need a lot more room than leopard geckos, but if a few individuals and a few institutions concentrate their efforts, and if the rest of us contribute our knowledge, our surplus animals, and our efforts, we can do it.

I would like to end with an advertisement: The Reptile Breeding Foundation is in the process of publishing the inaugural issue of a new journal called The Herpetoculturist. This is not a commercial venture, simply an effort to initiate a publication which would reach a global readership among herpetoculturists. Most North American herpetoculturists are ignorant of what their counterparts in Europe, Africa, Australia and other parts of the world are doing. And the reverse is true; they don't know what we are doing. It is my hope that a journal such as ours might correct that gap in our knowledge. We will soon have the first issue available, which will more fully explain our goals and objectives. We have not yet established subscription rates because they will be dependent upon the interest. I am pleased that already we have had orders from herpetoculturists in twenty-two different countries. If you would like further information, please write to us at the Reptile Breeding Foundation, P.O. Box 1450, Picton, Ontario KOK 2TO Canada.

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MAINTENANCE AND REPRODUCTION OF SELECTED SPECIES OF NEOTROPICAL IGUANID LIZARDS

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INTRODUCTION

The family Iguanidae includes some 700 species of lizards largely endemic to the New World. Although thousands of these lizards are still exported every year, primarily for the pet trade, most species have not fared well as captives due to their intricate nutritional and environmental requirements.

Since 1981, seven species of neotropical iguanids have reproduced successfully at the Fort Worth Zoological Park. Of these, the casque-headed lizard (Laemanctus longipes), the crested iguanid (Cortytophanes cristatus) and the long-tailed tree lizard (Uranoscodon supercilosa) are of particular interest, as they represent specialized forms largely ignored by other workers in the field. They live in primary and secondary forests of lowlands and foothills from southern Mexico to northern South America (Peters, 1970). Some general characteristics of each of these insectivorous and oviparous species are discussed below.

Laemanctus longipes is the most arobreal and alert of the three species. It has a body which is slightly compressed laterally body and a round tail usually exceeding three times the body length. In males, a flattened area just posterior to the vent is often evident. A bony projection on top of the head of this green lizard probably aids in catching rainwater for drinking.

Corytophanes cristatus is an arboreal or semi-arboreal lizard. This lizard has a laterally compressed body and round tail twice exceeding the body in length. The large crest extending from the top of the head to the back probably aids in catching rainwater for drinking, and functions in defense when extended with the gular flap (Davis, 1953). Some populations exhibit obvious characteristics of sexual dimorphism. The grayish males have larger crests than the brownish females. However, in some populations both sexes exhibit similar coloration and crest size (Porras, personal communication).

Uranoscodon superciliosa is a semi-arboreal and semi-aquatic lizard. This species often frequents the lower parts of trees surrounding water courses (Hoogmoed, 1973; Lamar, personal communication). The body is rounded and the tail, which usually twice exceeds the body length, is laterally compressed towards the end. The base of the tail is generally wider in males than in females. The spikes extending from the back of the head through the dorsum serve no known function in this primarily dark brown species.

Although all three species are territorial, none are nearly as aggressive, inter- or intraspecifically, as Anolis spp. Along with compatibility of size (slightly less than 1' snout-vent length as adults) this characteristic enabled us to house small groups of all three species in one exhibit.

Housing

Located in the "cool room", where ambient temperatures ranged from 76-78°F, this exhibit measures approximately 3'x 5' x 6'. Daytime thermal gradients using 50 and 75 watt incandescent spotlights enabled the lizards to bask at temperatures approaching 95°F. Additional lighting was provided by full-spectrum fluorescent tubes and backlights for additional ultraviolet radiation in the 300-400 nanometer range (Townsend, 1979). A skylight located on one side and not directly above the exhibit also provided some natural illumination. The exhibit was decorated with natural rock on the back and sides and live tropicals which were planted throughout the enclosure. Grape vines and limbs provided additional perches, basking sites and visual barriers. Three water drips positioned in different areas, provided constant access to fresh water for drinking as well as keeping the relative humidity at about 60 percent or above. Forced air systems for heating and cooling provided constant air circulation through 1/4" screen on the top and parts of the back and sides of the enclosure. Two additional exhibits in other parts of the herpetarium employed similar setups for housing groups or individuals of the three species.

Husbandry

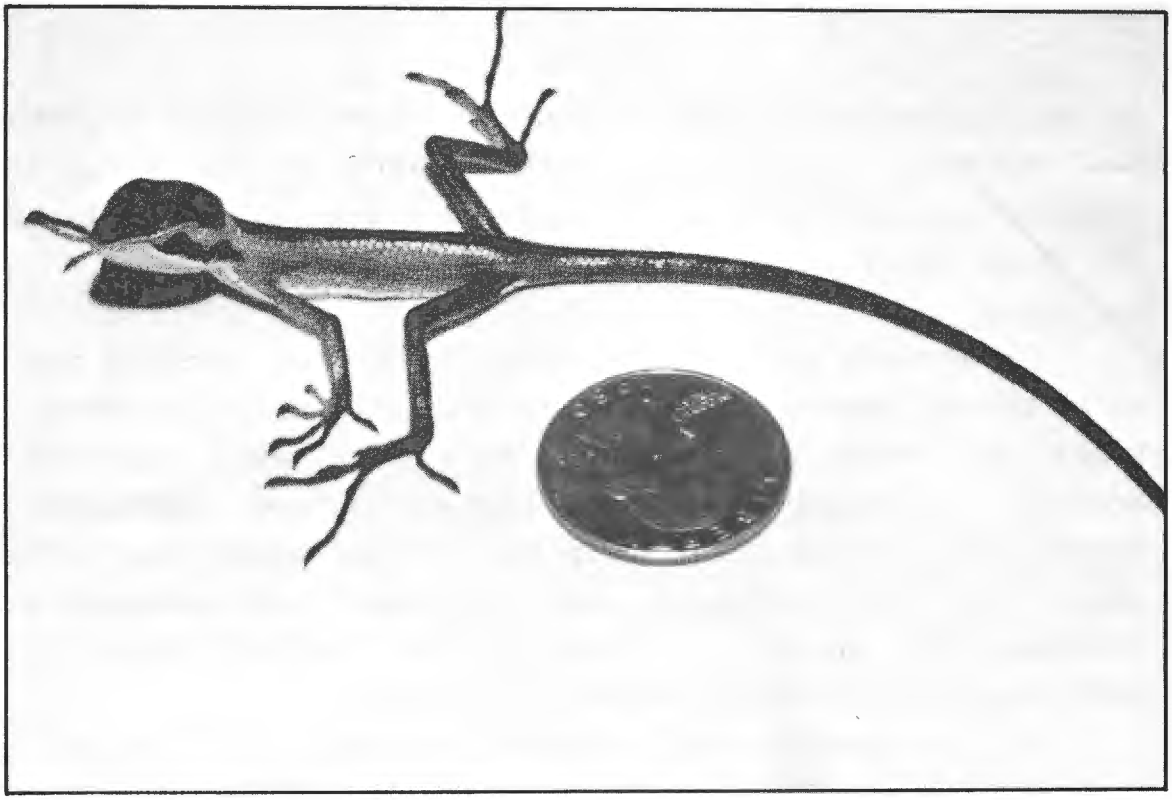
The lizards were fed crickets, mealworms, beetles and wax moth larvae dusted with vitamin and calcium supplements 1-3 times per week. Pre-weanling mice were also accepted on occasion by Uranoscodon and Laemanctus and earthworms by Corytophanes. Most of the captives became quite tame after a short time in captivity and accepted food presented on forceps. This method of feeding was used to fatten lanky individuals and females after oviposition.

All specimens were misted frequently (1-3 times per week). For some individuals this may be a necessary part of their husbandry as they were rarely seen drinking from the permanent drips throughout the exhibits. Almost without exception Laemanctus drank whenever misted on top of the head, as did Corythophanes on occasion. Uranoscodon, the most water loving of the three species, often soaked when provided with a pool or water bowl and drank water from a branch or rock that was misted. Direct misting on the head or body often caused individuals of this species to flee to another area of the enclosure and was thus avoided.

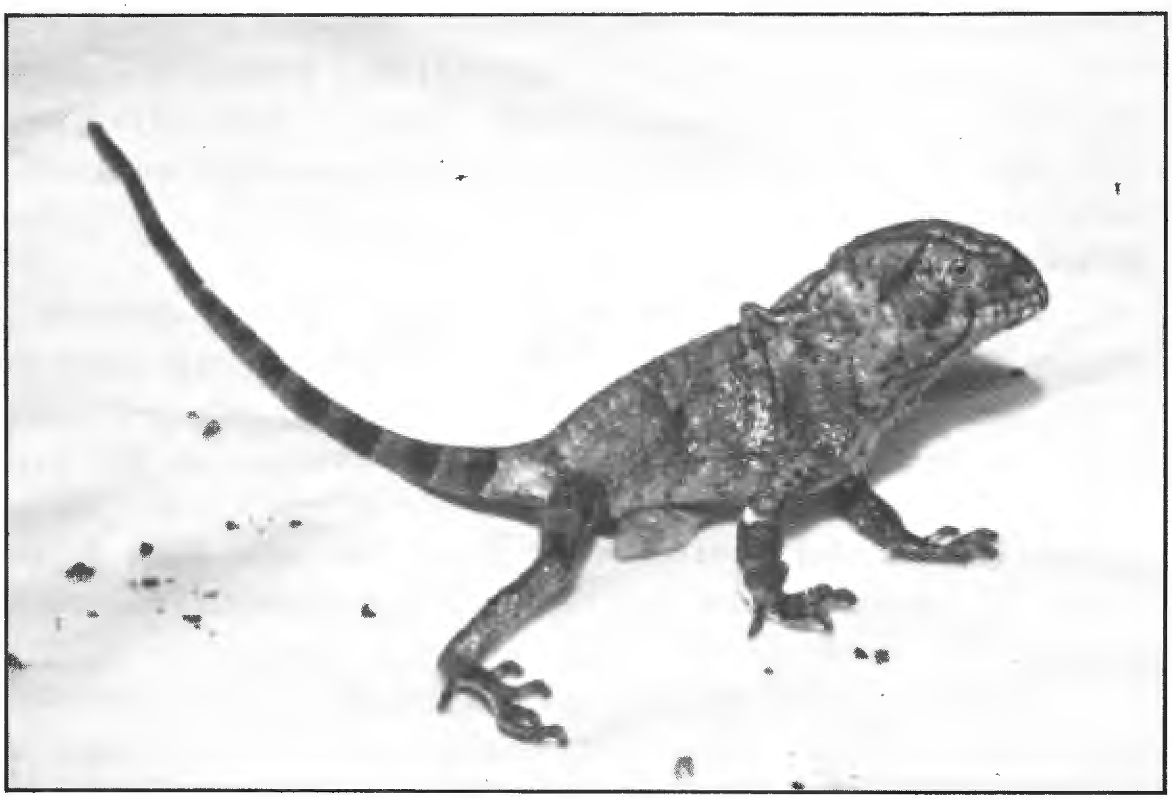
REPRODUCTION

A sex ratio of one male to one or two females per enclosure appeared adequate for all species. In courtship, which was seen on one occasion in Corytophanes and several times in Laemanctus, the male bit the females on the tip of the crest while trying to mount from the side. Although partially extruded hemipenes of males had been seen on occasion, it is uncertain whether successful copulation ensued during these observations.

Gravid females of all species became fairly conspicuous during the latter stages of egg production and eggs were easily palpated shortly before oviposition (1-2 weeks). At



Hatchling casque-headed lizard (Laemanctus longipes). Photo by R. Martin.



Hatchling crested iguana (Cortytophanes cristatus). Photo by R. Martin.

this time, the gravid females were separated into smaller enclosures for egg laying.

The eggs were incubated in a vermiculite media (Tyron, 1975) at tempertures ranging from 80-90^{O}F. Fourteen of 17 fertile eggs laid by two Laemanctus females in five clutches, hatched in 50-67 days. Two of five fertile eggs laid in one clutch by Corytophanes hatched in 125 days, while a clutch of three fertile eggs layed by Uranoscodon hatched in 86 days. Results of a wild breeding, a previous clutch of 10 eggs laid in a flower pot by Uranoscodon hatched after an unknown period of time. Six of the hatchlings were recovered from the exhibit in which the eggs were laid while the other four lizards were presumed to have been eaten by green basilisks (Basiliscus plumifrons) which shared the exhibit.

The babies of all species were raised initially in small enclosures ranging from screen-topped gallon jars for individuals to 10 gallon screen-topped aquaria for groups of up to four individuals. The tops of the enclosures were placed less than one foot directly under full spectrum and blacklight fluorescent tubes. Frequent misting was required as permanent water drips were not incorporated into the enclosures. With subsequent growth, the surviving babies were moved into larger enclosures.

Due to our small sample size (24 hatchlings) it is difficult to ascertain the exact reasons for our ability to raise some of the hatchlings to adulthood (Laemanctus) while others died. Rubbery bones, as seen in the metabolic bone disease, defined by Fowler (1978) did not appear to be a factor in most deaths. We are currently looking into the effects of relative humidity on raising hatchlings.

CONCLUSION

As deforestation continues at an alarming rate throughout most areas of tropical America, the future survival of many species of Iguanid lizards remains dubious. The more specialized forms, with their diverse morphological adaptations and intricate social behavior have classically not faired well as captives. It is hoped that this article will stimulate interest among co-workers in resolving some of the problems associated with keeping such forms in captivity on a long-term basis.

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LONG TERM INDOOR MAINTENANCE OF CHAMELEONS

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INTRODUCTION

The oldest surviving description of a chameleon was written by Aristotle, probably between 347 and 334 B.C., and until a few decades ago, most of the literature on chameleons consisted of species descriptions. More recently, articles have been written on their captive care (Bustard, 1958, 1959, 1960a, 1960b, 1962, 1963a, 1963b, 1963c, 1963d; Dowling, 1963; Francke, 1963a, 1963b; Hirschfield, 1962; Langhammer, 1962; Laszlo, 1969, 1978; Lilley, 1982; Mertes, 1949; Miller, 1973; Rose, 1955; Shaw, 1960; Switak, 1971; Van Mater, 1971 and 1972), but the husbandry of chameleons remains a real challenge. Very few zoos will even attempt it any longer. Almost all private parties enjoying success rely heavily on a local climate similar to the animal's native habitat.

Although chameleons seem to be ideal animals for some kinds of research, little is known of their natural history. Field study is difficult because they are arboreal, and most are native to a continent plagued with political problems. Additionally, access is often restricted by logistic and geographic barriers. Much could be learned about this family of lizards if they could be maintained indoors for reasonably long periods of time.

This paper presents insights into the indoor husbandry of chameleons gained from a questionnaire returned by 43 zoos and 18 private collectors with experience in the husbandry of Jackson's chamaeleons (Chamaeleo jacksonii) and from records kept on hundreds of individuals of 10 species maintained indoors by the author during the last seven years, exclusively indoors.

CAGE CONSTRUCTION AND FURNITURE

It has been reported (Bustard, 1963b; Francke, 1963a) that chameleons will not thrive except in large enclosures which mimic their natural environment. Indeed, in most cases, those who have successfully maintained chameleons on a long-term basis have kept them in large outdoor enclosures all or much of the time. However, it is possible to maintain them strictly indoors in small enclosures.

Glass aquaria are ideal in that they can be easily kept clean easily and provide good visibility. I use 30 gallon-long aquaria almost exclusively. The dimensions are approximately 36" x 12" x 16". No top is necessary provided the cage furniture is arranged in such a way as to prevent escape. For chameleons larger than about three inches snout-vent length an entire aquarium is provided per individual. For smaller chameleons the aquarium may be divided into two enclosures using a partition of translucent, not transparent, glass. Chameleons will often crawl the glass, especially when first introduced into the enclosure. This is interpreted by most as a sign of stress, but I am convinced that it does them no harm. Even in large enclosures with many plants, chameleons often wind up in the corner crawling the glass.

Maintaining one chameleon per enclosure is an important factor in successful long-term indoor maintenance of chameleons. Chameleons appear to be, in general, solitary creatures. In nature, encounters between chameleons are infrequent and of short duration. When chameleons are housed in pairs or within sight and close proximity they often spend much of the time displaying to one another. I believe this eventually takes its toll and that our anthropomorphic desire to see animals kept in pairs or groups is one of the major factors contributing to the poor success experienced by most keepers. Even new born or newly hatched chameleons display and attempts to raise them seem to be more successful when they are housed individually. Where there are adjacent aquaria the sides facing other aquaria should be covered with a translucent material unless the aquaria are separated by at least several feet.

Surprisingly, I have never found it necessary to furnish the aquaria elaborately or mimic the chameleon's natural environment. Live or plastic plants only provide hiding places for their insect food and obstruct the keeper's view. The result is that the chameleon's prey often avoids being eaten and the keeper assumes, because of the difficulty of observing either the chameleon or the insects, that the chameleon must be eating and doing well. Then by the time the keeper notices a problem, it's too late. Likewise substrate is unnecessry and undesirable since it makes keeping the cage clean and dry difficult, if not impossible. Further it may be ingested by the chameleon causing broken teeth, mouth infections, and other problems.

THERMAL AND LIGHTING REQUIREMENTS

Generally, it seems chameleons can be divided into two thermal types: "hot" species which are characterized by small homogenous scale patterns and "cool" species which are characterized by heterogenous scale patterns. Hot species prefer body temperatures around 32°C while cool species are stressed at that high a temperature and seem to prefer temperatures around 25°C.

Each of my aquaria is furnished with a ceramic incandescent light socket in one corner, located so that the chameleon can not climb from a branch to the connecting wire. At room temperature, I have found that a 25 watt bulb is quite sufficient for the cool species while a 60 watt bulb is better for the hot species. When lower wattage bulbs are used, the chameleons seem to spend an inordinate amount of time basking near the bulbs while higher wattage bulbs often produce signs of heat stress such as gaping, blanching or retreat to the farthest corner of the enclosure. The incandescent lights can be wired with the overhead fluorescent lights and controlled with a timer to create both a photoperiod and thermal-period. Some drop in temperature overnight does no harm. In fact, most chameleons can tolerate quite a sizeable drop in overnight temperature with no apparent ill effects, but I have never found a large drop in overnight temperature necessary, as has been suggested (Laszlo, 1978).

It has been suggested that light quality and/or intensity are important factors in successful long-term maintenance of chameleons. Lamps providing middle and near UV cause vitamin D_2 to be converted to D_3 in the skin. Vitamin D aids in the absorption of calcium from the diet and thereby helps prevent osteodistrophy or rickets (probably the most common problem in rearing chameleons). Full spectrum and/or high intensity lighting may also be beneficial, perhaps by stimulating normal hormone levels.

Though UV, full spectrum and/or high intensity lighting may provide some benefits, they do not appear necessary. The amount of UV most likely to convert D_2 to D_3 that is provided by full spectrum fluorescent lamps, such as Vita-lites, is no greater than that provided by regular fluorescent lamps. The amount of UV drops off quickly with distance from the lamp and as the lamp ages. Further, the intensity of most fluorescent lamps is so low that it seems impractical to try to mimic the sun's intensity at any part of the spectrum. Finally, some report enviable success using only simple incandescent lamps.

