Expedition Field Techniques

REPTILES AND AMPHIBIANS

by Daniel Bennett

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1 Kensington Gore
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Tel +44 (0)20 7591 3030   Fax +44 (0) 7591 3031
Email go@rgs.org   Website www.rgs.org/go

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Cover image: Adder and grass snake from "Knights pictorial museum of animated nature and complexion for the Zoological Gardens", circa 1887.

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Daniel Bennett, January 1999

Department of Zoology
University of Aberdeen
Tillydrone Avenue
Aberdeen
AB24 2TZ

About the author

Daniel Bennett was born in Glossop, Derbyshire. He has carried out fieldwork in Turkmenistan, Australia, Malaysia, India, the Philippines, Madagascar and Ghana. He is the author of several books about monitor lizards. As well as reptiles and amphibians his research interests include fruit bats and hippopotamus.

INTRODUCTION

Why study reptiles and amphibians?

Traditionally, reptiles and amphibians are neglected subjects for expedition studies. Very few reptile communities have been documented. Our knowledge of the behavioural ecology of most species of tropical lizards, snakes and amphibians is so slight that a carefully conducted study lasting just a few weeks can make a very significant contribution to the pool of scientific knowledge for most species. Because reptiles and amphibians are neglected groups their conservation is often overlooked. Important sites with rare species or assemblages are destroyed or modified in detrimental ways, simply because nobody knows anything about the animals that live there. Conservation strategies rely on baseline biological data that is all too often missing for these groups. As a result they gain little or nothing from conservation efforts or even suffer from well meaning, but ill informed, decisions. There is currently great concern that a worldwide decline in amphibian populations is occurring, possibly because of pollution or climate change. But because almost all amphibian communities are very poorly known, the true extent of the global decline is not clear.

As well as the urgent need for more information about reptile and amphibian communities there are several practical considerations that make their study particularly attractive for expeditions. Most importantly, much work can be done non-destructively, i.e. with the minimum risk of harm to the animals. Reptiles and amphibians tend not to go into shock with handling and capture. Such a response is relatively common in more delicate species of rodents, birds, bats and insectivores and may represent a major cause of mortality in the sample under study. Secondly, you are most unlikely to catch any diseases from the animals you are working with. Furthermore, most reptiles and amphibians can be identified on sight after a little practise and it is rarely necessary to preserve more than one voucher specimen of each species. Reptiles are much easier to find than other vertebrates; walks through forests or deserts almost always make amphibians or reptile sightings, whilst other vertebrates may appear to be much rarer at any time of day. Finally, and crucially, reptiles and amphibians tend to be much easier to catch than small mammals, birds or fish.

Despite these considerable advantages, very few expeditions make an effort to study reptiles and amphibians. Could this be because of a primeval fear of snakes and all things scaly or slimy? Or is it because techniques used in herpetology are quite different from those used for any other vertebrates? Most universities do not have members of staff with practical expertise in herpetology and most libraries can provide little in the way of theory. This book aims to outline methods that can be used in short studies of reptiles and amphibians. It aims to provide some theory but makes no claim to expertise. By necessity I have written the guide in an authoritative style, but there is “more than one way to skin a crocodile”. Many much more experienced workers than me have different ideas from those outlined here, and it is well worth the trouble finding them both at home and abroad to get their advice. Amphibians are treated separately from reptiles because many methods applicable to the latter would be harmful to the former. By necessity the book is written for students in Britain who are planning work in the tropics. For people planning work in their own countries some of the information given here might seem superfluous. Finally, and again by necessity, the emphasis here has been on lizards, at the expense of other taxa.

The study of reptiles and amphibians has been given the unfortunate title of **herpetology** - literally the study of things that crawl. Worse still, somebody who studies reptiles and amphibians is branded a **herpetologist**. With the exception of snakes, very few reptile or amphibians move by crawling and although these terms have near universal acceptance, I do not like them and have tried to avoid using them wherever possible.

Section One
PREPARATION

1.1 Getting help, students and supervisors

Unless you already have contacts in the country where you wish to study, you will need to make friends there at the earliest possible opportunity. The most important contacts are academic institutions (who may be able to provide students for you tol work with) and government departments (who are likely to supply the necessary permits and can introduce you to forestry or wildlife personnel).

Initially you should contact all the local universities. The *World of Learning, held* in the reference section of most libraries, list universities by country. Write to the Head of Department, indicating your aims and intended dates and ask if they would be interested in letting their students participate. Make it clear from the start that you will be able to cover their field costs. Try to get the names and addresses of the students you will be working with as early as possible so that you can involve them in the preliminary planning. If you don't get responses from institutions a combination of letters, faxes, emails and telephone calls should eventually elicit a reply. It can be very exciting to involve students from rival universities, of different disciplines or of different religions in the project. Many institutions that would not normally consider co-operating with each other may do so if the project has an "international conservation" element.