For my cages a single 24" 20W overhead fluorescent lamp is wired in with an incandescent lamp (used for thermoregulation) to a timer switch and the photoperiod set at about 14 hours.

DIET AND SUPPLEMENTS

The diet may well be the principal factor determining the success or failure of long-term maintenance of chameleons. Starving chameleons appear to be a captive phenomenon (Lin, 1975). In nature most species of chameleons have a large, seasonally changing, variety of insects available in substantial numbers. They may even select for novel prey. Experiments so far support this hypothesis (Eason, 1985). Frequently, I have seen chameleons refuse one kind of insect only to eat the next kind offered as if they were starving. For some time I felt that I was witnessing individual prey preferences but examining record sheets for over a year on six individuals of the same species, it became clear that each individual ate about the same proportion of each variety of insects but was switching from one type to another. Some species of chameleons do appear to be tolerant of a monotonous diet but at least three or four kinds of insects seem necessary to provide the variety needed for regular feeding.

Many who successfully raise chameleons depend on wild insects from sweep netting or house their chameleons in outdoor enclosures where the animals have access to a variety of wild insects. I have found that insects and other prey items easily cultured or purchased, such as mealworms, waxworms, crickets, and pinkie mice, are adequate, if a vitamin-mineral supplement is used.

The most obvious dietary problem among chameleons maintained indoors seems to be an incorrect calcium-phosphorus ratio in the prey provided. This shows up in defective or incompletely formed egg shells or as decreased hatching success. In the young it appears as osteodystrophy (rickets). Probably a proper calcium-phosphorus ratio in a reptilian diet approaches 1:1, but most cultured insects have calcium-phosphorus ratios from 1:3 to 1:20. The problem becomes one of increasing calcium absorption in the gut without increasing phosphorus absorption.

Adding calcium to the diet is one approach to solving the problem. Unfortunately bone meal (or calcium phosphate), which also contains phosphorus, is often used as a calcium supplement. It must be stressed that in most cases the problem is an improper ratio of calcium to phosphorus, not too little calcium. Therefore, no matter how much calcium is added, if phosphorus is also added, the problem is not solved. The ratio must somehow be brought closer to 1:1. Another way of attacking the problem is adding vitamin D to the diet or perhaps stimulating the natural production of D_3 by increasing UV radiation. Vitamin D aids in the absorption of calcium in the gut. Of the three forms of vitamin D, D_3 is by far the most active, requiring about 1/100th the amount of D_2 to produce the same results.

To help compensate for the fact that there is usually an improper calcium-phosphorus ratio in the diet and that there may not be enough vitamin D or UV provided, I dust insects with calcium lactate and a vitamin-mineral powder to which pure D_3 has been added. I prefer "8 in 1" vitamin-mineral powder because the fineness of the powder causes more of it to stick to the insect and because the vitamins and minerals seem to occur in the best proportions in this brand. For example, it has less vitamin A which can accumulate in the body to toxic levels. To a 3 oz bottle of the vitamin-mineral powder, I add one gram of pure D_3. This results in an extremely high D_3 content but, since D_3 breaks down so easily and since so little is likely to wind up sticking to the insect long enough to be ingested, I believe there is little danger of overdose. Vitamin-mineral supplements should be refrigerated to slow the breakdown of some vitamins, particularly D_3.

Finally, it should be stressed that the blood calcium-phosphorus ratio is not solely determined by the calcium-phosphorus ratio in the diet. A few of the many other complex interrelated factors are listed below:

- A high fat diet interfers with calcium absorption.
- Vitamin D_3 not only enhances calcium uptake but also enhances phosphorus excretion.
- A high protein diet enhances calcium absorption especially when the amino acids lysine and/or arginine are present in large amounts.
- Calcium absorption improves at lower pHs. For example, though calcium carbonate may have more calcium per unit weight than calcium lactate, calcium lactate may be a better calcium supplement since it tends to lower the pH while calcium carbonate tends to raise the pH.

WATER AND RELATIVE HUMIDITY

Chameleons will rarely drink from standing water so water must be provided in drop form. Rather than troublesome drip systems or bubblers, I have found that simply spraying a wall of the aquarium several times a day is quite sufficient. If deionized water is used there will be no mineral build up or scaling of the glass. The chameleons quickly learn to take water in this way and what they do not drink quickly evaporates.

Maintaining high relative humidity in the enclosures has not been necessary for the species with which I have worked, except in the case of babies which is discussed later in this paper. Relative humidity in the enclosures was usually about 40 percent. No problems with molting were observed among healthy individuals at this humidity.

RECORD KEEPING

The changes in the condition of chameleons are more subtle than in most animals. This may explain the sudden death syndrome so often reported. My experience has been that in almost every case there are problem signs well in advance of death. I believe these often go unnoticed because it is difficult to observe the animal (e.g., too much cage furniture) and because the keeper is not familiar enough with the animal's routine. Maintaining records forces the keeper to observe the animal critically and provides a history against which to judge the animal's current behavior. It takes only a few minutes per animal per day to update a record sheet but it is a valuable reference. Time and again my record sheets have demonstrated how wrong impressions can be. A sample record sheet is shown on the following page.

DISEASES AND TREATMENTS

Next to osteodystropy, the most common diseases among captive chameleons are respiratory and mouth infections. Treatments are often ineffective, possibly because they are initiated too late. The first sign of trouble is frequently a lack of interest in eating. It is important to begin hand feeding within a very few days, particularly if the animal is newly imported or underweight. In the early stages, it is relatively easy to hand feed by gently but steadily pulling on the gular pouch until the mouth opens. Once in the mouth, the insect is usually masticated and swallowed. Water should also be given at this time.

Though I have tried many different antibiotics, only two have proven effective. Septra (a sulfa based antibacterial drug) has been successful, especially when the problem is nonspecific. A 1:10 septra to water dilution is administered orally. The dosage is one drop per 10 grams of body weight per day, though I have given double and triple that amount with no apparent ill effects. Septra is a broad spectrum drug, effective against a variety of bacteria. However, it should not be given to an animal which has renal or hepatic problems or is dehydrated. I have also used a sterile benzathine penicillin G and procaine penicillin G suspension successfully to clear up a six to seven millimeter abscess which actually exposed the hip socket. The suspension was diluted with sterile saline solution and about 10 units per gram of body weight was administered every 48-72 hours by intermuscular injection using a tubercular syringe with a 26 gauge needle. In addition, the abscess was expressed frequently and treated with a topical antibiotic. Treatment continued well over a month and though effective it seemed protracted compared with antibiotic treatment in mammals and birds.

Finally, critically ill chameleons seem to lose their heat sense and will lie on the incandescent bulb provided for thermoregulation, resulting in severe burns. Care should be taken to prevent this by repositioning the bulb or branches, or perhaps, by substituting a hot rock or heating pad instead of an incandescent bulb as the heat source.

INCUBATING EGGS AND RAISING THE YOUNG

Almost everyone who has kept chameleons has at one time or another been presented with eggs or young. It seems most females are either gravid when collected, mate during shipment, or, in some cases, produce young from stored sperm. So there seems to be little problem acquiring young.

For egg laying species, some keepers suggest placing containers of substrate in the enclosure in hopes that the female will construct a nest. My experience has been that containers of substrate are usually ignored. Furthermore, eggs laid on the surface of a damp substrate dehydrate faster than those laid on glass. For these reasons I no longer add containers of substrate. The eggs are usually laid early in the morning and if collected reasonably soon will not dehydrate enough on glass to cause problems. Incidentally healthy eggs and/or young have been surgically recovered more than a day after the death of the mother.

The eggs can be incubated in a mixture of 50 percent water and 50 percent vermiculite by weight. I use a small snap-cap vial per egg. Though some puncture the lid to allow gas exchange, I believe this is not only unnecessary but may be harmful. It allows moisture to escape, making it difficult to maintain the proper environment. During the course of incubation the vial will be opened occasionally to check the egg, at which time there is ample opportunity for gas exchange.

Little information is available as to proper incubation temperatures, but eggs have been incubated successfully for <u>Chamaeleo tavetaenus</u> (a cool species) at about 75°F and for <u>C. dilepsis</u> (a hot species) at 80-85°F. Incubation can be as long as five or six months. In a few cases it seems hatching was induced by adding water to the substrate, perhaps stimulating the onset of rainy season. In nature, the eggs of some species overwinter through the dry season.

Whether live born or hatched, raising the young is a real challenge. Hardly anyone succeeds in raising young chameleons totally indoors. I have tried a variety of methods but have had the best results using half gallon plastic fish bowls which are flattened on two sides making them taller than they are wide. The babies are housed one per bowl and one side of the bowl is covered with translucent contact film in order to visually isolate young in adjacent bowls. The bowls are furnished only with bare twigs.

Dehydration is a special threat to newborns because of their high surface to volume ratio. The top of the bowl may be covered with plastic wrap. This allows oxygen to pass through but not water and maintains high relative humidity without constant misting.

Water is provided in the same way as for adults and the diet is also the same except that smaller instars are used. In addition, vestigial-winged fruit flies can be easily cultured and are relished. A few insects offered several times a day is preferable to many all at once. The young (and sometimes the older chameleons) will, after eating a few insects, often ignore the remainder. The result is that fewer insects are consumed overall than if they are offered in smaller quantities more frequently. The use of supplements is vital to raising healthy young. In particular, plenty of vitamin D_3 should be provided.

CONCLUSION

The most important factors contributing to successful long-term indoor maintenance of chameleons seem to be:

- Individual housing of the animals.
- Provision for thermoregulation.
- A varied diet.
- Supplements - particularly vitamin D_3 and calcium lactate.
- Close observation (record keeping)
- Enclosures which are easily kept clean, allow close observation, and afford prey little chance to hide.

Of these factors, diet presents the greatest challenge. Perhaps, further improvement will not be realized until more appropriate kinds of insects can be easily cultured and/or made available commercially.

MONTH/YEAR ____________________ SPECIES ____________________

WEIGHT (grams) ________________ SEX ____________________

TOTAL LENGTH (cm) ____________ ANIMAL NUMBER ____________

SNOUT VENT LENGTH (cm) ________

1	2	3	4	5	6	7
8	9	10	11	12	13	14
15	16	17	18	19	20	21
22	23	24	25	26	27	28
29	30	31				

C = CRICKET
MW = MEALWORM
WW = WAXWORM
S = SNAIL
P = PINKY
H_2O = WATERED
X = EXTRA
F = FECES

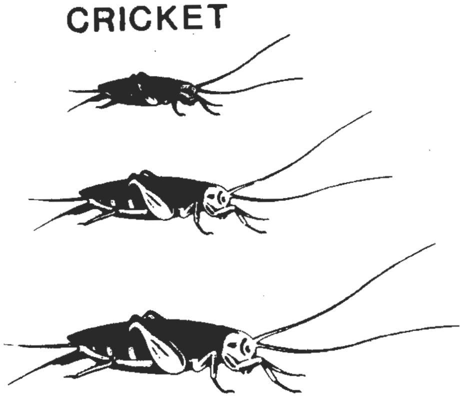
NOTE: Comments labeled by day

CRICKET

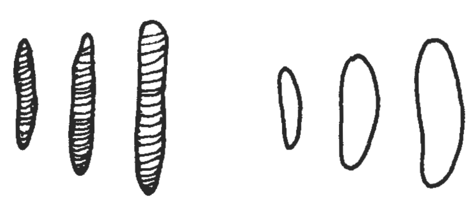
SM.

MED.

LG.



MEAL WORM WAX WORM



SM. MED. LG. SM. MED. LG.

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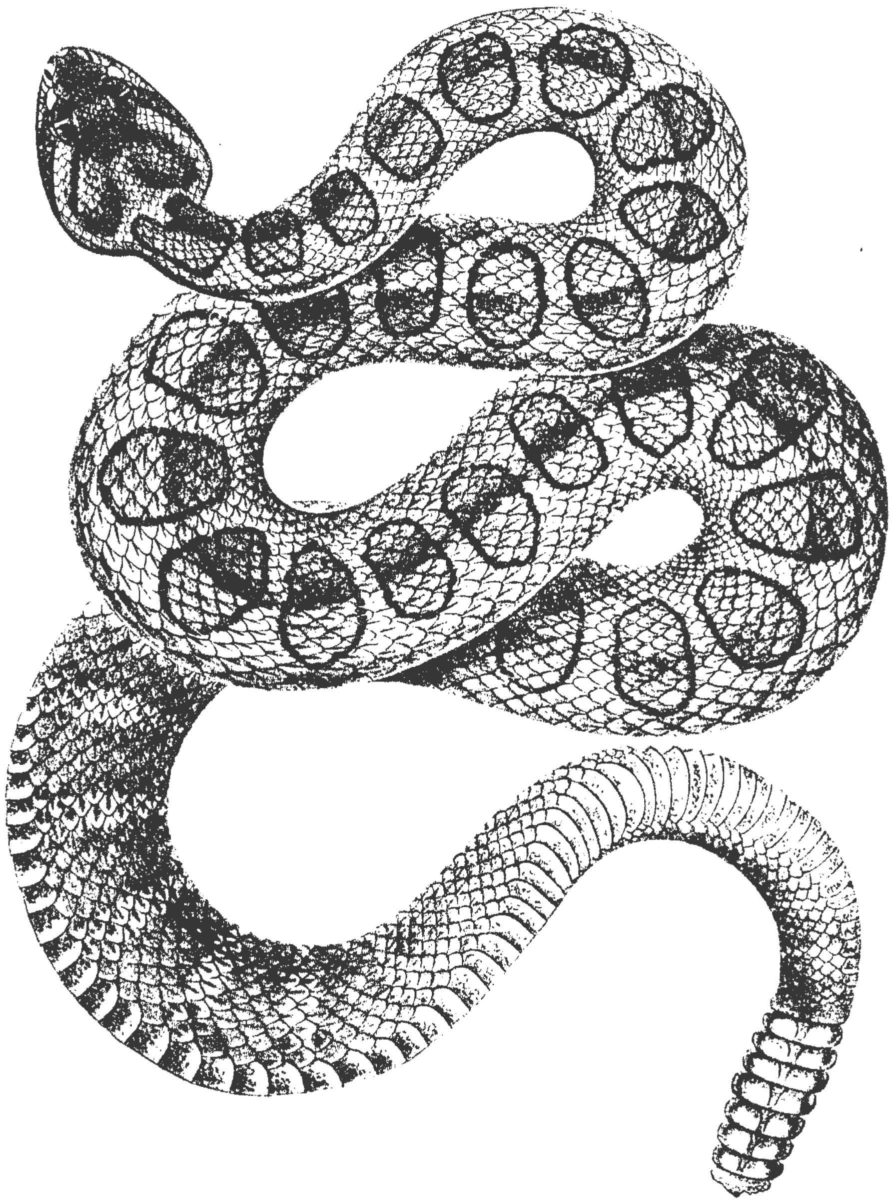

PROBLEMS ASSOCIATED WITH A COMMERCIAL COLUBRID BREEDING PROGRAM

Robert Applegate

INTRODUCTION

In the dairy business the number of cows one person can adequately care for has been well established. If a one person operation has a very specific number of cows and works at maximal efficiency, the maximum profit potential of the herd will be realized. Any fewer cows and the owner's time will not be fully utilized, and his potential income will suffer. Any more cows and the owner will have to hire help. However, unless the number of cows were doubled, the larger herd wouldn't provide the maximum income for two peole. If one person tried to take care of the larger herd the entire operation would suffer as the health, productivity, and profit potential of the herd would decline.

How many snakes can a one person operation properly take care of? The answers depend on many variables. These variables include, (1) how much time you can devote to the care and maintenance of a snake collection, (2) which species you intend to work with, (3) how well do you organize your time, and (4) what results or goals you intend to realize with your collection. Each variable is dependent upon the others and at present there is no single, established number of snakes that can be properly maintained by one person.

CHOOSING SPECIES TO WORK WITH

I have chosen to work with some of the colubrids (mostly albinos and what are known in the trade as "tri-colors"). These species were chosen because the <u>known</u> feeding and care requirements are similar and the same size cage can be used. My goals include providing a pleasurable activity for my spare time and to enjoy a hobby which can not only pay for itself, but the house as well, not to mention provide enough money to do some related traveling. I would also like to contribute something to herpetology that would benefit captive reptile breeding programs.

One of the problems in getting started is the choice of what you want to work with. If you don't have much experience in raising snakes pick species that aren't real expensive. If you make mistakes, the lessons are cheaper. Once you are confident in your ability to raise and breed snakes, pick whatever your interests and pocketbook dictate. If you choose a species already being bred in captivity, you will have an easier path. You can be assured enough is known about the species to breed them, and relatively parasite-free young can be purchased. There are many breeders and/or dealers making a business out of captive born species. A potential problem is that most dealers will not sell established adult breeding snakes, so be prepared to raise baby snakes to "adulthood", This may take one to four years depending on species and individuals.

Another problem is getting animals of the correct sex. It is wise to become proficient in determining sexes. For example, I once had an advance reservation for two pairs of Pueblan milksnakes. When the snakes became available, I wrote and requested advance payment. After a lengthy silence, I received a call from the gentleman explaining that a Florida dealer had made him a better offer, so he bought two "pairs" from that dealer. No hard feelings were felt on my part, but this type of occurrence is why I refuse to hold animals for people unless I have advance deposit. To end the story, about a year and a half later I received another call from this same gentleman asking about "extra" female Pueblan milksnakes. He explained that he had four nice <u>males</u> that needed mates. Even if the dealer were to take back two males, and trade for two hatchling females, or refund the money and take back all four, this gentleman would have been out the expected offspring and 1.5 years of work because he failed to verify the sexes of his new arrivals.

You can buy a large number of snakes at the onset of your "affliction" or, as I did, start with a few, produce young, and use the proceeds to expand into other species. The route requiring the least amount of time to produce offspring is obviously to begin with adult animals, rather than the young ones. Wild caught adults can be obtained through many dealers. Some obvious advantages are immediately realized: you don't have to raise babies and wild adults are good for expanding bloodlines, colors, etc. I find that with species which are not readily available for captive breeding that captive-born or wild caught are equally acceptable.

CAGING

The anticipated goals and future size of your collection should influence your caging choices. For a small collection, almost any type of caging that will provide adequate security, heat, light, and ventilation will do. Various aquariums, and commercially produced cages are, available, and home built cages can be constructed. My collection is quite large for the time I want to devote to it, so I have gone "assembly line" as much as possible to cut down individual cage maintenance time.

I start hatchlings out in plastic shoe boxes (3" x 7" x 12") with a pine shaving substrate, a folded paper for hiding, and a small "butter tub" with a 1" hole in its lid for an almost spill-proof water container. A record card which stays with the snake its entire life is taped to the top. A few small holes are drilled into both ends of the shoebox. These boxes are inserted into a rack which secures the lid, and provides heat via a strip of heat tape under each shoebox. The heat tape is regulated by a light dimmer and a room temperature thermostat. The snake can lay atop the heat tape and be warmed to 90^{o}F or move about in the box and choose a cooler temperature.

When the snakes outgrow the shoeboxes I transfer them to plastic sweater boxes kept in a rack or I place them directly into one of my breeding cages.

The "breeding" cages are glass fronted wooden frame, wood construction with double floor area including a drawer. I have

Bob Applegate inside garage snake room.

Bob Applegate's shoe and sweater box units.

two rooms with cages such as these. One room has 33 cages that each have a floor space of approximately 2' x 1.5'. The drawer ends about 4" from the back, providing an air space.

A length of heat tape runs through this space providing heat. For additional warmth the snake can lay against the back of the drawer while in the drawer or above the heat tape in the rear 4" of the cage if out on the top floor. The lighting is provided by "Powertwist Vita-Lites" above the 1/8" mesh wire open tops. The lights are on timers; the heat tapes are on light dimmers; and all power is routed through a master thermostat to prevent the room from overheating. There is also an electric vent fan programmed to ventilate the room for a few hours in the afternoon to reduce odors and freshen the air. These cages are stacked along the walls like rows of apartments. The substrate is silica sand with a water crock and an abalone shell above, with a folded sheet of newspaper in the drawer. The other room has 48 similar drawer-type cages with a floor area of 1' x 2'. This room is thermostatically controlled via 100 watt incandescent lights along the opposite wall, about 5' away. There are also 8' double-bulb fluorescent lights on the ceiling that are switch-operated and turned off and on at irregular and lengthy intervals. There is no direct light or heat in these cages. I have had good breeding success in both rooms.

I am not the original designer of the drawer-type cages. Many who built them before I did gave me helpful information on desirable and undesirable features, which I incorporated with my own ideas. I may, however, be the first one in this country to build wall units. The Europeans even have double side-by-side drawers under each cage, one for a dry hide area, and one that is moist to wet.

FOOD SUPPLY

What are you going to feed your snakes and where are you going to get it are questions that should have influenced your choices of species. The species I work with eat mice, so the "what" was easy for me. I decided that buying mice was too expensive and couldn't provide the smaller sizes always-needed for raising baby snakes. However, I didn't want to raise enough mice to feed 150-200 snakes. It may seem like an insurmountable problem, but it wasn't. Using approximately 32 commercial plastic mouse cages (12"x16"x8"), I raise enough of the smaller sized mice to feed all of my smaller snakes and sell the excess. I then purchase enough mice on a bi-weekly schedule to feed all of my larger snakes, and to resell some mice to local pet shops and friends. I purchase enough mice to get a discount. I sell enough of the "extras" at wholesale prices and pocket my discount, to pay for most of the mice I use. My mouse operation is almost self-supporting and almost feeds my snakes for free. Free, that is, if you don't figure the cost of the time involved.

Again, organization means efficiency and time savings. My mouse cages are in a line. When I see a pregnant female in a cage a nail goes in front in the wire top. When babies are in a cage, that cage is moved to the head of the line. When I need newborn mice, I review the cages with the nails first. If a cage has a nail, but no babies appear, cannibalism is suspected and the nail is placed in the back of the top. Any cage that stays at the end of the line, or gets two nails in the back is replaced with a younger colony of mice selected from offspring of the good colonies; males from one group, females from another. There are probably more efficient ways to raise mice but for my needs and an unwillingness to devote much time to mice, this works out very well. I clean cages and add food and water once per week. The water and food are all external, so at a glance while picking out feeder mice, I can check all 32 cages.

CARE OF HATCHLINGS

When a future breeder hatchling comes into my possession, it is weighed and sexed. This information, along with the source of the animal, who its parents are, hatching date, and any useful identifying markings are recorded on its record card. Then it is placed in a shoebox, kept warm the first year, and fed all I can get it to eat. The few which refuse to eat in the winter are hibernated. As long as the snake does not get "lumpy" and fat, I don't believe you can grow a snake too fast. Most of my snakes reach breedable size in 14 months. I don't have a regular vitamin supplement program, as I believe (or hope) that if the mice are healthy and breeding, the snakes should be able to do so also. I occasionally supplement "just in case" and I am receptive to new and proven ideas in this area.

CONDITIONING FOR BREEDING

At the end of the warm season (Nov. 1) all the adult snakes should be in prime condition. I stop feeding the snakes, wait two weeks for digestive tracts to clear, weigh them, and turn off all heat and lights. I then hope that the temperature of the cages will gradually drop and settle around 50°F. I then leave them in the hands of Mother Nature (her El Cajon, CA version), checking on them and changing water every 2 weeks. Meanwhile, I raise babies and take a vacation until March 1. I then weigh each snake again to determine winter weight loss. I warm up the room temperatures to 80-85°F in the day, and 70-75°F at night and start feeding them small frequent meals. Breeding activity should follow soon. This method has resulted in very dependable breeding success in most snakes. I have had a few snakes breed without "hibernation" but these are the exceptions to the rule.

BREEDING TECHNIQUES

While some of my snakes are kept as pairs, many are not. I feel for follicles to determine the probability that a female is ready. I separate and reintroduce cagemates. I spray the cage with a warm water mist. I place males together to induce combat behavior. I introduce individuals after either has shed. I even deliberately disturb them (I have had many cagemates copulate directly after I moved them around while cleaning the cage!) If I see snakes copulating, I record the time of copulation. After they separate I obtain a semen sample from the cloaca of the female, put it on a slide, and look at it under 200X on my microscope. If the sperm is rapidly swimming and in good numbers, I consider this a good mating. If I can't see anything, I introduce a second male and start over. In any event I will try to get that female to breed the primary male a few days later. I have had bad eggs result from a good mating, but I have never had good eggs result from a bad mating. I am convinced that I have gotten many good eggs that I would not have gotten if I hadn't followed up a bad mating with a good mating. I also try not to breed siblings, if I have a choice.

EGG LAYING

Most of my snakes are kept two to a cage. Most of the species I work with are predictable as to the time between the "pre-egg laying shed" and actual egg laying. When the egg-laden female sheds, she is isolated from her cagemate and given an appropriate sized "Tupperware" type container with an access hole cut into it, partially filled with wet sphagnum moss. She stays there until she lays her eggs. When this happens, she and the egg clutch are weighed, and the information is recorded on her card. The eggs are transferred to a container filled with moist vermiculite and placed in the incubator. When they hatch the date and number of each sex of the hatchlings is recorded next to clutch size and weight. The female is evaluated for condition and is either fed and introduced to a male immediately, or isolated from the males until she regains "prime" condition.

PREVENTATIVE MEDICINE

I hate to go back to the cows again, but it is a well-established veterinary principle of herd management that if you are treating sick animals, you are losing money. I feel the same way about my snake collection. Usually an animal that needs serious medical treatment is lost for the breeding season at best. I am fortunate to have a good veterinary friend, Dr. Winjum, who is helping me to establish a preventative medicine program for my collection. She has convinced me by demonstrated results that a full flotation fecal examination in addition to a direct smear is a valuable diagnostic tool for parasite determination. We are still in the beginning stages of this, but have already eliminted Trichomonas and some worms from a few of my wild caught breeders. Except for Trichomonas in a few captive-hatched animals which ate wild lizards (I have found Trichomonas in the feces of wild lizards), I am happy to report that we haven't found any internal parasites in any of my captive hatched offspring. I feel that elimination of parasites, proper nutrition, and good cage husbandry go a long way in reducing disease in captive reptiles. Year after year, better information on how to do this becomes available. Not too long ago mites were considered a serious problem. Now they are easy to eliminate in a number of ways.

LAWS

One of the most serious problems threatening our futures as captive breeders are local, state, and federal laws. For example, years ago I was breeding Lampropeltis zonata, as well as capturing and selling wild caught individuals. I was trying to produce a L. zonata with lengthways red stripes by selecting the offspring with the most incomplete bands. A law was then passed whereby you could only have one L. zonata so I got rid of most of my animals. Then you couldn't have any L. zonata and the federal "sting operation" materialized. I got rid of the rest (I am well known by Fish and Game and have a high visibility) and destroyed several years worth of "incriminating" data, just in case. Now you can have one again, but it can't come from one side of a certain freeway. The point is that the herpetological newsletters are full of examples of "can't own a non-domestic pet", "can't have snakes", etc., that are real or proposed laws. I wish I could say it's our fault and we should educate the lawmakers and convince them of our good intentions, but if you fail in this effort, years worth of developing and maintaining your collection are in jeopardy. I don't have a solution for this problem. There is something to be said for remaining "underground" if you have a small collection.

CONTRIBUTIONS TO THE STATE OF THE ART

Breeding reptiles in captivity would still be doubtful if it wasn't for the spirit of sharing that brings us all together here. All of us can recall a word, hint, or direct instruction that has helped us, and most of us feel good when we can help another. I could tell you about the countless (wishful thinking) eggs that I hatched this year, and be considered a snake guru of sorts or I can admit to producing a large number of bad eggs, and feel very incompetent. I am making a limited admission to both. I am doing some things right and I am doing some things wrong. Maybe there are ten important items involved, any combination of four will insure breeding success. I don't know all the answers yet. But I am keeping detailed records and hope that by comparing my notes with other breeders' notes, some solutions to as yet unsolved problems can be reached.

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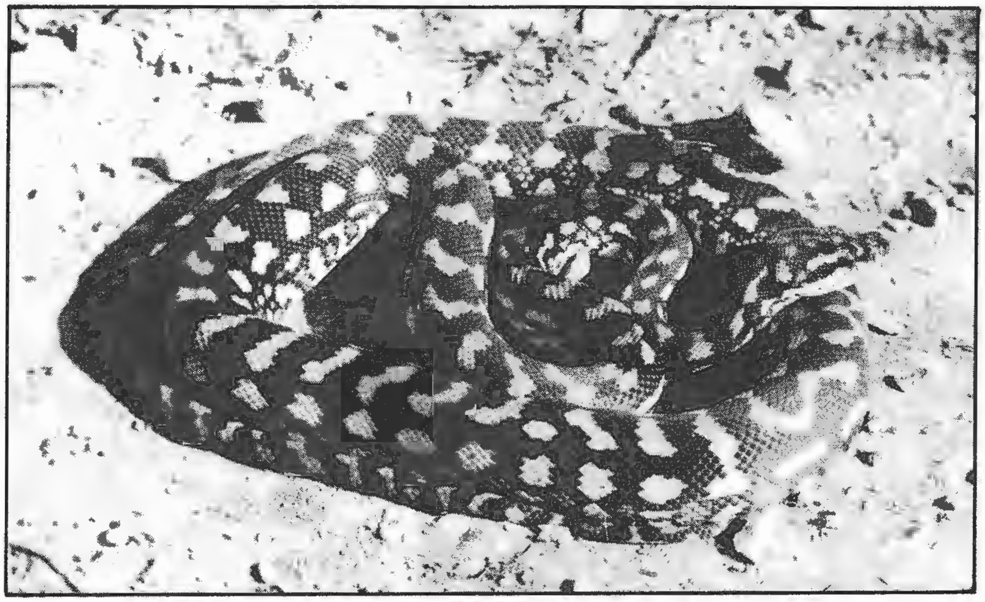
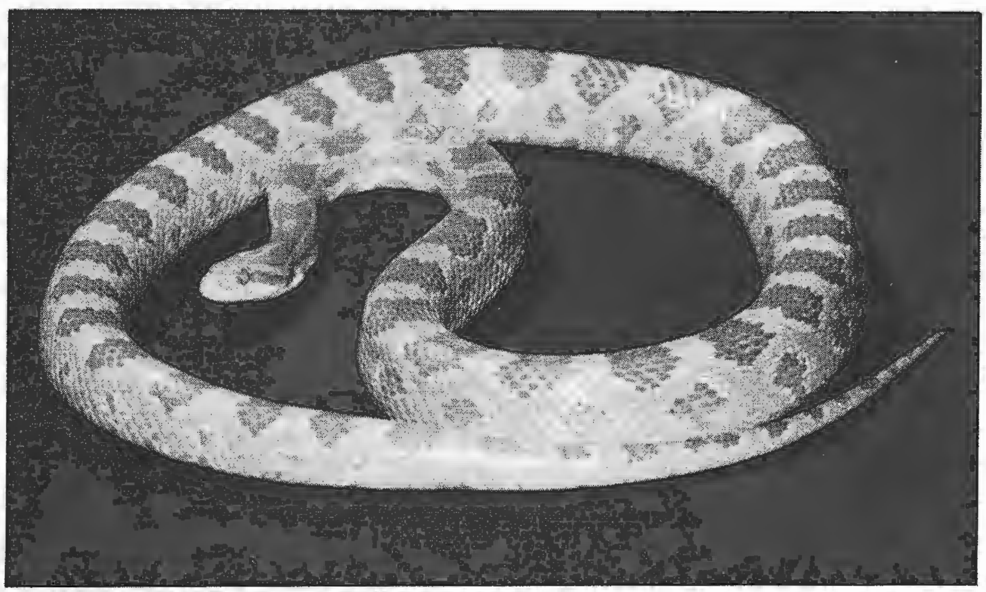
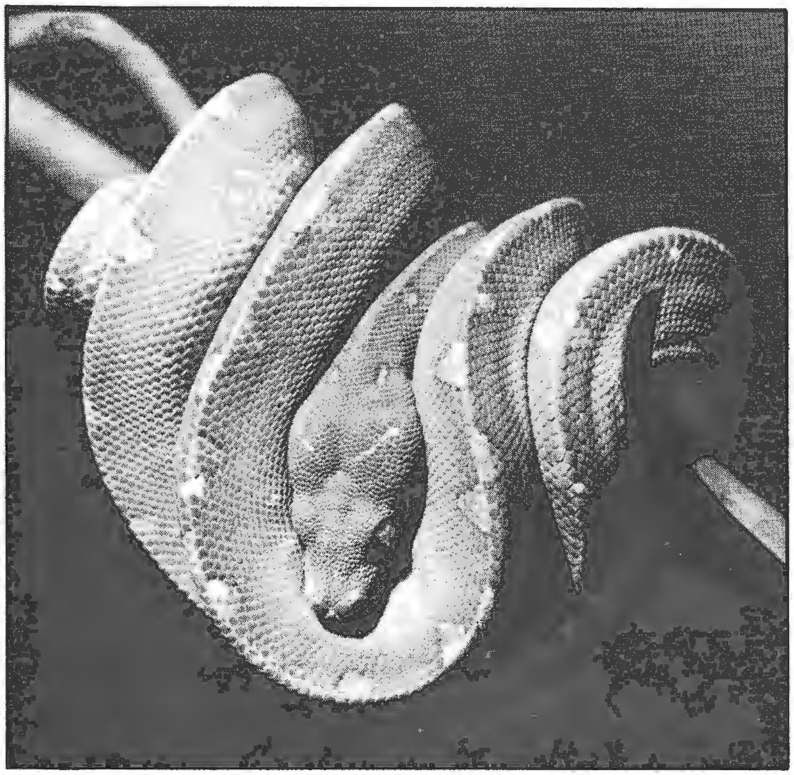


Carpet python (Morelia spilotes). Photo by D. McCampbell.



Amelanistic corn snake (Elaphe guttata). Photo by D. McCampbell.



Green tree python
(Chondropython viridis).
Photo by D. McCampbell.

Sonoran black kingsnake
(Lampropeltis getulus nigritis)
Photo by D. McCampbell.

CAPTIVE HUSBANDRY OF WILD-CAUGHT EMERALD TREE BOAS

Ernie Wagner

During the last few years, wild-caught emerald tree boas (Corallus canina) have become much more readily available and less expensive than in the past. With the addition of several of these snakes to my own collection, I encountered a series of specific problems which caused me to change the way in which I housed and cared for them.

Upon receiving a new emerald tree boa, I would always examine the snake for any sores or injuries, check the mouth to make sure it was clean, and then expose the snake to a "No-Pest Strip" overnight to eliminate any mites which it might be carrying. In the past, I would then set the snakes up in groups of three or four, in large cages which had attached branches and a substrate of fir shavings. Under these conditions I discovered that it was difficult to get food to all the snakes and that some would overeat and regurgitate if the amount fed was not controlled. In addition it was not unusual for them to get shavings in their mouths when they struck at a rodent and missed; and because of the length of their teeth, it was difficult for them to clear their mouths.

Moving the snakes for any reason also became a chore, involving considerable disruptions and stress on the snake. They held onto branches so well that snake sticks really did not work. In the end, I found myself just grabbing the snake behind the head whenever I wanted to move it. The final straw came when I discovered that several of these snakes had mouth rot. I believe the mouth rot was caused from stress since I had eliminated shavings and gone to newspaper.

At this point, the advantages of housing the animals individually, along with the need to develop a nonstressful method of moving them, had become obvious. To do this, I took a series of 10-gallon aquariums and fitted each end with a "U" shaped clip which I attached to the glass with silicon rubber. I then took a one and one-half inch wooden dowel and cut it to length so it would rest inside the aquarium on these cips, but could be easily lifted out. I cut screen lids and attached "Velcro" fasteners to both the screen and the glass of aquarium and then added a newspaper substrate and a water jar to complete the arrangement.

To facilitate a series of these tanks, I constructed a series of shelves in one corner of the room and then draped the whole area with translucent plastic. A room humidifier placed on the floor inside this area could then be used to raise the humidity of this whole area. Two heat lamps mounted outside the plastic but shining at the tanks provided heat. Using a thermostat I am able to obtain 85^{O}F during the day, and then drop the temperature to about 80^{O}F at night. I usually humidify this area on the afternoon of the day I plan on feeding, and then I feed live mice or rats to the snakes after the lights have gone out. Lighting in this area is provided by a four-foot fluorescent light fixture attached to the ceiling. I eventually added a 15 watt incandescent light over each tank on the bottom two rows in order to bring their tempertures up to the required 85^{O}F during the day.

Under these conditions the emerald tree boas are doing much better, although there are still some problems which have to be watched for and managed. While shedding the snakes seem to need extra humidity, so I add a plastic tray of damp shavings, cover the cage with plastic and spray the snake daily until it has shed. If there is an incomplete shed, the snake is soaked in water for half an hour and then shed by hand. Regurgitation is an occasional problem in newly arriving snakes and I am generally able to resolve it by treating with Emtryl^{R} and then starting the snake out on small meals. Lack of appetite can also be a problem in newly arrived snakes and, if they refuse rodents, I will try chicks. If nothing works and the snake appears thin, I will force-feed several meals, and usually the snake will begin feeding on its own. I force-feed a combination of skinned, blended mice, egg yolk, and powdered vitamins, and administer it to the snake with a basting tube.

As mentioned above, the most irritating problem with these snakes is their propensity for developing mouth rot. Newly arrived snakes that are doing only marginally well, feeding occasionally but are still thin, or animals that are not feeding must be watched closely for this problem. The difficulty is that they will develop a case of mouth rot and not show any gaping or other symptoms until it is really advanced. Because of this, I have a rule that any snake that refuses food twice in a row is checked for mouth rot. If caught in the early stages, it is much easier to treat. I usually begin treatment with topical application of Polysporin[R] ointment on a daily basis until it has cleared up. Animals which do not respond to topical treatment need systemic treatment with antibiotics, depending upon culture results. Snakes that have settled into captive conditions, are feeding well, and have good weight rarely develop this problem.