1.2 Keeping on the right side of the law

Permits are almost always necessary for fieldwork and can take a long time to be issued. It is very important that you apply for them as early as possible and get all host country partners to support your application. You may need to apply to more than one government department - your academic supervisors should advise you on this. The permits needed depend on the sort of work you want to do. If you want to work in a protected area a simple entry permit may be sufficient. If you want to catch animals other permits will be required and if you expect to have to preserve specimens you should get written permission from the relevant authorities. If you need to export specimens you will require export permits. For any species (or samples from species) on CITES lists you will also require an import permit to bring them home. In Britain the Department of Environment (address in the appendix) issues CITES import permits. They are only granted on receipt of a copy of the export permit from the country of origin. This means that you must get the export permit several months before you come home in order to get the import permit issued in time. There are severe penalties (large fines and imprisonment) for failing to comply with CITES regulations.

Figure 1.1. Sneferus' bent pyramid at Dolshur, Egypt. In the foreground is the burrow of a Varanus *or* Uromastyx *lizard. Digging for lizards in areas like this can cause great suspicion.*

1.3 Finding literature and maps

Before leaving for the field you should:

1. Know what work has been conducted in the area previously
2. Have as much information on identifying animals as possible.

The easiest way to conduct literature searches is through on line databases such as BIDS or EDINA. But it is very unwise to rely on computer databases alone. Most only contain recent citations and may only cover a very limited range of literature. Leads are often found in the reference lists of other works, by asking people to recommend literature or by sorting through card indexes. Even if useful literature exists it may not be easy to find. *Zoological Record* is one of the best reference sources. It goes back to the Nineteenth Century, comes on CD and lists references both taxonomically and geographically. Much of the literature will not be in the average university library. The easiest way to get it is via inter-library loan services. This is especially true if you expect to have to do a lot of photocopying; the costs to non-members of large institutional libraries can be horrific. But if rare publications are needed or the volume of literature is enormous visits to other libraries will be required. In Britain the Natural History Museum has the best collection. The Zoological Society of London and the British Herpetological Society also have good libraries. A lot of the best herpetological literature is in German, and it may be necessary to recruit translators to get the required information.

The Internet is a very valuable source of information, allowing you to contact professional herpetologists of many disciplines all over the world. It is worthwhile making a list of authors who have produced papers on your subject of interest, or have worked in the same area, and then trawl through the Internet looking for their email addresses. A polite message with a specific question can yield very useful hints on where to find more information. Expedition reports can be an invaluable source of information. In Britain the Royal Geographical Society has a large collection of reports. Similar institutions abroad may also keep expedition reports.

It’s unlikely that you will find maps with enough detail to base survey work on - study areas usually have to be mapped on site. Useful sources of maps in Britain are the Royal Geographical Society Map Room, Stanfords (a shop) and some of the larger libraries. In the host country universities, government departments and non-governmental organisations are potential sources of good maps.

1.4 Finding, making and using keys

Dichotomous keys are the standard way of identifying animals. It is not possible to make identifications from photographs alone. In theory you work through the couplets, deciding which category your unknown creature fits into until you reach the correct species. In practise it can be much more difficult than that. Keys sometimes rely on what seem to be very obscure characters and employ lots of terminology, which no dictionary can explain. It takes, time, practise and discipline to use keys properly. But first you have to find them. Literature searches should have revealed any keys that exist. Otherwise field guides often contain useful keys, or the necessary details can be found in type descriptions and subsequent revisions, often along with references to other useful work. Even if keys cannot be found it is often possible to construct them if you have access to some descriptions and enough specimens.

If you find keys - how do you know if they are any good? There are some truly dreadful keys, often accompanied by the wrong photographs, just for good measure. Find people who may have used the keys before and ask their opinion. If you are lucky they may have improved the keys for their own purposes and will send you a copy. For some taxonomic groups, good keys have been produced which cover most if not all species and can be used to identify living animals. For others, keys may be so old that many species are omitted, keys may demands details of internal characters or there may be no keys at all.

1.5 Visiting museums

Once you have read the relevant literature you are in a position to test your identification methods. The next step is to visit zoo or museum collections and look at the sorts of animals you hope to encounter in the field. I have found it useful to make a theoretical key from the literature and then test parts of it against actual animals. The ideal way to practise is with a collection of live animals in a public or private collection. International Zoo Yearbook or herpetological societies can help you with this. More usually, collections of preserved animals have to be used. Visits to museums are essential for survey work, but staff at museums and libraries are often overworked and however much they want to help they will not be able to spare you much time. In Britain the Natural History Museum and National Museum of Scotland hold large reptile collections. Some of the largest collections in Europe are those in Paris, Frankfurt (Senckenberg), Leiden and Berlin. Published catalogues will reveal which collections hold animals you are interested in. A useful list of catalogues can be found in Crumly (1990). Write to the curator saying what you want to do in the field and which specimens you need to see. Request an appointment well in advance and stick to it.