Under my new conditions I am now able to clean the snakes and move them about from tank to tank with a minimum of stress as they will usually ride on the dowel without even looking around. This type of caging is more work to maintain than the old system but has resulted in healthier snakes. I find it takes newly imported emerald tree boas a very short time to settle into the routine of being moved from tank to tank during cleaning, but it usually takes about four to five months for them to gain enough weight to resemble long-term, healthy, captive-bred snakes. At this point, they can be placed together in a group for display or reproductive efforts, and they should continue to thrive.

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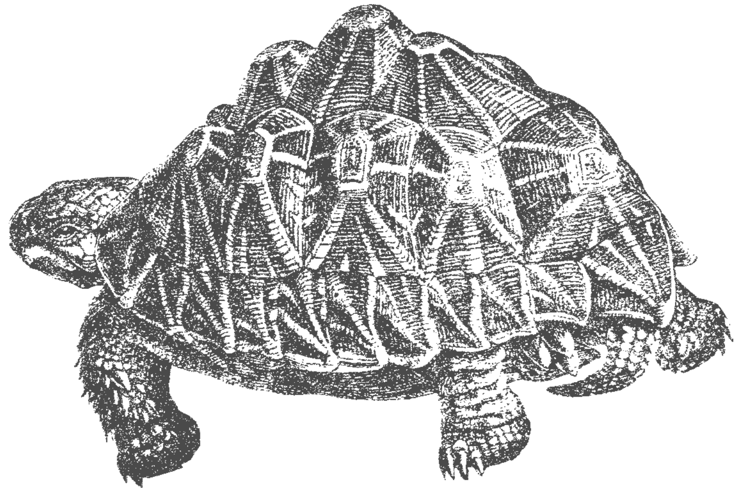
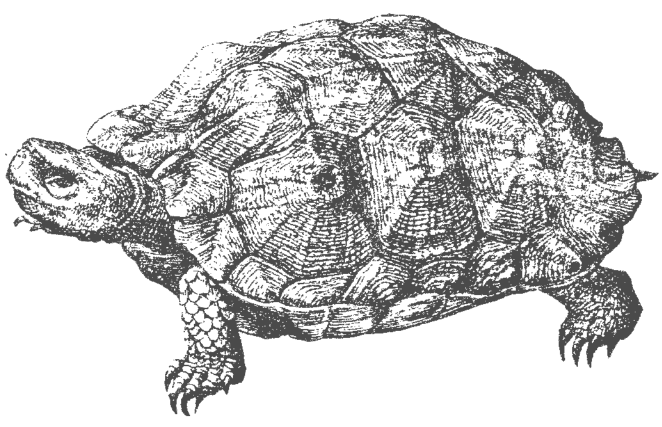




CAPTIVE HUSBANDRY AND PROPAGATION OF TORTOISES

Brett C. Stearns

INTRODUCTION

This paper reviews the basic requirements for maintaining tortoises in captivity and specifically addresses such topics as housing, diet, medical problems, breeding, egg incubation, and care of hatchlings. Also, a survey of tortoise types is made, with observations regarding their suitability to captivity. Although the paper's broad scope has been at the expense of some detail, it is hoped that it will nonetheless be useful to those inclined to keep tortoises.

SURVEY OF TORTOISE TYPES

The world's tortoises may, for simplicity's sake, be divided into four groups: the genus Gopherus; the genus Testudo; the genus Geochelone; and the various small tortoises from Africa and Madagascar.

Undoubtedly, the tortoises most familiar to herpetologists in the United States are of the genus Gopherus, the only tortoises native to North America. Perhaps the most familiar of these is the desert tortoise (Gopherus agassizi). Prior to being protected, large numbers of these tortoises found their way into captivity and, according to any reasonable estimate, are still in many backyards scattered throughout the West. Others of this genus are: the Texas tortoise (G. berlandieri), smaller than, but similar in habits and appearance to the desert tortoise; the gopher tortoise (G. polyphemus) from southeastern United States; and the Bolson tortoise (G. flavomarginatus), from northern Mexico, largest and rarest of the genus.

The genus Testudo is comprised of nine tortoises. Most common in collections are: Herman's tortoise (T. hermanni) from Europe; the marginated tortoise (T. marginata), from Greece; Horsfields' tortoise (T. horsfieldi) from Russia and the Middle East; and the spur-thighed tortoise (T. graeca) from Spain, southern Europe, North Africa, and the Middle East.

The genus *Geochelone*, in terms of variety and types of specimens, distribution, and size of individuals, is the largest. Most noteworthy are the Galapagos tortoises (*Geochelone elephantopus*) and the Aldabra tortoises (*G. gigantea*) but others reach remarkable sizes. The largest non-insular tortoise is the African spurred tortoise (*G. sulcata*). These tortoises are known to reach lengths of at least thirty inches and weights of at least two-hundred pounds. They were once fairly common in captivity in Europe, being imported primarily into France from the then French colonies in North Africa. They have always been rare in the United States and, even today, few are in private collections and even fewer are in zoos. Given proper care, they take readily to captivity, and are aggressive and active--perhaps the most of any tortoise. They have been bred in the United States only at the San Antonio Zoo and by the Institute for Herpetological Research. An African tortoise which occurs more frequently in zoos and private collections is the leopard tortoise, (*G. pardalis*). While not attaining the size of the African spurred, the leopard tortoise is still large in its own right.

Madagascar's representatives of *Geochelone* are the radiated tortoise (*G. radiata*), perhaps the most attractive of all tortoises, and the angulated tortoise (*G. yniphora*), considered to be the world's rarest tortoise. Unfortunately the latter has been bred only once outside its range--recently at the Honolulu Zoo. A concerted effort is now being made to insure the continued existence of this tortoise.

South America (excluding the Galapagos Islands) has three representatives of this genus, the least known of which is the Argentine tortoise (*G. chilensis*), a small tortoise of uniformly brown color. In recent years, it has been imported into the United States in disturbing numbers. Frieberg (1981) divides this species into *G. chilensis*, *G. petersi*, and *G. donosobarrosi*, and they do, in fact, seem significantly varied in appearance. The red-footed tortoise (*G. carbonaria*) and the forest or yellow-legged tortoise (*G. denticulata*) are similar to each other in behavior, appearance, and maintenance requirements, although G. denticulata reaches lengths in excess of twenty-four inches, whereas G. carbonaria rarely exceeds sixteen inches. These tortoises are active, personable, and do well in captivity. As they are tropical tortoises, they are fond of fruit and partial to damp, humid environments.

The balance of the genus comes from Asia and nearby islands. India claims three of these, one of which is the star tortoise (G. elegans). Large and dramatically patterned star tortoises also occur on Sri Lanka and a smaller star tortoise (G. platynota), comes from Burma. The latter is rare in captivity and its habits are not well known. India is also home to the Travancore (G. travancorica), and the elongated (G. elongata) tortoises. These are similar except that G. travancorica lacks the nuchal scute which is almost always present in G. elongata. Closely related to the elongated and Travancore tortoises is G. forsteni, principally from the Celebes Islands. Finally, there are three species from Southeast Asia: the Burmese brown tortoise (G. emys); the closely related G. nutapundi; and the impressed tortoise (G. impressa). The latter is not known to thrive in captivity and is seldom found in collections. The other two do well in captivity, and have bred quite readily. The Honolulu Zoo has experienced considerable success breeding these tortoises. They become large, sometimes weighing up to one hundred pounds. G. nutapundi has recently been taxonomically separated from G. emys based on differences in the configuration of plastral scutes.

Seven genera from Africa, south of the Sahara, and from Madagascar are relatively small--some mature at no more than four or five inches. The tortoises are: the bowsprit tortoise (Chersine angulata), from South Africa; the spider tortoise (Pyxis arachnoides), and the flat-shelled tortoise (Acinixys planicauda), from Madagascar (combined by some authorities into a single genus); the pancake tortoise (Malacochersus tornieri), from East Africa; Psammobates and Homopus from South Africa; and the hinged-back tortoises (Kinixys) from East and West Africa. With the exception of the pancake and hinged-back tortoises, these are rarely found in collections as most are threatened in the wild and thus have been legally protected.

Several tortoises seem to be particularly well-suited to captivity. Desert tortoises come to mind due to their popularity in years past, but they are now protected and no longer legally available. However some Testudo (e.g., T. hermanni, T. graeca) are available and are also well-suited to captivity. Generally from temperate regions, they are more tolerant to colder temperatures than are tortoises from desert or tropical areas. Under proper circumstances, they can be induced to hibernate. As a rule, none of the small African tortoises should be kept since little is known of their requirements and, with few exceptions, they have not thrived in captivity. Several Geochelone are endangered, and therefore unavailable. Of those available, perhaps most easily maintained are the red-foot, yellow-leg, leopard, and elongated tortoises. All have a varied diet, grow quite rapidly, and often breed readily in captivity.

HOUSING

I admit to a strong bias in favor of maintaining tortoises outside if weather, security, and housing space allow. However, keeping tortoises outdoors presents the keeper with numerous decisions and considerations as different tortoises have different requirements. Red-foots and yellow-legs, for instance, prefer damp and dark areas and thus large, leafy plants and mudholes should be provided. On the other hand, tortoises such as the African spurred, desert, and leopard prefer open, sunny areas and should be provided with a grassy expanse upon which to graze. However, these also need shady areas, as all tortoises must be able to escape the sun during the heat of the day. Shade is best provided by planting trees or shrubs in the enclosure.

Water should be constantly available in a shallow container with gradually sloping sides which allows tortoises to move in and out of it freely without risk of turning over. Water-proof cement may be used to form the container. Water should be changed frequently since tortoises often defecate in it.

Smaller tortoises should be kept in a predator-proof enclosure with a welded wire top which permits sunlight and fresh air to pass through, yet keeps out birds, cats and racoons.

Whether to leave a tortoise outside overnight is essentially a function of its type, size and overnight low temperatures. Some tortoises are more cold-resistant than others; for instance, *Testudo* may remain out in weather too cold for tropical types such as *G. carbonaria*. Generally, the larger the tortoise the less susceptible it is to cold weather as long as there is adequate warmth during the day. The body mass of large tortoises absorbs considerable heat during the day, much of which is conserved overnight. On the other hand, smaller tortoises cool more quickly and are, therefore, more at risk. Tortoises may be acclimated to cooler weather but the process, a gradual one, should not be started when the weather is cool, but be deferred until summertime when evenings are consistently warm. I have acclimated large tortoises over many years to the point that they have remained outside with overnight temperatures as low as 40°F. They have showed no ill effects and, during the days following these cold evenings, have actually been active and feeding. Tortoises being acclimated to cooler weather should be monitored daily. This procedure, while not necessarily recommended, is mentioned to illustrate the acclimation ability of larger tortoises.

Another decision keepers may confront is whether to allow a tortoise to hibernate. First it must be determined if the tortoise is of a type that will normally hibernate. Some that will are the desert tortoise and those of the genus *Testudo*, whereas most, if not all, of the genus *Geochelone* are not inclined to hibernate and should be kept active year-round. Only healthy tortoises should be allowed to hibernate; those that are ill or malnourished may die in hibernation. If your tortoise is a type that hibernates in the wild, it will most likely dig a burrow as the weather starts to cool. Be sure it does not do so near a fence--it may emerge on the other side or in an area that may become flooded by rain. Alternately, tortoises may be hibernated in a cardboard box or doghouse in a cool, dry area such as a garage. Keep the garage below 50^{o}F so that the tortoise remains inactive. Tortoises kept active during winter should be fed and watered as usual, although food consumption will, undoubtedly, be reduced. Check tortoises emerging from hibernation for signs of an illness that may have developed or worsened during hibernation. Soak them several times in warm water as they are usually inclined to drink. Additionally, food may be offered although several days may pass before they eat.

Housing tortoises inside also presents the keeper with a variety of aspects to monitor and decisions to make, such as type and size of enclosure, type of substrate, the manner in which water will be presented, the manner in which temperature and humidity will be maintained and monitored, and the type and number of tortoises to keep in an enclosure.

Glass aquariums or wooden cages are adequate indoor enclosures for smaller tortoises; a portion of the garage converted into a pen, for larger tortoises. Every enclosure should be large enough to allow freedom of movement and creation of a temperature gradient. By having one end of the enclosure hotter than the other, tortoises can seek preferred temperatures (thermoregulate). The hot area may exceed 100^{o}F but the temperature should gradually decrease to about 70^{o}F at the other end of the enclosure. A good average temperature is around 85^{o}F. Heat may be provided by heatpads or tapes placed under smaller enclosures or by overhead heat or spotlamps. Indoor pens may be heated by a bank of heatlamps secured so they won't fall on the substrate. A sheet of plywood above the lamps helps retain heat. It is good practice to keep a thermometer (preferably a high-low type) in the enclosure and, in larger enclosures, one at both ends. All lamps may be placed on a timer and left on daily from twelve to fourteen hours, but should be off at night to provide a photoperiod. If it is necessary to heat the enclosures overnight because they will otherwise get below 55 or 60^{o}F, do so by non-illuminated heat sources (e.g., heatpads and tapes). Large tortoises maintained in a garage pen may not need supplemental heat at night if they are allowed to become thoroughly warm during the day, unless garage temperatures fall below 50^{o}F at night.

Humidity for tortoises that require or prefer it may be created by spraying down or keeping a water dish in a covered enclosure. Don't keep tortoises requiring a dry atmosphere with those requiring humidity, as the former may develop medical problems.

Keeping tortoises, particularly young ones, on hard surfaces such as concrete may cause leg problems. Inorganic substrates such as rocks, sand, or gravel should not be used as tortoises tend to eat these along with food. This may cause intestinal blocks leading to death. Use a digestible substrate such as alfalfa pellets for larger tortoises and Litter Green cat litter for smaller tortoises. A minimum of an inch and one half layer should be provided, more for larger tortoises. Larger tortoises penned in a portion of the garage should not be kept on a cement floor but should be provided a substrate of alfalfa or prairie hay. Though this will get messy and needs occasional changing, the effort is justified in terms of healthy tortoises.

As tortoises don't like to be constantly on display, provide a hiding place. Large flower pots, split in half, may be used for smaller tortoises; a retreat made out of wood in one portion of the pen for larger tortoises.

A dog bowl or plastic tub containing a brick to keep it upright is an adequate water container for larger tortoises. A shallow pan may be used for smaller tortoises. Place a low, flat rock in the pan so that tortoises cannot become immersed in the water and drown. Change water regularly to insure cleanliness. If tortoises from arid areas are being kept, do not keep a water pan in their covered enclosure if covered, as it will create unwanted humidity. Instead, remove the tortoises from the cage weekly and soak them in warm shallow water in a tub.

It is ill-advised to keep dissimilar tortoises in the same enclosure. Small tortoises can be dominated or overturned by larger tortoises. Avoid mixing those of an aggressive nature with those of a shy nature as the former may dominate and get most of the food. Also avoid overcrowding, which causes the enclosure to be fouled more quickly and reduces the possibility that all the tortoises are receiving adequate food.

Tortoises kept indoors for any length of time should be provided with a broad spectrum light source such as Vita-Lites (by Duro-Lite Lamps, Inc.) and black lights. These fluorescent tubes, of various wavelengths, should be used in conjunction with each other, placed between fifteen and twenty inches above the substrate, and left on up to twelve hours a day.

FEEDING

What and how often to feed tortoises is often a source of frustration. Some tortoises will eat a wide variety of food, while others are very selective and frequently refuse to eat. Offer a wide choice of foods and experiment with new ones; but don't fail to offer a different food on the assumption that it will be refused. A caution must be made about lettuce, as tortoises readily come to rely on it, to the exclusion of other foods. This should be avoided as lettuce alone does not provide enough nutrition. However, a moderate amount of lettuce, particularly romaine, is beneficial, but should be offered more as an afterthought than as the main staple. In other words, lettuce may be fed after tortoises have eaten more substantial foods.

The following diet used by the Forth Worth Zoo for Aldabra tortoises demonstrates the need for variety:

1 head of cabbage
½ head of lettuce
2 heads of Romaine, when available, instead of lettuce
1 handful of alfalfa
6 stalks celery or celery tops
2 squash
3 carrots or 1 sweet potato, raw and sliced
1 small Irish potato
6 bananas
3 oranges
6 apples
1 cup dogchow, not soaked
1 cup monkeychow, not soaked
1 cup bird of prey diet

Not all tortoises will accept all these items, but this diet indicates the variety needed to maintain proper nutrition.

Examples of some unlikely foods which tortoises have eaten are radishes, spring green onions, large onions, and green peppers. Other good foods include parsley, strawberries, zucchini, melon of all types, peas, and beans--in short, any mixture of vegetables and, to a lesser extent, fruit.

A diet too heavy in fruits, especially bananas, will often cause loose stools, although this does not seem to apply to red-foots and yellow-legs, which undoubtedly have a large fruit intake in the wild. Tortoises such as the desert, leopard, and Argentine should have their fruit intake kept in check. The best vegetables seem to be the rougher ones such as squash, green beans, and broccoli; and leafy greens such as chard, spinach, and small amounts of cabbage. Larger tortoises may be fed a diet of alfalfa, supplemented with fruits and vegetables. Most tortoises take readily to backyard grasses and weeds.

The protein content in dogfood is too high for tortoises; however, many tortoises eat dogfood readily and thus some keepers include too much in the diet. This can lead to several undesirable results, one of which is a sometimes fatal buildup of hardened uric acid, often in the bladder. The unattractive pyramiding effect of the carapace that captive tortoises often demonstrate is another. A small amount of dogfood, fed once weekly, is acceptable.

During warmer months animals kept outside should have food available at all times. This, in simplest form, can be a matter of grasses and weeds in the backyard, supplemented by fruits and vegetables. Tortoises housed indoors should be offered food about three times a week. Food scattered randomly on the floor of the enclosure will often be trampled and defecated upon by tortoises; therefore, place the food in a bowl or other container. Larger tortoises may be fed food placed behind a grate or grillwork through which they can reach to obtain the food but which precludes them from walking on it.

Many keepers recommend the use of diet additives such as vitamins and bone meal. One such product is Vionate (manufactured by Rich Health, Inc.) which, in powdered form, can be sprinkled on top of the food. It has been reported that excessive amounts of bone meal can cause an intestinal block and, therefore, a caution on its use should be observed. While these additives may be of some benefit, they are not a substitute for a well-balanced, nutritious diet and ample sunshine.

BREEDING

Certainly the most rewarding aspect of tortoise keeping is a successful breeding program. Unfortunately, this is often difficult to achieve. As the differences between sexes are often subtle, it is not always obvious whether you have a male or a female. One indicator is that male tortoises generally have concave plastrons. The extent of the concavity is variable; some are extremely concave, while others present much less difference between the sexes. Likewise, tail size is usually an indication, as males have larger tails. Body size is not always an indicator, as you may not have fully grown individuals. Additionally, the male of certain species can be substantially larger than the female whereas, in other species, the reverse is the case. It is preferable to keep more than one male and several females for a breeding colony; two males provide competition and stimulation for each other.