When you get to the host country a visit to the museum and university collections should be one of your first priorities. Although the collections of books and animals might not be as large as those of the imperial museums you are more likely to find details of collections made close to your study site and are likely to come across new literature. It is much better to find out about this work before you start your project than to discover it while you are writing your report. A great deal of valuable information may be found in the libraries of wildlife and forestry departments.

Section 2
BASIC TECHNIQUES

2.1 Asking questions

It's very, very easy to overestimate the amount of work that can be done in the field and so it is always best to restrict your aims to simple questions that can be realistically answered in the time available. Usual themes are:

1. Species inventory of the site;
2. Densities or relative abundance at the site;
3. Distributions within the site.
4. Aspects of the natural history of selected members of the community (e.g. habitat use, activity patterns, reproductive biology)

Unless keys are available that will allow you to identify live animals it will be difficult to produce accurate species lists without killing and preserving some animals and finding someone who can identify them for you. Unless you are able to catch or mark animals, attempting to measure abundance and densities will be very difficult. Unless the animals you are working with are fearless of mankind or live in very open habitats it is unlikely that you will be able to make many observations on movement or behaviour without relatively sophisticated equipment.

2.2 What is possible

Most expedition surveys aim to be of use in the long-term conservation of an area or of a specific species. Typically they want to be “utilised in management plans”. The most basic data required for management plans is a list of species present (species richness), with an indication of their relative abundance (diversity) and distribution. This data cannot be collected by a single approach, a variety of techniques and experiments are required. For individual species some knowledge of their basic biology can go a long way to help formulate conservation plans. It is rarely possible to collect this sort of data for all members of the community, and it can be difficult to combine studies of community structure with studies of the niches of individual species. Furthermore, sponsors may be sceptical about the amount of work that the team can do in the short time available. For these reasons it’s a good idea to keep the aims of the project simple and well defined, resisting the temptation to make the expedition’s aims mirror long term, heavily funded projects. At the same time, although having very clear research objectives is essential, you also need to be flexible, especially if you are going to work in a little known area and do not have much information about the study sites/target animals before you arrive.

2.2.1 Species richness

The researcher tries catch as many different species as possible. This will require sampling at all times of the day and night, searching and trapping in all microhabitats that the animals of interest could possibly be found in. The researcher must be in a position to describe the climatic conditions under which the surveys were performed and be able to identify every animal found to species level.

Species richness can vary with:

1. Size of study area: larger sites will contain more species than smaller ones
2. Remoteness of study site: remote areas (such as oceanic islands) tend to contain fewer species
3. Elevation; species richness can decline or increase with altitude;
4. Latitude and longitude; the most speciose communities are found around the equator (although the most species rich groups of turtles, lizards and salamanders occur in more temperate zones;
5. Plant diversity or habitat complexity; species richness may increase with habitat complexity;
6. Previous weather; prolonged droughts reduce the richness of communities;
7. Time since last fire or other catastrophic activity; species richness may decline after fires, volcanic eruptions, earthquakes etc.

This data must be collected for each site. Luckily, all are easy to measure. Local people will tell you about fires and the nearest meteorological station should be able to provide summary details of temperature and rainfall for at least several years prior to the study.

In general a few species in the community will be common and the rest will be rare. In a short study it is unlikely that you will catch representatives of all species and the question of how many species you are likely to have missed will arise. This can be estimated by plotting the data as a rarefaction curve (Sanders 1968 in Krebs 1992) which gives some idea of what proportion of species in the community should have been sampled after a given amount of effort. Plot the number of trap hours against number of species caught. The closer the line comes to levelling out the better your chances of having caught all the species present.

2.2.2 Relative abundance and densities

A key element in the study of diversity is the relative abundance of different species at a site. It is nowhere near as easy to measure as species richness. The simplest measurement of abundance is a rank in which the common species is listed first and the rarest last. Alternatively they can be ranked in nominal classes such as very rare, rare, common, very common etc.). In order to produce a quantitative measurement some indication is needed of what proportion of the population of any species is being sampled. In practice, it is usually not possible to determine abundance of whole communities of animals in a short study and the best that can be done is to get a measure of the relative abundance of the commoner species.

Removal experiments measure abundance from the decline in capture rate as individuals are removed from the population (Zippen 1958). One problem with this approach is that removal of individuals allows others to immigrate to the area. This is suspected when the number of animals caught fails to decline as expected. Removing all (or most) of the individuals of a species from a study area is very rarely justifiable.

In mark recapture studies animals are caught, marked and released. This continues until a large proportion (hopefully nearly all) of the population has been marked. An estimate of the total population is made using the ratio of marked vs. unmarked individuals in later samples. Ideally the number of animals caught will be relatively constant while the proportion of marked animals in samples increases steadily over time. Mark recapture methods make some important assumptions that do not hold true for many studies. Essentially, all individuals have to have an equal chance of being caught, catching an individual does not alter its chances of being recaught subsequently and there is no effect of immigration, emigration, births or deaths on the population. For studies of a few months the effects of births can be discounted if it is possible to distinguish juveniles from adults. Capture methods largely determine the success of mark recapture studies. If capture is too traumatic animals will flee the study area. If it involves rewards (such as food in baited traps) animals might come flocking to the study area from all over the district. Catching animals while they are inactive or asleep is the ideal way to conduct a mark recapture study.

2.2.3 Resource use

Common species may be common because they are able to utilise a greater range of resources than rare species. Many rare species have specific habitat requirements that may relate to a single resource. Short-term studies cannot expect to be able to measure niche breadth (Pianka, 1973) but they may be able to identify areas that contain rare species. In order to sample different habitat types within the study area it is necessary to characterise the vegetation in each area. Each habitat type is then surveyed (using standardised techniques) for animals and attempts made to relate changes in animal community composition and diversity to differences in vegetation. (Pianka, 1967, 1986).

Not all expedition fieldwork will be directed towards discovering exactly what species of animals live in an area. Often the object is to collect data on the ecology of certain species within the community. For projects lasting 1-3 months investigations of patterns of habitat use, activity patterns, population structure, reproductive behaviour or diet might be tackled. These studies are particularly appropriate when they target rare or endangered species.

2.3 Producing reproducible studies

Science progresses by careful use of reproducible experiments that provide evidence for or against various theories. Field biologists work under the unhappy affliction that very little of what they do can ever be truly reproducible. Events in a biological system are varied and dynamic and so communities cannot be the same at two different periods of time. Work needs to be planned very carefully to minimise these effects. The best that can be done is to standardise surveys in ways that allows them to be compared with other surveys at the same site and with surveys elsewhere.

The preceding section discussed reasons why species richness might vary with time at any site. We now have to recognise that the *observed* species richness and diversity at any time depends on the levels of activity within the community, which in turns depend largely on current weather conditions. For example if we survey an area twice, targeting active animals, once when the air temperature is 12^{o}C and once when it is 30^{o}C we might intuitively expect to find more active reptiles in the warmer conditions. If our survey techniques targeted inactive animals the scenario might be reversed. We can try to eliminate this source of variation by targeting animals when they are expected to be inactive (e.g. searching for diurnal animals in their nocturnal retreats or vice-versa). But in practise this may result in too little data being collected. As well as temperature, amount of cloud cover, rainfall patterns, humidity or even the phase of the moon might influence the number and types of animals that are caught. In order to be reproducible survey work for reptiles and amphibians **must** include measurement of basic climatic conditions;

1. air and substrate temperatures;

2. cloud cover or light intensity;
3. relative air humidity;
4. wind speed;
5. time since last rainfall.

All of these can be recorded with inexpensive equipment. It is usually most convenient to record climatic data at a nearby field camp.

There are other, more worrying, reasons why two surveys might yield different results. The effort put into each survey will have an important bearing on how many animals it detects. “Effort” is based largely on time, energy and expertise. When surveys are conducted using trapping methods alone, effort can be standardised by using the same number and type of traps over the same length of time. Where surveys collect data by active searching, an individual’s ability to spot or capture animals has an important influence on the results. Although there is little you can do about standardising ability in surveys other than ensuring that each person does an equal amount of work at each site, you should look for bias in results. If sites searched by one worker consistently contain more animals than those surveyed by others, this might suggest that their expertise in finding animals is having an influence on the results obtained.

2.4 Working with local people

Ideally your team will be a mixture of people with and without experience of the animals and the habitat you are working in, but ultimately the results you get will depend largely on how much help you receive from local people, both officially and unofficially. If you are lucky you will find people who know the animals more intimately than anyone else in the world. The possibility of meeting these people by chance is extremely slim. For survey work the best help you could hope to have is from professional animals collectors for the pet trade, who specialise in catching animals without harming them. In countries which export live reptiles and amphibians, the local Wildlife Department licensing office may hold lists of registered animal collectors and be able to recommend some. Do not be put off by collectors who are unable to speak any English. Don't commit yourself to engaging anyone for the duration of the project until you are convinced that they know what they are doing. Trappers are usually paid per animal but for survey work this will bias the results, because you will invariably find yourself working in areas where abundance is unusually high. It is important to survey areas that have low as well as high diversity. Local hunters are also essential companions, but they may not be as good at catching animals without damaging them. Obviously you should never employ children to catch animals for you.