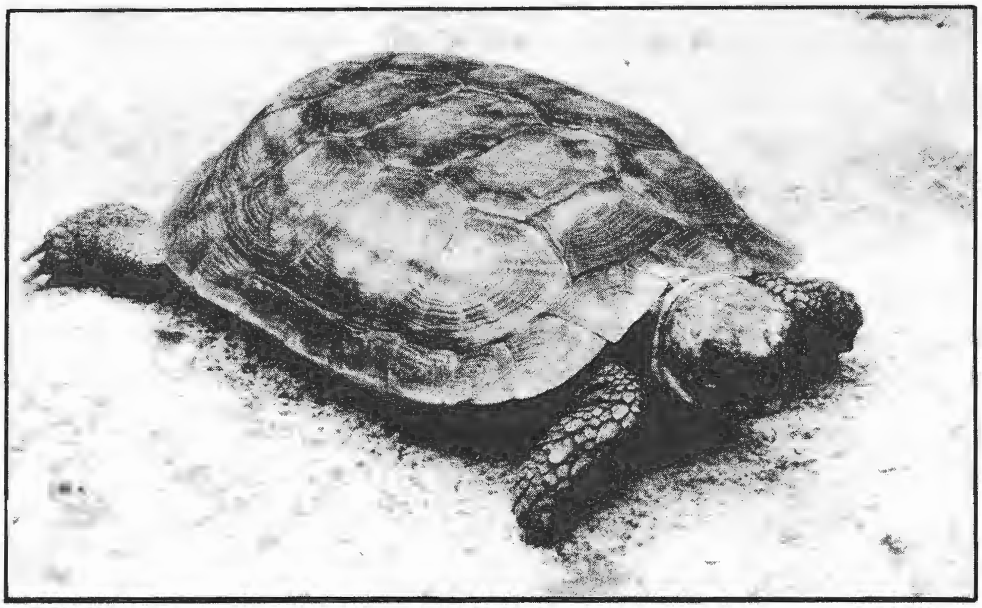
Breeding is more likely to occur outdoors than indoors and will probably occur seasonally. Warm weather stimulates breeding and, with species such as the red-foots and yellow-legs, warm rains or spray from a hose or sprinkler will often stimulate breeding. If the male shows little or no interest in the female, separate them for awhile and then reintroduce them. Outdoor enclosures should include a nesting area of sandy, loamy soil to facilitate egg laying.

Do not get discouraged as breeding programs can take time to establish. When you least expect it you may get a clutch of eggs, whereas with things apparently in good order, you may have no results whatever. Much is not known about captive breeding of tortoises but at a minimum, keep them healthy, well-nourished, in the best set up possible and, again, be patient.

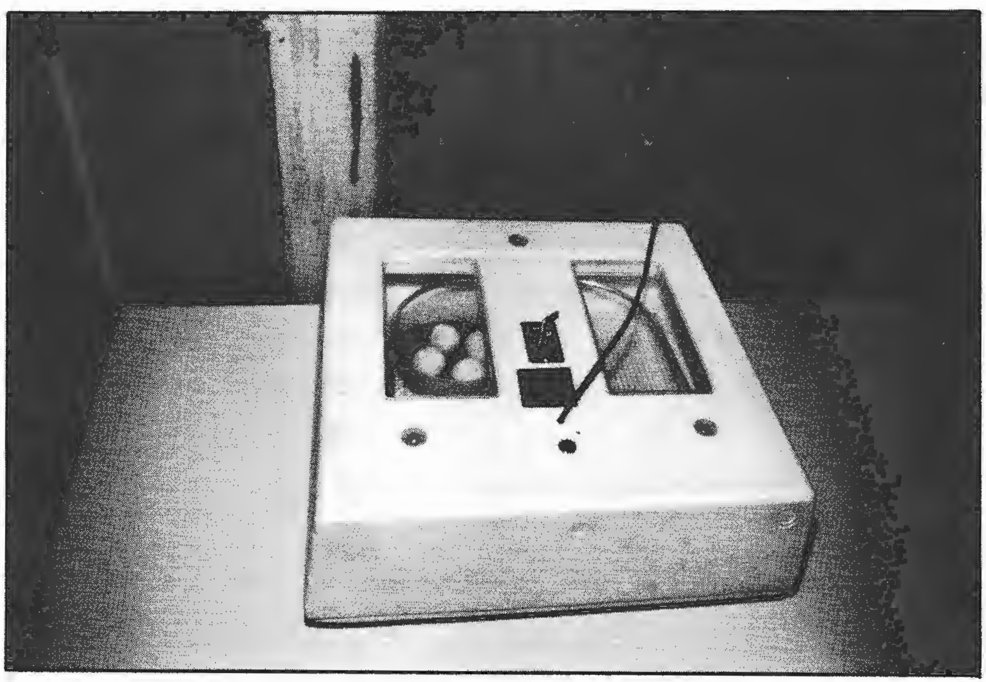
INCUBATION

Incubation of tortoise eggs is by no means a perfect science. Many methods have been used, successfully and unsuccessfully. The Taronga Zoo in Sydney, Australia successfully incubated Aldabra tortoise eggs in a plastic container, lined with damp paper towels. The container was kept in a dark area of the reptile house which had an interior temperature of between 80^{o} and 85^{o}F. Two eggs hatched after one hundred and sixty two days; seven more hatched over the next twenty-one days.

The San Antonio Zoo has used a variation of this method to successfully hatch Geochelone sulcata. Vermiculite and water, mixed equally by weight, are placed in plastic shoeboxes. Eggs, marked on top to indicate the position in which they were laid, are placed in this mixture with the bottom third of each egg settled into it. Boxes are covered and kept



African pancake tortoise (Malacochersus tornieri). Photo by D. McCampbell.



Chicken incubators can be used for hatching tortoise eggs. Photo by B. Stearns.

in the reptile house at about 82°F. Once a week the tops of the boxes are removed for two hours to facilitate the interchange of fresh air, but no additional water is added. The incubation period, though quite variable, averaged four months.

A private breeder has had steady success incubating leopard tortoise eggs in an incubator set at about 87°F. Eggs are placed on damp vermiculite and hatch, in approximately four months, although incubation times ranged from eighty-eight days to about seven months. On occasion, there has been more than a month's difference in hatching time with eggs from the same clutch incubated side by side in the same incubator.

The San Diego Zoo has hatched Galapagos tortoises, incubating eggs at about 82°F, with hatchlings emerging from ninety-six to one hundred and twenty-four days later. The National Zoo has successfully incubated red-foot eggs over a one hundred and sixty-five day period.

The Institute for Herpetological Research has successfully incubated Geochelone sulcata eggs using two different methods, the first of which is a variation on the shoebox method of the San Antonio Zoo. Covered shoeboxes, containing eggs on moistened vermiculite, are placed on a layer of alfalfa pellets in an aquarium, with a heatpad underneath it to provide warmth. The layer is of a depth which maintains a temperature of between 82 and 87°F. In the second method, eggs are placed on moistened vermiculite in small, open plastic cups that in turn are placed in a chick incubator set at 82°F. The incubator top is removed weekly for fresh air. These methods resulted in a high hatch rate with variation in incubation times from about 90 to 170 days. Since incubation times are extremely variable, eggs that have not hatched should nonetheless be incubated until it is certain they are infertile.

CARE OF HATCHLINGS

Hatchlings require special care and constant attention. They should be fed daily, given a vitamin and calcium supplement (such as Vionate), housed inside, measured and weighed periodically to monitor growth, placed outside regularly to obtain sunlight, and soaked in warm, shallow water twice a week.

If, after hatching, their yolk sac is still relatively large, hatchlings should be placed on a non-abrasive, moist surface such as damp paper towels, in a shoebox. Keep them this way until the yolk sac is absorbed, usually within two or three days.

Since some hatchlings will eat within a day or so after hatching, food should be offered almost immediately, even though others may not take food for up to two weeks. Diet may consist of leafy greens, such as raw spinach, broccoli leaves, and romaine lettuce; and chopped fruits and vegetables such as tomatos, apples, papaya, broccoli, parsley, beans, peas, bananas, and carrots; and, of course, grasses and weeds.

Hatchlings placed outside in warm, sunny weather should be in a draft-free enclosure, which is covered to protect from birds, cats, skunks, and the like. A portion of the enclosure should always be shaded.

Hatchlings should be kept indoors at least for the first year, in an aquarium on a substrate of Litter Green at about an inch and a half deep. Litter Green absorbs wastes, provides ample traction, and may be eaten without harm by hatchlings. Do not use inorganic substrates (e.g., sand or gravel) as these are undigestible and are often taken in by hatchlings while eating; this can cause death by impaction. Constipation may be precluded by allowing hatchlings to soak and drink in a tub of shallow, lukewarm water twice a week.

Provide heat by a heat or spotlamp at one end of the enclosure, by a heatpad or tape under a portion of the enclosure, or by a combination of both. Hatchlings should not get below about 70°F at night. Heatpads or tapes under the aquarium will maintain warmth in the enclosure though the room drops below these temperatures overnight. During the day, heatlamps create a thermogradient (e.g., 70°F to 100°F), which allows hatchlings to select their preferred temperature. Automatic timers may be used to control the lamps, pads, and tapes.

MEDICAL CONSIDERATIONS

Medical problems affecting tortoises include internal and external parasites, eye problems, and respiratory ailments.

The most serious problem is pneumonia, which is usually caused by some combination of excess humidity, damp and drafty confines, or insufficient heat. Symptoms are a nasal discharge and watery eyes (however, as some tortoises, such as <u>G. carbonaria</u>, normally have watery eyes, this is not always an indication). Many antibiotics have been used with varying degrees of success. A sick tortoise should be kept at 90-95^{o}F, have ample exposure to sunlight if possible, kept out of drafts, and offered food. However, sick animals often lose their appetite; thus, the fact that it does not eat is not, in and of itself, cause for alarm.

With severe respiratory problems, a culture should be made before an antibiotic is administered. Once the bacteria type is determined, the appropriate drug may then be selected. Squeeze all air bubbles out of the syringe before injecting. After injection, usually in the tortoise's "hind quarters", withdraw the plunger slightly; if blood is withdrawn into the syringe, remove the needle and inject at another location.

A variety of antibiotics has been used to treat pneumonia. However, recent developments indicate that some of these are not necessarily the drug of choice. The reader is referred to Ross (1984) for excellent, current information in this area. Antibiotics that have been used, and their doses (based on total weight, including the shell), are:

Ampicillin	50-100 mgs/kg/day
Tylosin	25-50 mgs/kg/day
Chloramphenicol	50-100 mgs/kg/day
Tetracycline	25-50 mgs/kg/day
Gentamicin	10 mgs/kg/48 hours

Amikacin	10	mgs/kg/48 hours

Amikacin has a wide spectrum and is effective against gram-negative bacteria such as Pseudomonas. It is an aminoglycoside and organisms found to be resistant to other aminoglycosides, such as Gentamicin, have been found to be susceptible to Amikacin. It is also found to be active against some gram-positive organisms but probably would not be the drug of choice with a gram-positive problem. These drugs have proven toxic to some reptiles. They should be used only after consulting an expert or proper literature.

For internal parasites, culture the fecal matter, then treat with injections of levamisole phosphate (Tramisol) diluted one part to eight parts water. The dose is .01 cc per ounce of body weight. Three shots should be given, ten days apart. External parasites may be gently removed with tweezers.

Eye problems are usually remedied by applying Polysporin ointment twice a day directly to the eyelid.

Uric acid buildup (cystic calculitis) is another, and potentially fatal, problem. The symptom is the change in the consistency of the white uric acid discharge from a gelatinous one to a coarse, granulated one. Suspected causes include insufficient water intake and/or excessive amounts of protein in the diet. Treatment would certainly include making water available through frequent soakings and reducing, or preferably eliminating, protein-rich foods, such as dog food, from the diet. The cystic calculitis often is revealed by an x-ray. X-rays through the shell may require up to three times the normal dose; if through the soft parts, use the same dose as for humans, dogs, or cats.

It is important to keep records on your tortoises. Hatchlings and those newly-acquired should be measured and weighed monthly; those that have done well in your collection for some time, less frequently. Record this information and refer to it as needed.

CONCLUSION

The foregoing review of captive management of tortoises is broad in scope. Major topics have been covered in a general sense; less critical topics have not been covered at all. However, it is hoped that this paper, which addresses many of the problems involved in keeping tortoises, will benefit tortoises and their keepers.

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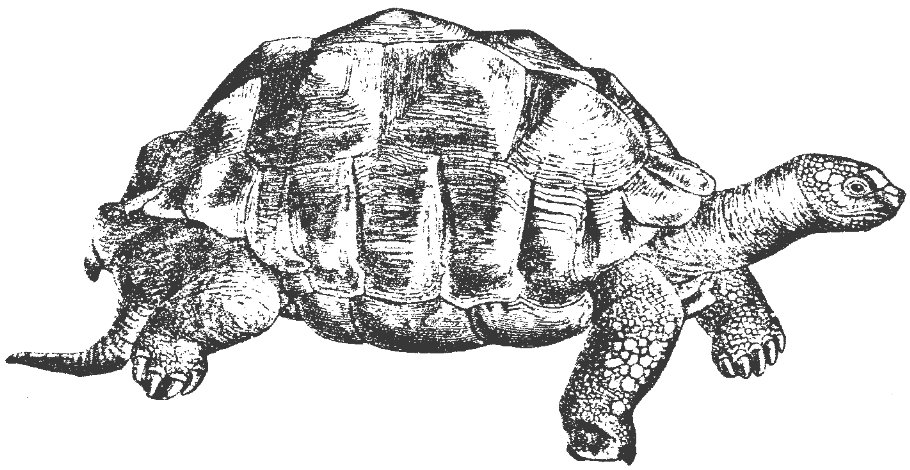
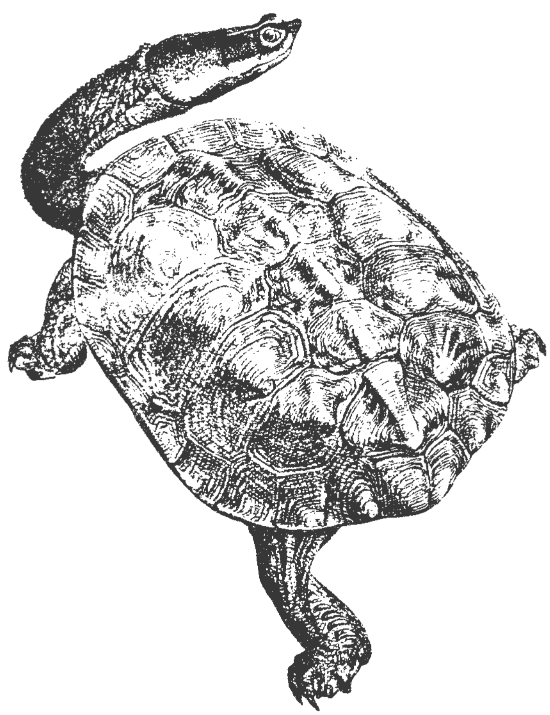




CAPTIVE PROPAGATION AND HUSBANDRY OF LEOPARD TORTOISES

Rochelle Freid

Between July and November of 1979 a female leopard tortoise (Geochelone pardalis) laid six clutches containing a total of 60 eggs. The eggs were collected as laid, placed in styrofoam egg cartons, and put into an incubator set at 85°F. I had seen a male mating with this female and hoped that these eggs would be fertile.

Upon checking the incubator toward the end of November 1979, I noted that some of the eggs from Clutch I were hatching. The hatchlings were left in the closed egg cartons in the incubator until the egg sac was completely absorbed. The hatchlings were then removed from the incubator and placed in a heated terrarium. Each hatchling was identified by letter marked on its shell with an indelible marker, and its shell design, which is unique in each animal, was diagrammed to aid in identification.

A total of 40 young hatched from the 60 eggs, giving a hatch rate of 66 percent. Two of the animals, born with deformed shells, died within the first year, but the other 38 animals are alive and thriving five years later. There was great variation in the number of days the eggs took to hatch. Some eggs took as few as 118 to 120 days to hatch and some took as long as 155 to 160 days to hatch (see Table I). There was also a wide variation of hatching times within each clutch.

Table 1. 1979 Hatching Data for Leopard Tortoise Eggs

Clutch No.	Date Laid	No. of Eggs	No. Hatched	(%)	Incubation Time Days	(X)
1	July 7	11	5	(54)	137-159	(141)
2	Aug. 4	12	7	(58)	124-143	(134)
3	Aug. 26	10	8	(80)	120-160	(138)
4	Sept. 16	9	5	(56)	132-161	(148)
5	Oct. 12	9	5	(44)	118-137	(127)
6	Nov. 10	10	10	(100)	131-152	(137)

HOUSING.

The hatchlings were removed from the incubator and placed in large heated terrariums. Each tank contained a large hot rock type reptile heater, a thermometer, and a light. Incandescent lighting was used, as I have raised several species of exotic tortoises using these lights with satisfactory results. Of course, the mild climate in southern California allows me to carry the animals outdoors for some sun several times a week during most of the year. Litter Green, a cat litter product made of small pellets of pressed alfalfa, was used as a substrate. If this product is inadvertently ingested by the animals, it is completely digestable. As the tortoises grew older and were moved to much larger terrariums, I began to use rabbit food pellets as the substrate material since this was considerably less expensive in the large quantities I was using, and seemed to work nearly as well.

During the day, I kept the lights on in the terrariums and attempted to maintain a temperature of 85^{O}F. At night the lights were turned off and the terrariums were covered with blankets. The hatchlings readily learned to sit on the hot rocks for a source of additional warmth. A small cardboard box was placed over the hot brick to serve as a hideout for the hatchlings. Water was not kept in the tanks as I found that if saucers of water were left in the tanks, the tortoises would continually wander in and out of the water causing the substrate to become wet. This promotes the growth of bacteria in the tank and considerably increases the humidity. The tanks remain clean and dry for a long period if, instead of including water in the set-up, you soak the tortoises in a shallow pan of lukewarm water for a few minutes, two or three times a week. This seems to work well for tortoises which come from relatively dry climates such as California desert tortoises (*Gopherus agassizi*), Texas tortoises (*Gopherus gerlandieri*), or Hermans tortoises (*Testudo hermanni*). In the wild these animals would not necessarily have water available to them at all times. Another benefit from soaking the tortoises individually is that sometimes the young male tortoises will extrude their penises, indicating the sex of an animal before there are any other visible clues.

DIET

Some of the items which may be beneficial when incorporated into the diet in moderate amounts include mung bean sprouts, tofu (bean curd), French style green beans and other varieties of fresh or frozen mixed vegetables, Gerber's high protein baby cereal, which can be mixed with the vegetables, and dry dog or cat food which has been soaked until it is soft enough for the tortoises to eat. Thrive cat food seems to be especially appealing to the hatchings because it is so brightly colored. I soak the dry chow in water to which I sometimes add children's liquid vitamins and powdered calcium lactate. Wayne's dry dog food, which I moisten and feed occasionally, is especially low in fat compared to other brands (only 8 percent).