Local people will almost certainly take you at your word when you arrive asking for help and will be very keen to oblige. It is important that you do not cause bad feeling by failing to live up to their expectations. Make it very clear exactly what you intend to do and what help you need. Fix a reasonable daily wage, which you can top up with generous expenses. Wages can be kept pitifully small, but by now most of the expedition’s money will have been spent on airfares and equipment and it is good to spend as much of the remainder as possible locally. Don't make any promises you are unable to keep. A colleague of mine once hinted to a group of children that our soon-to-arrive team might bring them football boots. The following morning I was given a jubilant greeting and had to spend a very uncomfortable hour explaining that he had been talking nonsense.

Sometimes you will find very few animals or even none at all. People who catch animals for a living know that this happens sometimes for reasons that no one can explain. It is an accepted fact of life. They may have trouble sympathising with your despair and desire to improve the situation. Be patient.

2.5 Handling animals

If you can identify animals from a distance there may be no need to catch and handle them. More usually animals must be handled, at least during the early stages of the project. Ideally you should get some practical experience of animal handling before starting fieldwork.

Figure 2.1 A safe way to hold a small monitor lizard.

2.5.1 Bags for animals

Snakes, lizards, crocodilians and chelonians should be transported in strong cloth sacks tied with drawstrings. **Remember that wet cloth bags become airtight and will suffocate their occupants.** Bags should be at least twice as deep as they are wide, with double stitched seams and a fast closing action. Expedition teams rarely have too many bags. On busy days animals have to be left or transported in socks and pockets if there are not enough bags available. Its nearly always cheaper to have bags made locally - take a few samples with you. The neck of the bag is twisted before it is tied and snake bags are knotted twice. At regular intervals cloth sacks should be boiled clean. If faecal samples are being collected bags must be cleaned after every use.

Small lizards should be held between the thumb and finger across the pectoral girdle. Hold larger animals around the pectoral and pelvic girdles, keeping the hands away from the claws. Some long necked species can turn around and bite in this position so it is necessary to hold them by the neck or at the base of the head. Where necessary, gloves can be worn to protect wrists and forearms. Never put pressure on the head and never pick animals up by the tail.

2.5.2 Handling snakes

Although even the most potent serpents would rather run away than bite, they respond to capture in the same way other animals do, by fighting for their lives. The vast majority of snakebites occur when people are trying to kill or catch them or the snakes are trodden on accidentally. Unless expedition members have experience of handling venomous snakes their collection should not be considered. Even with experience, it is worth bearing in mind that snakes in zoos behave very different to wild animals in fear of their lives. If you are employing local people to catch venomous snakes you should be prepared to take full responsibility for any accidents.

The first rule of snake handling is: never touch a snake unless you know what it is. But for our purposes when we know what it is we often have no need to catch it. **So every snake caught should be treated as if it were venomous.** Never handle potentially dangerous snakes alone. Snake catching requires two people (one to handle the snake and one to handle the bag), but not more.

Snakes are delicate creatures and are easily damaged by incorrect handling. Often injuries are not immediately apparent and animals die hours or days later. Whenever possible they should be coaxed into a bag without being handled. Hooks and tongs are the usual way of handling larger snakes. The head end of the animal is kept well away from the handler with the first hook, whilst a second hook may be required to support the bodies of larger specimens. Good balance and concentration are required. Tongs (usually called Pilstrom tongs) are used to grasp snakes behind the head and completely immobilise them. I have found that the use of tongs usually causes scale abrasions unless the jaws of the device are padded with foam rubber. Remember that many snakes feign death in the hope of escaping. Nooses do not work well on snakes.

Most "accidents" with snakes occur getting them in and out of bags. For slow moving species a bagging stick is very useful. It consists of a metal ring attached to a long pole. The bag is held open on the ring and snakes picked up with hooks or tongs are simply dropped in (Figure 2.2A). Often cornered snakes will voluntarily enter the bag without being touched. When the animal is in the bag the, neck of the bag is sealed with a pair of hinged sticks (2.2B) before being tied by hand 2.2B & C). Very fast snakes can rarely be held in the bag for long enough to secure them. Their heads must be immobilised from outside the bag before it can be tied safely. I always knot bags twice using tight knots that can be undone quickly.

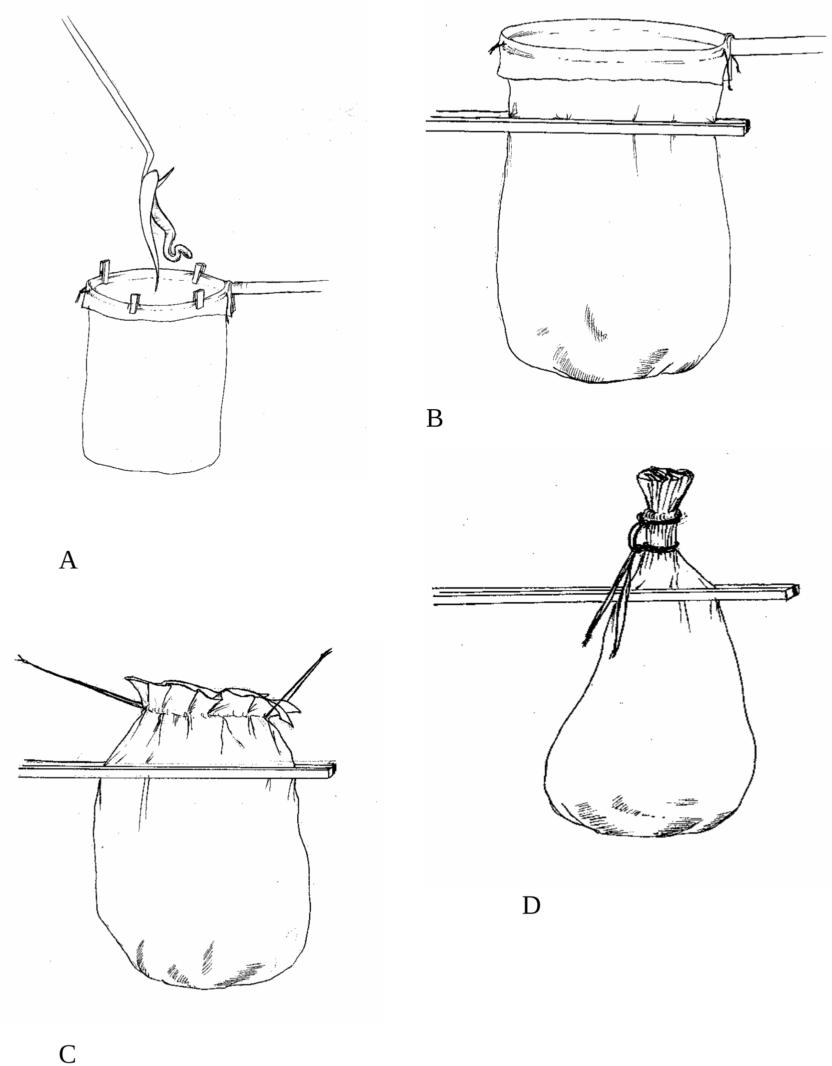


Figure 2.2. Safe procedure for putting snakes into bags

Snakes are quite capable of biting through bags. Therefore bags containing snakes should only be picked up by the handle and always kept well away from the body. Snake bags should be transported in sturdy boxes with the contents clearly labelled.

To get a snake out of a bag to study it, secure the head from the outside of the bag then open it and secure the snake directly before releasing the grip from the outside of the bag.

2.5.3 Releasing snakes

The simplest way to get snakes out of bags is to hold the bag on a tong or hook with a clamp over the neck of the back below the knots. When the bag is untied the snake is unable to escape until the clamp is removed, whereupon the snake can be gently shaken out of the bag if it does not leave of its own accord.

The above notes are given as a guide, but you cannot learn to handle animals (dangerous or not) from books. If you are planning to survey snakes, and venomous species are expected, you must get training before you leave and ideally you should be accompanied by somebody with experience of handling wild snakes.

2.6 Short term captivity

Being caught induces trauma in reptiles and amphibians, but if the experience is short little or no harm is done. If animals are to be kept for less than 12 (amphibians) or 24 hours (for reptiles) they should be kept in the bags they are transported in and released exactly where they were caught. The faster animals are returned to the wild the better.

Always store animals in a cool (20–24^{o}C for tropical reptiles), dark place. I prefer to keep small animals suspended from "washing lines", using separate lines for animals that are awaiting examination, release or preservation. Keeping them on lines prevent them from being trodden on or lying forgotten in the corner.

In some circumstances it may be necessary to keep animals in captivity for a few days (e.g. to collect faecal samples). The dangers facing captive animals are from dehydration, overheating, suffocation and predation. Keep animals individually in smooth-sided plastic boxes or jars with screen lids, a hiding place and a supply of drinking water. Write the field number of the animal on the box (not the lid) with a permanent marker. Larger snakes can be kept in bags for at least several weeks, but they must be checked and given water to drink daily. Wash the containers well and clean off the old field number with a drop of alcohol.

When a lot of animals are passing through the field station it is a good idea to appoint a "zookeeper" responsible for the well-being of animals.