In reviewing my dietary regime for this group of hatchlings and other tortoises which I have subsequently hatched and raised, I feel that I was so anxious to ensure the survival of this original group that I went quite overboard with both the quality and quantity of the diet provided. This diet included large amounts of various high calorie, high protein foods which are certainly not what these animals would find in the wild. As a result of the overly rich diet, the hatchlings grew very rapidly but their shells became somewhat bumpy. This is not what is generally observed on wild tortoises which tend to grow more slowly on the limited food supply that is available to them. An additional factor which encouraged me to overfeed these animals is that they quickly learned that I was their food source, and every time I walked by their terrariums they would frantically begin to climb the plexiglass, silently begging for food. You wonder if you have fed them enough and, fearing that you may not have provided sufficient food that day, you find yourself conned into feeding them again. Other tortoises which I have raised on what I would call the "benign neglect" method (less frequent feedings of lower quality foods) have shells which are relatively smooth and natural looking.

However unaesthetic the bumpy or pyramiding effect on the shell carapice may look, this group of animals seems active and healthy. The 38 hatchling tortoises are now five years old. The animals only receive a moderate amount of supplementary feeding since they now have access to a 3,000 square foot outdoor enclosure heavily planted to provide plentiful grazing. I feel that the bumpy shell growth is definitely leveling out.

One of the problems involved in feeding large numbers of animals is that, without careful observation, it is difficult to be sure that all of the animals are actually eating. When the animals are young, I try to observe each animal actually eating, at least two or three times a week. I place a few animals around a mound of food and watch to see if they are all eating. Any tortoise not observed eating has its identification letter noted, and is closely monitored for the next several feedings. One of the first signs that a tortoise is having a problem is its failure to eat. If you just put a mound of food in the terrarium and don't observe the animals actually eating, you may not be aware of a feeding problem until one of the animals is in quite poor condition. It is also helpful to weigh your animals, perhaps as often as once a month when they are young, and keep a record of their weights. Older animals may be weighed less often. Tortoises losing weight or not gaining well should be watched closely.

It appeared to me that during these controlled feeding situations, those tortoises which turned out to be males tended to spend less time eating and were usually the first to leave the feeding area.

GROWTH

By the time this group of hatchlings was a year old, several of them weighed over two pounds and were over six inches long. By the end of the second year, most of the animals were in the five to six pound weight range and were over seven inches long. By the end of the third year, many of the animals weighed over 13 pounds. By the end of five years, some of them weighed over 20 pounds.

Before the animals were two years old, I could see that the tails of the tortoises which would later be sexed as males were becoming considerably longer and larger than the tails of the tortoises which were going to be determined females. A slight concavity began to appear in the plastron of the males. Thirty-one of the hatchlings (82 percent) were evidently females and seven of them (18 percent) were males. Those hatchlings that turned out to be males were clustered at the end of clutch three and included all of the tortoises in clutch four. Perhaps incubation temperature at a critical period was a factor in this clumped distribution of sexes.

MATING

Mating was observed in this group of hatchlings by the time the animals were three years old. Mating usually seems to occur at times when the air temperature is over 85^{O}F. There is no special courtship behavior. Usually the male approaches the female from the rear and mounts her. Males can be heard to make some grunting or gasping noises during the mating.

I observed the males spending more time moving actively around in the enclosure than the females on most occasions. The females moved around most actively in the enclosure during the period prior to laying eggs.

REPRODUCTION

During the summer and fall of 1983, before the oldest of the tortoises was four years old, several of the females began laying eggs. Many of the females laid four or five clutches of eggs, about one month apart, between July and December. Clutch size varied from 5 to 15 eggs. Most eggs were laid in the late afternoon or early evening. By January of 1984, 450 eggs had been laid. By December of 1983 some of the eggs laid in July began to hatch. Only five percent of the eggs that were laid that season actually hatched.

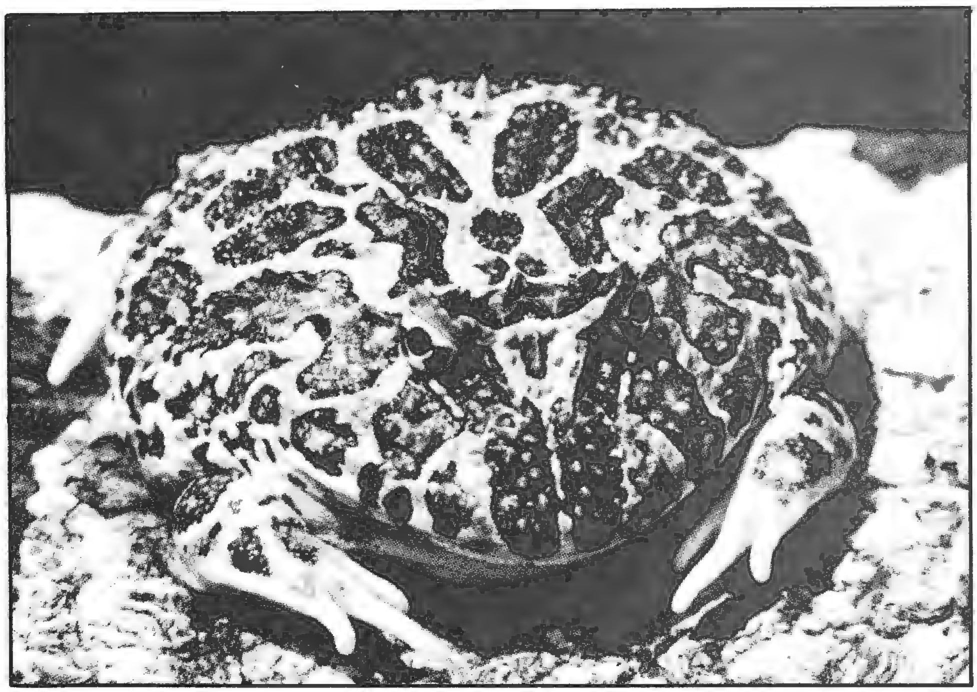
During the egg laying season of 1984, between the months of July and December, the original group of hatchlings laid over 900 eggs. Most of the females laid several clutches of eggs about a month apart, with clutch size varying from six to 20 eggs. By December of 1984 some of the eggs laid in July had begun to hatch.

EGG LAYING

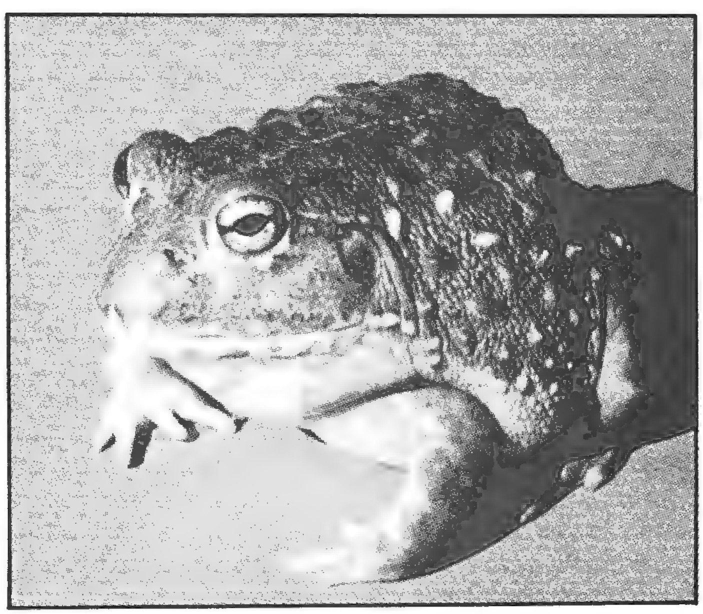
Prior to egg laying, the female tortoises become more active than usual, roaming around the enclosure searching for the perfect spot to begin digging a hole in which to deposit their eggs. (At this time, the tortoises are kept in a 8' by 14' heated house which opens into a 3,000 square foot enclosure.) They may be observed to drink more water than usual, and they feel quite dense and heavy when you pick them up, due to the weight of the eggs and the extra water they have consumed. When they find a suitable spot, they begin laboriously digging with their hind legs, alternately scooping out small amount of dirt with first one leg and the other, and urinating in the hole to soften the soil and facilitate the digging. If they are digging in a spot where the earth is quite compacted, I assist their efforts by pouring some water into the hole and by loosening the soil. If the tortoises are still digging as the temperature begins to fall in the early evening, I cover them with a heating pad to keep them warm enough to complete the digging and egg laying. When they have excavated a hole from eight to 12 inches deep, they stop digging and begin to lay their eggs. The eggs appear from a few seconds to several minutes apart. They slide down a column of viscous mucus, making a slow descent into the hole. This reduces the possibility of egg breakage. I remove the eggs from the hole as they are laid to minimize possible breakage. Upon completion of the egg laying, the tortoise begins to slowly cover the hole, alternately pushing the dirt back in with first one rear leg and then the other. The whole process of digging, laying, and covering may take several hours. When the tortoise is done covering the hole, it is so thoroughly camouflaged that you would never find the eggs, if you had not observed the digging. I check the enclosure at least every hour during the egg laying season (July to December) to make sure that I do not miss any of the animals that are laying eggs.

3701 Hermosa Place, Fullerton, CA 92635

Barking tree frog
(Hyla gratiosa)
(Unknown Mexican subspecies).
Photo by D. McCampbell



Argentine horned frog
(Ceratophrys ornata).
Photo by D. McCampbell



African burrowing frog
(Rana (=Pyxicephalus adspersa).
Photo by D. McCampbell.

ECOLOGICAL BALANCE IN ARTIFICIAL SYSTEMS

Niall F. McCarten

INTRODUCTION

The design of cages for amphibians and reptiles is continuously evolving as we better understand the requirements for keeping and breeding them. The size, shape, and accoutrements must always be for the benefit of the species we are interested in, and not for utilitarian or aesthetic reasons. The skittish behavior of many animals is most often observed when we feed or clean the cages of these animals. Their mad dash for safe cover is undoubtedly a natural response to the perceived presence of a potential predator. In any event, human interference results in a decreased amount of time an animal spends in its normal behavior of resting, feeding, thermoregulating, socializing and breeding.

My approach to terrarium design, therefore, has been to decrease the amount of time in which I might interfere with the amphibians in my care. It is first necessary to analyze the activities which disrupt animal behavior patterns. The various frog species which I keep seem to panic easily. The activities that result in terrarium chaos are: feeding, removing excrement and dead crickets, changing water dishes, direct handling for sexing and checking for maladies, photographing and general observation and turning on and off of lights. Obviously, feeding and some physical handling are necessary. However, lights can and should be put on automatic timers. Photography and observation from a distance will result in limited disturbance if one exercises a little organization and patience.

The chore of cleaning the terrarium is a job which is definitely disturbing to most animals. Frogs, in particular, present a problem with respect to the frequency and amount of time involved in cleaning their terrarium. Owing to their fast metabolism, the frog terrarium often requires cleaning within

36 hours after feeding. This is especially true when they are on a diet of crickets. Therefore, I use a biological filtration method similar to one developed for use with fresh-water fish and invertebrates.

BIOLOGICAL FILTRATION

Biological filtration utilizes bacteria which live on surfaces such as sand, gravel, and filter materials and which biochemically convert waste products into useable fertilizers for plants, etc. The nitrogen cycle (Fig. 1) is a bacterially mediated process which converts potentially toxic ammonia, derived from animal waste products, into nitrates which can be used by plants and algae. Therefore, if waste produced by our animals can be circulated into the nitrogen cycle we may eliminate the arduous chore of terrarium cleaning.

In the semi-aquatic environment ammonia may not be the main concern in terms of animal health hazard. The build-up of solid waste can stimulate growth of toxic or infectious bacteria, molds or fungi. Therefore, the breakdown of excrement and uneaten food to the point where it can enter the nitrogen cycle is a very important step. In order to break down and diffuse the waste into the aquatic portion of the terrarium where it can circulate and be filtered, I use an artificial rain system. The rain washes all of the terrarium surfaces and physically breaks up the animal feces. Once the waste particles are in the aquatic system they can be circulated through the biological filter of gravel and sand.

MATERIALS AND METHODS

Presently, I maintain nine marbled tree frogs (*Phrynohyas venulosus*) from Veracruz, Mexico in a 55 gallon show aquarium. The frogs are fed six to seven small crickets per frog per week in two or three feeding periods. The crickets are powdered with Blair's Super Preen vitamins for added nutri-ents. Occasionally, mealworms are offered in a petri dish.

The terrarium (Fig. 2) contains a submersible water pump which feeds the artificial rain system and circulates the water from the pond area through the gravel. The rain is

THE NITROGEN CYCLE

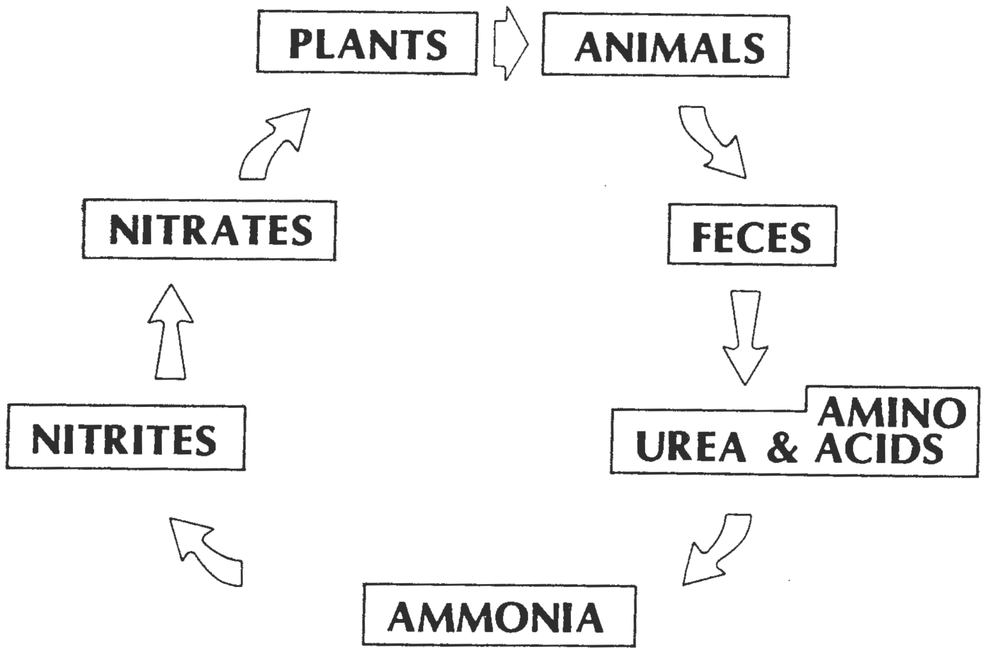


Figure 1. The Nitrogen Cycle, a bacteria mediated system which breaks down organic waste products into chemicals (i.e. nitrates) which can be utilized by plants.

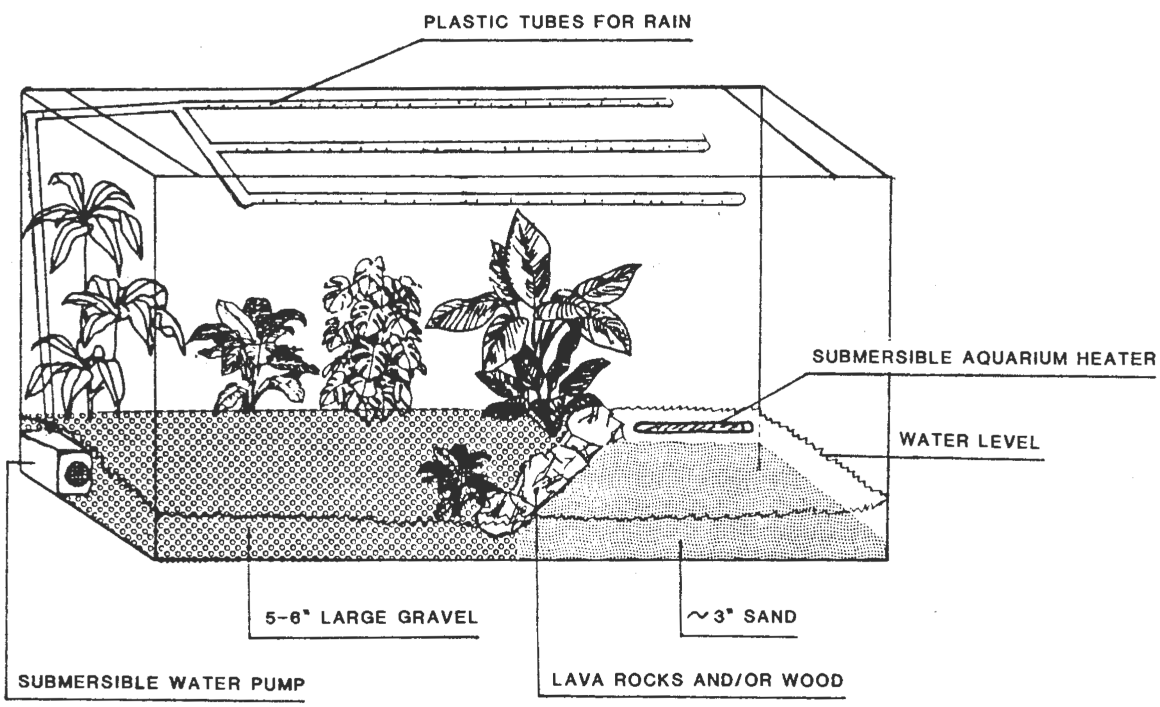


Figure 2. Diagram of a self-contained terrarium system.

produced by several three foot lengths of perforated rigid plastic tubing. The perforations are at various angles and of various sizes so that all parts of the aquarium are washed. River gravel of 3/4" to 1" diameter surrounds the pump and extends over 60 percent of the aquarium bottom with a vertical height of six inches. Washed building sand was used for the pond area and has a vertical height of three inches. Volcanic rocks and waterlogged wood line the pond in order to maintain the different levels.

The water level covers the submersible pump completely, yet about ½" of the gravel is exposed above water level. The sand is covered by nearly 3" of water which forms the pond area. A fully submersible aquarium heater is attached horizontally to the rear wall of the aquarium approximately ½" above the sand.

The artificial rain system is run continuously for up to two days after a feeding. The gravel, plants and wood dry off quickly once the rain is turned off. The water is evenly circulated especially through the gravel and over the sand where the biological filtration takes place. The feces are quickly fragmented within a couple of hours after starting the rain system. Phrynohyas venulosus, as with many other amphibians, is believed to be stimulated into mating behavior by rainfall. If this is the case then the artificial rain may be providing yet another important function.

In the aquatic area of the terrarium I have also added pond snails (Physa fontinalis). Although these snails are the bane of the aquarist trying to grow aquatic plants, they serve a significant purpose in the terrarium system. The snails help reduce frog excrement and dead crickets into pieces which are readily processed by bacteria and other organisms.

Finally, plants utilize the end products of decomposition, i.e., the nitrates. The plants used must be consistent with the enviroment in the terrarium. Since Phrynohyas venulosus is a lowland tropical frog, the plants must be able to survive the temperature (26-30°C), humidity (over 80 percent) and abuse by the jumping frogs. On the gravel, or terrestrial region, I use cuttings of small-leaved philodendrons (Philodendron oxycardium and P. rubra). They are wrapped around pieces of wood leaning up against the rear wall. The cuttings readily send out long trailing roots which attach to the wood. There is no soil or fertilizer used in the system! Aquatic plants which can grow immersed such as Amazon sword plants (Echinodoris spp.) and Anubias spp. are rooted in the sandy pond area. As in freshwater aquaria using biological filtration it is important to use a lot of plants in order to take up the nitrates.

Although this terrarium system requires more work in design and setting up, it definitely reduces the need to disturb the animals later on. Based upon their increased activities, rates of growth, and general health it appears that this system is working well for the frogs. Measurements of pH, ammonia, nitrite and nitrate levels further support my contention that the nitrogen cycle and overall biological filtration process is working. The terrarium has been set up for nearly six months without having to clean it by hand or change the water. Fresh water is added to maintain the desired level.

Further advantages to this design include the capability of keeping more species due to decreased maintenance time as well as the important potential that breeding under semi-natural conditions will occur.

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AN OVERVIEW OF THE AMPHIBIAN COLLECTION AT THE STEINHART AQUARIUM AS AN INTRODUCTION TO AMPHIBIAN CARE

Bruce I. Hiler

INTRODUCTION

With a display collection of over sixty species, Steinhart Aquarium maintains one of the largest amphibian collections in the West. It is the purpose of this paper to correlate this collection in terms of scope and direction, as well as utilize the collection's diversity to introduce a systematic approach to amphibian culture in captivity.

The Steinhart Aquarium opened in 1924, and has been one of the world's best aquariums ever since. It features a huge collection of both freshwater and saltwater fish, displayed in a long gallery of large tanks around a stylized gothic alligator "swamp". Amphibians were not on the site until the 1963-1964 renovation at which time a battery of small vivariums was constructed along the south edge of the Swamp. The amphibian gallery was equipped with two of the Aquarium's freshwater systems and has remained essentially unchanged since 1964. The utilization of the Aquarium's heated and chilled freshwater systems not only satisfies two of the systematic requirements for amphibian care, it also helps to make the Steinhart Aquarium ideally suited to house such a large colletion. There are eight topical requirements for amphibian capture culture, which are:

- An understanding of the species' natural history.
- The temporal function.
- The enclosure (spacial function)
- Substrate/decor
- Moisture
- Temperature
- Lighting
- Food

The first and second topics are precursors for the other six. The proper understanding and application of all eight topics will guarantee success with any animal species, whether vertebrate or invertebrate.

THE SPECIES' NATURAL HISTORY

There can be no long-term success with a captive amphibian without a good understanding of the animal's natural history and how its native environment and ecology applies to captivity. To gain information on a species' natural history, several things can be done such as field research, library research, and personal discussion with people who have studied or maintained the beast or close relatives of it. How this information is applied to captive stock will determine the degree of keeper expertise.

A common trap, which is easy to fall into, is to overgeneralize the requirements of a particular species. For example, the climbing toad (Pedostibes hosei) and the hill toad (Bufo asper) both originate from south-east Asia. It would be easy to assume that both toads could be housed together and be given the same food, temperature, surroundings, etc. However, a close look at each of these species' natural histories shows several opposing needs. The hill toad is a cool-loving, high elevation animal, feeding mainly on soft-bodied invertebrates. The climbing toad is arboreal, from warm low-land rainforests, and feeds mainly on flying insects. Under certain circumstances, generalization of an animal's natural history can be helpful when unexpected animals are obtained or when there is very little information available on the ecology of unusual species. It is important to remember, however, that speciation is a natural process whereby species acquire unique characteristics as a reflection of the animal's environment. Therefore, while the keeper's body of knowledge of these characteristics increases and is applied to its husbandry, the animal's captive state should be in a dynamic state.

THE TEMPORAL FUNCTION

Amphibians as a family are most noted for their dual life cycle, a fully aquatic larval stage followed by an adult terrestrial stage. They are also most active at dawn or dusk and at night. These two biological functions along with their seasonal reproductive cycle, act as a temporal function. As a requirement its use is straightforward. For example, most tadpoles need different care than their parents and anyone raising an amphibian through its full life cycle will need to satisfy both sets of requirements. The same need for two sets of requirements will be necessary when considering amphibians that need to go through a hibernation period or a rainy season.

ENCLOSURE

The type and size of an amphibian enclosure is the most important consideration of the five captive environment topics. The enclosure is equal to a spacial function and is the limiting factor of species maintenance, determining the number of animals, their health, and their breeding success. Ideally the enclosure should be large enough to allow its occupants to carry out all their biological functions throughout their life. In practice, the enclosure is usually a fish tank or a variation of one, and the animals are moved or the tank is altered depending on the inhabitants' temporal function requirements.

The amphibian gallery at the Steinhart Aquarium has fiberglass boxes with a front viewing window. The enclosures are of five different sizes and range from 1.25 cubic feet to 10 cubic feet. There are 29 enclosures in all, 24 of them are 3 cubic feet or smaller. The smaller vivariums are inadequate for long-term culture of most amphibians. For this reason the collection's trend is towards smaller amphibians that adapt well to the available space, such as poison arrow frogs (Dendrobates spp.), fire-bellied toads (Bombina spp.) and newts of the family Salamandridae.

SUBSTRATE/DECOR

The surroundings within the enclosure allow the animal to behave "naturally". This includes fine sand or soil to burrow in, rocks and wood to hide under, branches and plants to climb on, ponds or places to breed. The important consideration is to include the proper substrate and all the "props" to allow the animal to carry out all of its natural behaviors.

Since the Steinhart Aquarium's enclosures are also public displays, they typically are "natural" looking habitats that are well planted. Hard substrates, rocks and gravel, are usually avoided since they can cause abrasions and digestive blockage. Instead, fine sand, soil, wood, leaf litter, sphagnum moss, and living substrates of Java moss _Vesiculuria spp._ and fontinalis (_Fontinalis spp._) are used. These are better for the animals as well as creating for the viewer "mind images" of the amphibians' native home. A beautiful, well-planted, well-balanced amphibian vivarium is the nicest bonus about keeping amphibians.

MOISTURE

No other land vertebrate is more tied to water than amphibians. This is no surprise to anyone, but can be a great advantage to the amphibian keeper. A few groups of amphibians, notably the south-east Asian leaf frogs (_Megophrys spp._), the African rain frogs (_Breviceps spp._), the African grey tree frogs (_Chiromantis spp._), and various other desert dwellers like the spadefoots (_Scaphiopus spp._) like dry conditions. Most other amphibians flourish in damp enclosures. Water being the universal solvent, coupled with its thermal properties makes it ideal for use as a cleaning agent and as a means of regulating temperature, as well as its basic function of preventing desiccation. In order to take full advantage of this groups' love of water, it is important to design the enclosure properly and have plenty of clean, fresh, aged water available.

The vivariums at the Aquarium are of two basic designs. The first type is an open drip system in which water continuously and slowly enters the enclosure and drains through a drain line drilled at the appropriate height on the back wall. The other design is more applicable for home amphibian keepers. The vivarium has a two inch thick substrate of #20 aquarium gravel covered over by one of the softer substrates such as Java moss. A four square inch corner is left bare down to the base of the tank. This can be done by making a retaining wall with rocks, wood, or pieces of brick. The enclosure is then periodically, usually twice weekly, flooded well above the soft substrate level. By using the bare corner as a sump, the water is siphoned off. The process is then repeated, but then siphoned to the level the animals need until the next cleaning day.

TEMPERATURE

Amphibians generally have to be cooler than their reptile cousins from the same area. This is one reason tropical amphibians are found in trees or under logs and desert amphibians are found underground. Tropical and desert amphibians enjoy temperatures ranging from 68°F - 79°F (20° - 26°C), and temperte and cloud forest amphibians will do well between 64° - 72°F (18° - 22°C). Additionally, many need to be kept cooler or even hibernated below 41°F (5°C) in order to complete their life cycle. As stated above, each animal deserves unique consideration, and generalized temperatures should be used only on a temporary basis.

There are many ways of maintaining the temperature in an amphibian vivarium. Most of the methods used for captive reptiles will work, such as incandescent lighting, heat tape, and small room heaters. An easy and reliable method is to use a fish tank heater. This can be submerged in the "pond" part of your vivarium as long as the water is not stagnant. If it is, the heater may not work properly and get dangerously hot. You can circulate the water with a small air pump and airstone or sponge filter that will keep the water clean. If the vivarium is mostly terrestrial, the heater can be housed in another tank or commercially available canister and the water pumped through the vivarium. Several enclosures can be heated by one canister, and temperature regulated individually by flow rate. Using low wattage incandescent bulbs in conjunction with an aquarium heater will simulate the extra heat produced by sunlight as well as form temperature gradients inside the vivarium. Cooler-loving amphibians will often do well in the cooler spots in a home, such as the garage or basement. Some amphibians such as Pacific giant salamanders (Dicamptodon ensatus) or Sierra ensatina (Ensatina eschscholtzii platensis) will do well only if kept in expensive refrigerated systems. Animals can be hibernated in plastic shoe boxes in a refrigerator. Make sure the temperature never drops below 38^{o}C (4^{o}C) and check the animal's weight and the box's moisture every few days.

LIGHTING

Light duration is more important than light intensity since most amphibians enjoy dimly lit surroundings. The duration of the light or day length is often a visual key to the temporal function of an amphibian's reproductive cycle. Paralleling an amphibian's native day length can be done with an inexpensive light timer adjusted every wekk to simulate the changes in sunrise and sunset. If plants are being kept within the vivarium, full-length fluorescent lights will provide uniform but not harsh lighting, making the animals and plants easy to observe and pleasant to view.

FOOD

Very little research has been done on the dietary habits of amphibians, therefore most amphibian diets are modified reptile and fish diets. The amphibians at Steinhart Aquarium are fed a great variety of food, most of which is cultured at the Aquarium. Along with a varied diet the food is fortified with a vitamin-mineral powder such as Vionate or a liquid such as Avitron. The types of food used can be placed into four categories:

1. Vegetable (for vegetarian tadpoles): green water, romaine lettuce, green algae, and fish foods such as Tetramin tablets and trout chow.
2. Soft-bodied invertebrates: white worms, black tubifex worms, earth worms and slugs.
3. Hard-bodied invertebrates: wingless fruit flies,

mealworms, blow flies, and crickets of all sizes.

4. Vertebrates: filets of fish, horse meat, liver, goldfish, mice.

The category of food an amphibian prefers can usually be determined by looking up the gut contents in field reports of wild-caught specimens. Many amphibians will do better on soft-bodied invertebrates with a coating of liquid vitamin supplement versus the traditional cricket diet. It is often possible to "trick" an amphibian into eating a food type it would not naturally find in the wild. Toads (Bufo spp.) leaf frogs (Megophrys spp.), horned frogs (Ceratophyrs spp.), and African bullfrogs (Pyxcephalus adspersa) will often take filets of fish dangled in front of them by forceps. Fire-bellied toads (Bombina spp.), and poison arrow frogs (Dendrobaties spp.) will take worms out of an inverted jar lid. So little is known about amphibian diets that good health can only be achieved by using supplements and by offering the animals the widest selection of food possible.

SUMMARY

Eight topics have been briefly discussed in order to gain a general understanding of each. Specific rules on amphibian care have not been stated due to the scope of the paper as well as the diversity of amphibians. The intent herein is to understand the topics and utilize them for each species being cared for. This can easily be done in a notebook type format (see below). Since amphibians are harder to keep than most other land vertebrates, this topical systematic approach will help trouble-shoot problems. It should also help beginners and experts alike think clearly about the animals' captive needs. The notebook method can also be used as a means of communicating technique and difficulties encountered, via sharing their contents with peers.

The amphibian collection at the Steinhart Aquarium will remain large and diverse. Hopefully, this will continue to make the aquarium a good source of information on husbandry for a wide variety of amphibians.

An Example of the Notebook Format

ORIENTAL FIRE-BELLIED TOAD (Bombina orientalis)

Purchased: 10/12/83, 10 animals, 5 ♂ , 5 ♀ .

Natural History: Subtropical marshes and swamps of south-east Asia, fairly aquatic, feeds on insects and worms, tadpoles vegetarians, will not tolerate low temperatures, breeds spring through summer, have larger forearms than .

Enclosure: 36"l x 18"h x 18"w fiberglass box with viewing window.

Substrate/Decor: fiberglass rockwork with #20 gravel in pond, Chinese evergreen, fontinalis, dwarf sedge.

Moisture: On drip system (hot fresh), 3" deep pond covering 1/3 of vivarium.

Temperature: Air 68° - 77°F (20° - 25°C), water 68° - 74°F (20° - 23°C).

Lighting: Two 3-foot Vitalite fluorescent bulbs, 10 hours a day year-round, indirect sunlight.

Food: 3-week old crickets, fish filets, 3 times a week.

Observations: Active and hardy, very good at escaping from enclosure. Spend 75% of their time in the water. Laid eggs 3/15/84, 3/22/84, 3/30/84, 4/10/84, 4/26/84, 25 to 50 eggs per clutch, hatch in 3 days. Feed on Tetramin tablets. Tadpoles removed from vivarium at 10 - 14 days old. Metamorphosed at 4 to 5 weeks. Froglets small and delicate, grew rapidly on wing-less fruit flies and white worms, and soon graduated to 2 and 3 week old crickets and tubifex worms. Reached adult size in 10 to 12 months.

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THE EFFECTS OF GAS SUPERSATURATION ON AMPHIBIANS

Kris Orwicz

INTRODUCTION

Water quality has a major effect on the well-being of aquatic animals. A serious disease of amphibians in both laboratory and natural conditions is "Red leg" which causes mortality due to high systemic levels of the bacteria Aeromonas hydrophila (Gibbs et al., 1966; Hird et al., 1981).

Exposure of amphibians to dissolved gas supersaturation may produce clinical signs similar to those of "red leg" disease (Colt et al., 1984a). Gas supersaturation may be present in both natural and laboratory systems. This paper will focus primarily on gas supersaturation in laboratory systems.

WHAT IS GAS SUPERSATURATION?

The composition of dry air is 78.084 percent nitrogen, 20.946 percent oxygen, 0.934 percent argon and 0.032 percent carbon dioxide. The solubility of each of these gases and water is a function of barometric pressure and temperature. The pressure a given gas exerts in the atmosphere in the absence of all other gases is its partial pressure. Total gas pressure consists of the sum of the partial pressures in the liquid plus the vapor pressure of water:

$$\text{Total Gas Pressure} = P^{\ell}_{O_2} + P^{\ell}_{N_2} + P^{\ell}_{Ar} + P^{\ell}_{CO_2} + P_{H_2O}$$

At equilibrium, total gas pressure is equal to the local barometric pressure. Three conditions may exist:

Total Gas Pressure = Barometric Pressure (equilibrium)

Total Gas Pressure > Barometric Pressure (supersaturation)

Total Gas Pressure < Barometric Pressure (subsaturation)

The difference between the Total Gas Pressure (TGP) and the local barometric pressure (BP) is the differential pressure

(ΔP). Total gas pressure is equal, therefore, to BP + ΔP.

ΔP can be measured directly, thus this parameter is used to express gas supersaturation levels with;

$\Delta P = 0$ (equilibrium)

$\Delta P > 0$ (supersaturation)

$\Delta P < 0$ (subsaturation)

The traditional method of reporting gas supersaturation (Colt, 1983) is the expression of the total gas pressure as a percentage of local barometric pressure:

$$TGP(\%) = \frac{[BP + \Delta P]}{BP} \times 100$$

A major effect of gas supersaturation on aquatic organisms is formation of emboli. When the $\Delta P > 0$, emboli may form within the organism causing Gas Bubble Disease (GBD). If the $\Delta P \leqslant 0$, gas emboli can not form regardless of the partial pressure of a single gas.

OCCURANCE OF GAS SUPERSATURATION

Gas supersaturation can be produced by a variety of processes. The major mechanisms are heating, bacterial action, air entrainment and photosynthesis. The occurance of gas supersaturation in the hatchery may be a result of a supersaturated water source or due to production of supersaturated conditions within the hatchery system.

Natural supersaturation commonly occurs in groundwater, springs, streams, lakes, and bays at some time of the year (Bouck, 1976; Harvey, 1967; Lindroth, 1957). The usual mechanism in these situations is heating of the water, since the solubility of gases decreases with increasing temperature. In large bodies of water such as lakes and reservoirs, heating occurs to a significant depth. Spring or well water is commonly supersaturated due to the water temperature being higher at discharge than at the time the water went underground.

This mechanism may be coupled with bacterial action, which may modify dissolved gas levels in natural water sources. Bacterial respiration results in decreased oxygen and increased carbon dioxide concentrations (Matsue et al., 1953). Under anaerobic conditions, significant amounts of methane and nitrogen may be produced resulting in supersaturation (Koyama, 1964).

Incoming hatchery water may be supersaturated at some time during the year, therefore, the water should be degassed before use. The use of degassed water (i.e., water at equilibrium) does not assure that the system will be free of supersaturation problems. Gas supersaturation may be produced inside the culture system due to heating without re-equilibrium of dissolved gases, air entrainment, and green water culture.

It is common practice to heat water to achieve optimum hatching, survival, or growth. The mixing of water of different temperatures may cause gas supersaturation since the solubility of gases decreases with increasing temperature. A problem with closed heating systems (common water heaters) is that elevated hydrostatic pressure will prevent gas transfer until the water is discharged into the culture system.

A common mechanism for production of gas supersaturation is air entrainment. If the screens of a hatchery intake pipe clog, the pipeline may not be full, thus entraining air (Wyatt and Beiningen, 1971). Air leaks around pump seals, loose valve connections, pinholes in tubing or pump intake pipes not being fully submerged can cause subatmospheric pressures that result in air being drawn into the water system. Various aeration methods such as airlift pumps or submerged aeration may cause supersaturated water (Colt and Westers, 1982).

The raising of algae (green water culture) for feeding some young amphibian species may cause water to be supersaturated with dissolved oxygen (Supplee and Lightner, 1976). During the day, algae produces dissolved oxygen levels that may range from 200 to 400 percent of saturation.

MEASUREMENT OF GAS SUPERSATURATION

There are many methods and instruments available for the measurement of gas supersaturation. The most commonly used instrument is the "Weiss saturometer." This instrument is simple to operate and measures ΔP directly. The instrument consists of 200 ft of silicone rubber tubing connected to a pressure gauge. The silicone rubber is permeable to dissolved gases. At equilibrium the pressure inside the tubing is equal to ΔP in the water (Fickeisen et al., 1975).

Most other products available use this basic semi-permeable membrane concept. They vary in size and in their capabilities of measuring other water quality parameters. Examples include hand held battery operated tensiometers to multiple channel electronic systems which use a solid state electronic pressure transducer.

EFFECTS OF GAS SUPERSATURATION ON AMPHIBIANS

Aquatic organisms respond differently to gas supersaturation, but the process is fundamentally the same (Weitkamp and Katz, 1980). When the ΔP inside an aquatic animal is greater than zero (i.e., supersaturated) there is a tendency for bubbles to form. Bubbles that form in the blood are called emboli while bubbles formed in the tissues are called emphysema. The quantity, size and location of emboli and emphysema is related to the degree of supersaturation, exposure time, and tissue type.