If animals are kept for longer than two days they will probably require food. If they need food they will also need to thermoregulate in order to digest it properly. Over longer periods stress-induced disease becomes a major factor in mortality. If you expect to keep animals for more than a few hours consult a good book on the subject (e.g. Mattison 1987) and ask the advice of vets and reptile keepers before you go. To reduce the danger of spreading disease between animals wash all equipment and hands as often as possible.

2.7 Mapping and GPS

Knowing exactly where you have surveyed is obviously very important. Don’t presume that your local counterparts will have good maps. Take the best ones you can from home and try to find better ones when you arrive. To pinpoint the exact location of study sites, and the routes taken by transects, a Global Positioning System (GPS) is invaluable. GPS is particularly useful when working along rivers, which are too dynamic to be depicted properly on maps. GPS systems do not work well under trees and you should take multiple readings at each spot if possible. The altitude measurements of GPS systems can be unreliable.

2.8 Keeping notes

Failure to keep adequate notes will destroy the scientific content of the project. Fieldwork is often very exciting and in the heat of the moment it is easy to forget to write down essential details. For this reason I carry a supply of bags which already have field numbers written on them. Ball-point pen is best for writing on plastic bags, permanent marker for cloth bags. Do not place labels in the bag with the animal. They can end up causing all sorts of problems.

I also carry a stack of pre-printed forms which I complete as fully as possible before leaving the spot. The most important information is:

1. exact time date and place of capture;
2. habitat description;
3. behaviour;

4. method of capture;
5. weather;
6. tentative description.

The tentative identification should always be given even if it is very basic ("green lizard") as a safeguard against mix-ups. Often the exact location of capture is difficult to find when it is time to release the animal, so I usually tag the spot with a piece of brightly coloured tape on which the field number of the specimen is written in permanent pen. At a later date the exact co-ordinates of the site can be taken with a GPS system. Data sheets are only reliable when they are printed on waterproof paper and written in soft pencil. Make copies of all field notes as you work, and keep the duplicates in a separate place.

2.9 Taking measurements

Handling is very stressful to animals and unless there are good reasons for taking lots of different measurements only the essentials should be recorded. They are:

- sex;
- weight;
- snout-vent length;
- tail length;

These measurements allow the main aspects of population structure to be characterised. Taking all these measurements is straightforward except for distinguishing sex. Chameleons and agamids tend to be clearly sexually dimorphic (i.e. the sexes look different), but in many reptiles the differences are less obvious, particularly in juvenile specimens. Male lizards have a pair of hemipenes that engorge with blood and are everted through the cloaca. The sex of many species of reptiles can be determined by probing the cloaca with a blunt instrument (Honneger 1978). This is a delicate procedure that cannot be learned from a book. Most zookeepers or reptile breeders would be happy to show you how to do it. For some taxa this method is not reliable and the only way to distinguish the sexes without internal examination is by observing any eversion of the hemipenes. The matter is complicated by the fact that some females reptiles have hemiclitoris which, to the uninitiated, look just like hemipenes (Ziegler and Bohme, 1997). Adult male amphibians can be distinguished by the presence of vocal sacs and nuptial pads on the feet.

Measure small animals on a flat metal ruler with a stop at one end. Hold the animal so that its snout is touching the stop and view from the side to read measurements. For larger animals flexible tapes may be more appropriate. Measuring animals alone is almost impossible, and it's always more fun to work in pairs.

For many reptiles weight is not very meaningful because of the large meals they can ingest. Tail circumference, taken with a flexible tape, can provide a very useful index of body condition in animals such as juvenile monitor lizards that ingest large (up to 30% of their body weight) meals and store fat in the tail. In many other reptiles tail base circumference varies with sex. Tail lengths tell you nothing about population structure, but differences in relative tail length sometimes occur between individuals of different sexes, ages and geographical populations. Care is needed when measuring the tails of delicate geckoes and skinks.

Taking precise measurements from struggling animals can be difficult. In studies where the accuracy of measurements is of crucial importance, or animals are dangerous to handle, anaesthesia should be considered. For reptiles both injectable and inhalable anaesthetics have been used. Ketamine is often used to anaesthetise lizards but is considered dangerous for some species of snakes (Font and Schwartz, 1989). Standard dosage is 0.01 ml per 10g of body weight, injected into the body cavity through the ventral surface. This induces anaesthesia in 20-30 minutes, but is very variable and it can take days for animals to recover enough to be released safely. Sodium brevital injected at 15mg per kg of body weight is known to be an effective anaesthetic (Wang *et al*. 1977). Miller and Gutzke (1998) record that 5mg per kg is suitable for rattlesnakes and that high doses are harmful. Halothane is an inhalable anaesthetic with a vapour concentration of 2-4% being the recommended dosage (e.g. Reinert and Cundall, 1982). However it is difficult to control vapourisation in warm conditions and mortality has been associated with its use in some snake species (Aird, 1986). Methoxyflourane is less susceptible to over vapourising at higher temperatures and is the standard anaesthetic used by zoo vets, in Europe at least. Animals recover quickly and completely. Its only disadvantage is its cost. You should consult a vet for advice on using anaesthetics.