Clinical signs of gas bubble disease vary in aquatic animals but amphibians follow the basic pattern seen in fish and invertebrates. Extremely high levels of gas supersaturation (ΔP = 200-250mmHg) can cause a high percentage of mortality within a short period of time (1 - 3 days). This is due to large emboli occluding critical portions of the cirlatory system (Beyer et al., 1976; Colt et al., 1984a). In the bullfrog (Rana catesbeiana) external clinical signs are rarely observed, but internally bubbles may be present under the skin and in the vascular system, along with a large degree of internal hemorrhaging. Also, in the Eastern newt (Notophthalmus viridescens) bubbles in the eye or a swollen abdomen have been occasionally observed.

As the ΔP is decreased (115-185mmHg), internal effects are less severe, however external effects become more prominent and death may result from a secondary bacteremia. Some clinical signs are quite obvious yet others are subtle. Abnormal behavior is a nonspecific sign that may be observed. This may include the inability to locate or ingest food, loss of equilibrium, and floating [due to the accumulation of gas under the skin (Figure 1) or in the gastrointestinal tract]. In bullfrog tadpoles (Rana catesbeiana), exposure to gas supersaturation results in the inflation of the gastrointestinal tract. Affected tadpoles floated on the surface, either with their left sides elevated or on their backs. Tadpoles exposed for 10 days to a ΔP of 160-170mmHg showed increased levels of systemic Aeromonas hydrophila bacteria (Colt et al., 1984b). Clinical signs in frogs (Rana catesbiana, Xenopus laevis) begin with the formation of gas bubbles in the interdigital webbing, which can occur within 24 to 48 hours. The bubbles then expand in size and number throughout the webbing (Figure 2). An ascending hyperemia progresses up the legs and abdominal surfaces due to the rupturing of small capillaries (Figure 3). The forelegs follow the pathogenesis of the hind legs. External bubbles disappear, leaving petechial and ecchymotic hemorrhages which enlarge and progress into eroded areas of necrotic skin. At this point, the frogs typically lose their ability to stay submerged. Death may result due to accumulation of gas in the vasculature system or from a secondary bacterial infection (usually Aeromonas hydrophila). The develoment of these clinical signs occurs within 3-4 days for Xenopus held at a ΔP of 180mmHg (Colt et al., 1984a). These signs may be reversed within a few days by reducing the TGP to equilibrium.

The clinical signs of gas bubble disease in fish and invertebrates parallel the signs reported as "red-leg" in amphibians--except for the presence of emboli. Failure to observe the emboli may be due to the small size of the emboli, their ease of rupturing and the rapid development of hemorrhaging that destroys the bubbles.

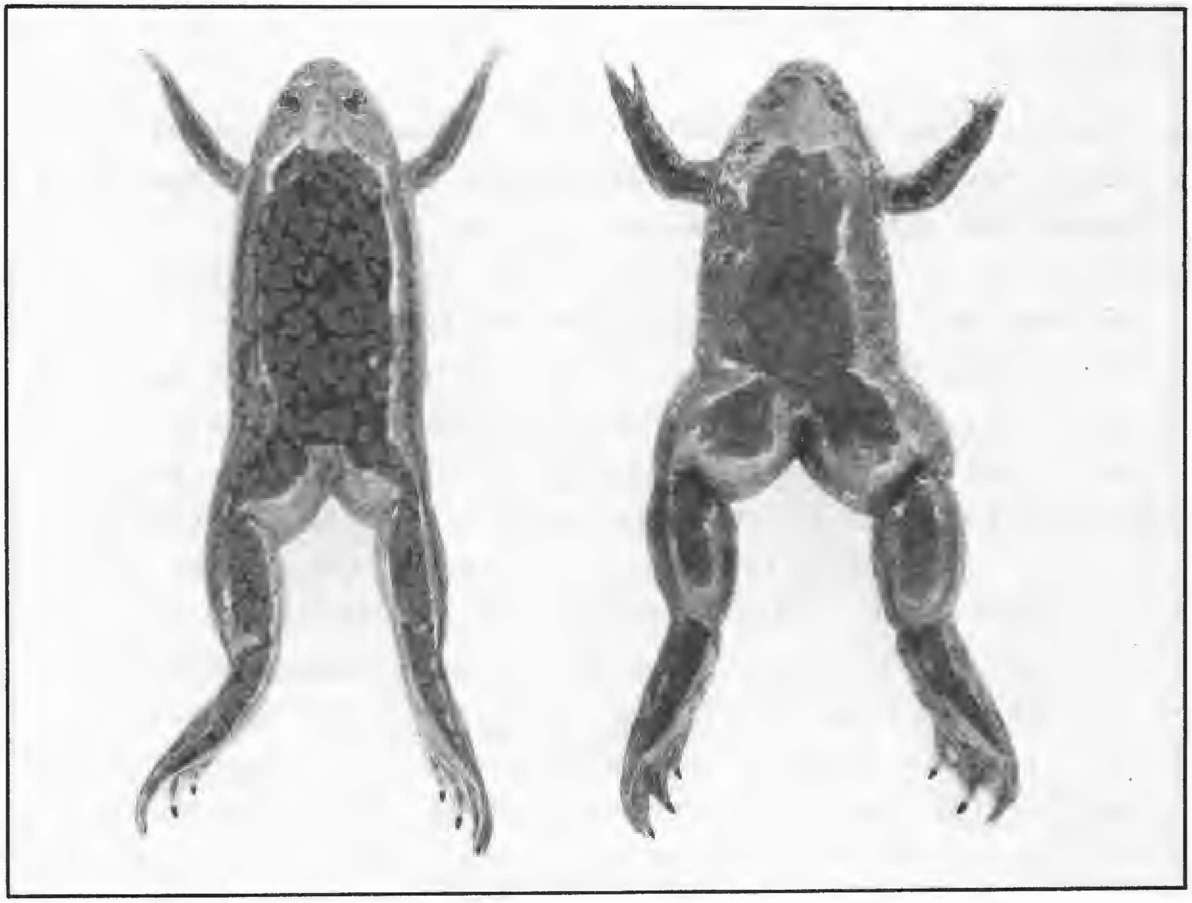

Figure 1. Comparison photograph of <u>Xenopus</u> <u>laevis</u> with accumulation of gas under the skin.

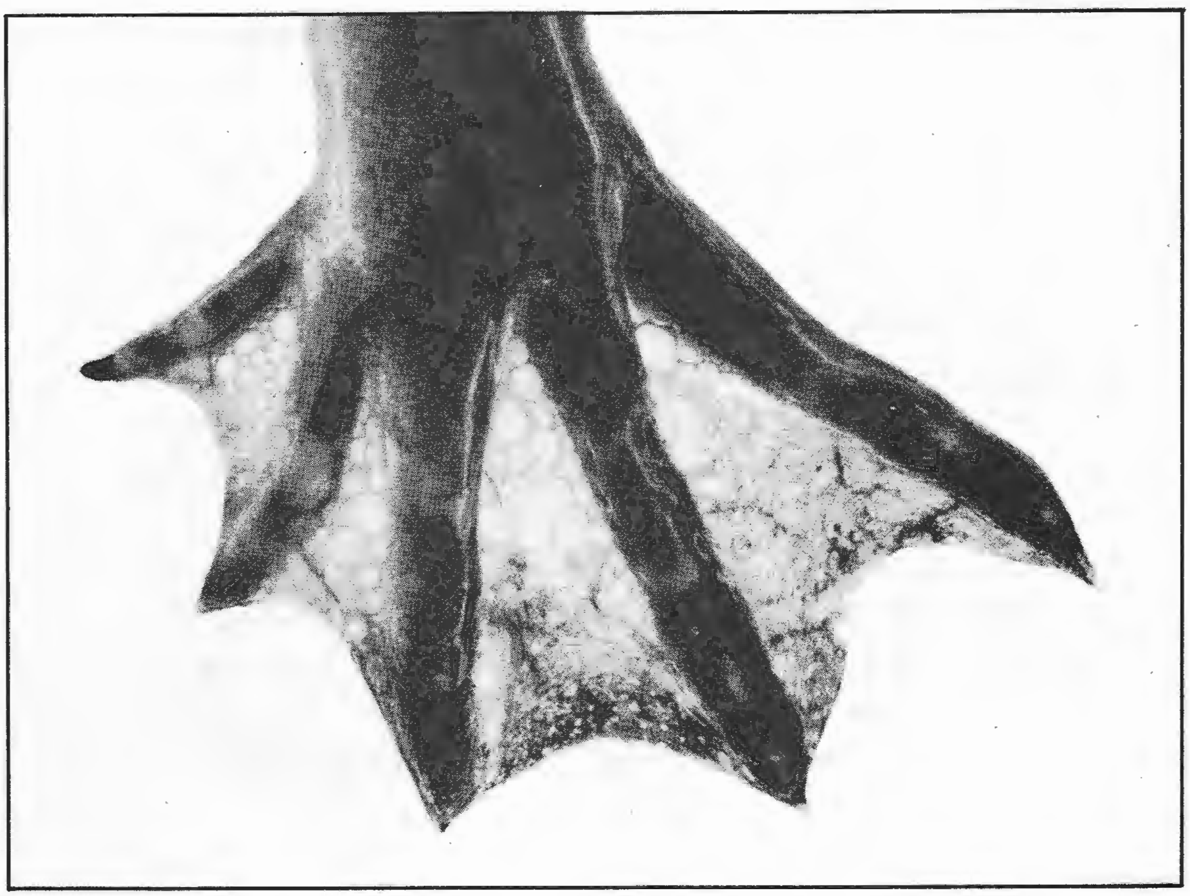

Figure 2. Bubbles in the webbing of <u>Xenopus</u> <u>laevis</u> exposed to dissolved gas supersaturation.

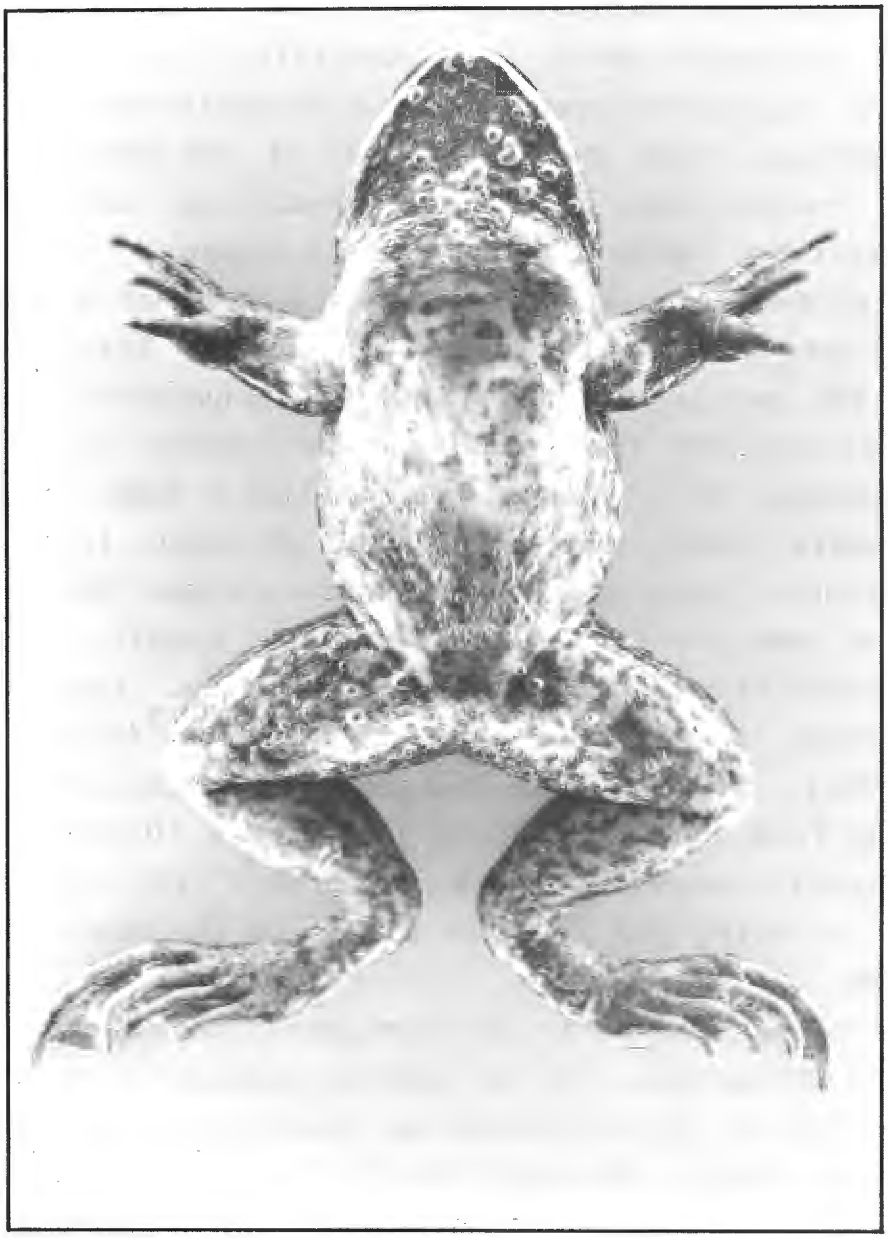

Figure 3. Bubbles on the ventral body surfaces of *Rana catesbeiana* .

Amphibians appear to be able to withstand a higher degree of gas supersaturation than fish. In fish, a ΔP in the range of 120-140mmHg will kill 50 percent of the animals in 4 days. In amphibians no clinical signs are seen below a ΔP of 100 mmHg--when held for 2 weeks or less. However, chronic levels may prove to be a significant stress.

REMOVAL OF GAS SUPERSATURATION

Supersaturated water will naturally lose gases to the atmosphere. In static systems, this mechanism may be adequate for prevention of gas bubble disease if the water is allowed to stand several days before the animals are added. In flow-through systems, natural degassing is generally too slow and the rate of degassing must be increased by other methods.

The most efficient methods or devices for gas removal maximize the gas-liquid interface. Two convenient methods are packed columns and fine nozzles. The packed column aerator (PCA) consists of a column filled with a high surface area packing media. Water flows down over the media in a thin film thus causing a large gas-liquid surface area. The key design parameters are column height, surface loading rate (flow rate/cross-sectional area) and media size. Typical column loading rates are in the range of 50-200 $m^3/m^2 \cdot h$ (Colt and Bouck, 1984). The height of the column depends on the P and may range from 1-3 m. For low flow rates (0.01-0.03 m^3 h), drip irrigation nozzles placed inside 2-3" PVC pipe produce a fine mist allowing for gases to escape to the atmosphere.

CONCLUSION

Gas bubble disease is not recognized as a serious problem in amphibian culture. It is usually assumed that amphibians are not affected by dissolved gas supersaturation since they are air breathers. However, 35-70% of the oxygen uptake of amphibians is through cutaneous respiration, and during hibernation this may reach 100%. This makes amphibians susceptible to GBD.

In all aquatic organisms, high levels of dissolved gas supersaturation can cause rapid mortality. At lower levels it may cause chronic mortality, or be a predisposing factor to secondary bacterial diseases (e.g., "red-leg").

Dissolved gas supersaturation is easily measured and can normally be alleviated using standard degassing devices developed for fish hatcheries. With properly designed facilities, successful propagaion of amphibians may be enhanced.

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PANEL COMMENTS

During the conference a panel was assembled to answer questions from the audience. Several of the panel members prepared written responses to topics they felt were important. The responses are included below.

Question: How can cooperation and communication between professional and amateur herpetologists in matters such as propagation and husbandry be improved?

Jim Murphy: At the Dallas Zoo we have a large collection of diverse herpetofauna. Due to our location and the size of the collection, a considerable number of amateurs interested in herpetology visit our facility. Often, these people are primarily interested in breeding colubrid snakes. It is perplexing to me that often the only snakes that interest these peole comprise the genera *Elaphe* and *Lampropeltis*; all of the other taxa in the collection are basically ignored. The conversation is often composed of the nuances of pattern and color in a particular taxon of kingsnake. Rarely does the conversation deal with the biology of the animals or the interesting diversity of other reptiles and amphibians.

Furthermore, when I have attended some regional herpetological society meetings, discussions have centered on the cost of a particular colubrid snake, the crosses between various subspecies of kingsnakes and recent legislation. The likelihood of improved communication between the groups would be enhanced if the topics discussed were broader in scope and encompassed other elements in herpetology, such as behavior, ecological parameters, zoogeography or taxonomic discussions of other reptiles and amphibians. My plea is simple - let's expand the dialect.

Question: What is the main problem with the state of the art of captive reproduction in reptiles and amphibians?

Ernie Wagner: The main problem with the state of the art of captive reproduction is the lack of definitive parameters of environmental manipulation needed to induce reproduction. For instance, how much light is needed and what type? How long a hibernation period is required and what is the minimum effective temperature? I find myself in the same situation as many of the other peole who occasionally present reproduction papers at conferences. That is, I can tell what worked for me, but when it comes to answering questions later about specific details (i.e., will two months hibernation work instead of three?) I just don't have the answers.

What is needed in the field of reptile and amphibian breeding is for someone to do the detailed empirical studies necessary to define these environmental parameters. I would like to see someone in the research field set up a large group of one species of reptile or amphibian and begin to exactly define what will and won't work in terms of husbandry, diet, and environmental manipulation. I realize this is a major undertaking and that's probably why it has not all ready been attempted, but these sorts of parameters have been worked out for other fields of animal husbandry, and should be within the realm of possibility in ours. Possibly, then, someone could give a better, more definitive answer to questions from people who wonder why they keep getting infertile eggs.

Question: What do you see as the future in reptile and amphibian propagation?

Ron Tremper: There is quickening evolution occurring among captive breeders towards species diversity, sharing important information and establishing profitable "hobbies" or actual businesses based on self-sustaining populations of herps.

As the pool of successful private breeders enlarges throughout the world there will emerge a few individuals who begin "reptile ranching". The use of free-living outdoor pens will be the main focal point of such commercial operations. Lizards and chelonians will be mass produced, as well as a large variety of amphibians. As the importance of outdoor pens is realized by the herp community and the techniques of such pens are perfected, a number of snake species will be experimented with and successfully reared outdoors.

As the ever decreasing ability to import enough animals from the wild becomes apparent a great demand for captive-bred animals will only speed up this process of reptile ranching. Where there is a need, there will be someone to fill that market.

Too much energy and time is wasted by trying to fool "Mother Nature" indoors. Not only is it harder to breed reptiles compared to outdoor efforts, but we are now seeing many problems associated with poor sperm viability and vitamin/mineral deficiencies.

In closing, I can clearly see that there will soon not only be a greater interest in breeding amphibians and reptiles but there will be a big need to do so. Within five years every serious breeder who lives where there is any type of favorable climate will have one or more outdoor breeding pens for year-round care of select species. Also, the growth in numbers and quality of the new information being gleaned will bring the status of our expanding profession to new heights and approval throughout the country. Herpetoculture is here to stay!