2.10 Taking temperatures

I find it best to take two sets of temperatures, one recorded at a convenient fixed location (such as camp) and another taken wherever an animal is found. This temperature should reflect the ambient temperature experienced by the animal. Usually this means taking both an air temperature and a substrate temperature. The problems arise largely in deciding where exactly to take the temperatures from; thermal gradients can be extremely complex in some habitats, such as sunny riverbank, where a huge range of temperatures occur at the land/water interface. The problem can usually be solved by recording the temperature where the animal's head was first seen.

It may be a good idea to record the body temperatures of newly caught reptiles. The standard way of doing this is to insert a fast acting thermometer or thermal probe into the cloaca. It is imperative that the temperature is taken within seconds of capture, particularly for small animal who respond rapidly to changes in thermal conditions. Data on body temperatures might lead to clues about the thermal niches of individual species or suggest differences in the thermoregulatory behaviour of different members of the community. Gans and Pough (1982) and Pough *et al.* (1998) provide a detailed introduction to this topic. Avery (1982) discusses the limitations of taking single body temperatures in field studies.

Figure 2.3. Taking the body temperature of a Puff adder.

2.11 Taking DNA samples

There are many good reasons to take DNA samples even if you have no idea what to do with them. Well-labelled samples are very easy to take and store, and it may not be difficult to find someone who can make use of them. Where identifications are uncertain or tenuous and no whole animal specimens are collected, a DNA sample can make all the difference between worthwhile and worthless surveying.

The literature recommends storing samples in liquid nitrogen, but for most expeditions this will be too expensive and laborious. I take small (0.5-2mm) pieces of tail tissue (either tips or notches cut out of the top of the tail) and put them straight into cryotubes half full of absolute ethanol with a label. Hundreds of such tubes fit into a small bag. Whenever possible I store them in a freezer. Recent reports (Bridner *et al.* 1996, Clark 1998) indicate that high quality DNA can be obtained from sloughed reptile skin.

You must be aware that legislation often makes no distinction between DNA samples and whole animals. You will usually require permits to take DNA samples from animals you catch and almost certainly need them to bring it back home. The best way to find a use for your samples is often with a PhD student in the host country. Failing that, leave them with a local institution. Unfortunately electricity is in short supply in many universities, let alone the equipment to perform molecular analyses. In these cases it is best to apply for permission to bring the material back home and keep it frozen until a use can be found for it. Many workers are currently using molecular tools to investigate the phylogenies of different groups, and the chances are that your material is the only available DNA for those species. If you advertise your samples on the Internet be careful to screen requests for DNA from idle fools who have no intention of using it, but can't resist the idea of having it sent to them.

2.12 Looking at diet

Many reptiles defecate in response to capture and so if animals are being caught for examination or measurement, any faeces produced should be labelled and stored for subsequent analysis. There are little or no health risks associated with reptile faeces. Samples are best stored in individual tubes of 70% ethanol with labels written in soft pencil on parchment paper. Dried samples are susceptible to attack from fungus or being crushed to powder. For analysis rinse and tease samples apart and list the parts found. If a reasonably large, properly labelled collection can be made, and there is no time to work through it the collection should be left at a local university where it can provide a local student with a very interesting thesis project. Diet analyses of these types do have some serious limitations; Frogs or slugs in the diet are rarely detected in faeces and it is difficult to calculate what time span of feeding the sample represents. Often, however, it provides baseline data that is not available elsewhere. Animals preserved as specimens will probably have food items in their alimentary tracts. You should resists the temptation to remove these until the animals have been positively identified and lodged in the relevant collection, unless you are unlikely to see them again, in which case they should be neatly removed and stored in a small jar bearing the same number as the specimen they have been taken from.

Stomach flushing can be a very useful way of determining diet without harming animals. A tube is inserted into the animal's stomach through the mouth and food present is simply rinsed out with water. A variety of methods have been described for lizards, amphibians and crocodilians, details of commonest methods are given in Legler and Sullivan (1979), Fitzgerald 1989, Taylor & Webb 1978, Leclerc & Courtois 1993 and Rivas *et al.* 1996. Stomach flushing can be very time consuming and it takes practise to perfect the technique. The methods are harmless if care is taken not to put water into the lungs or cause damage to the jaws or alimentary tract. Usually a wooden or rubber "bit" with a hole to accommodate the water tube is put between the animal's teeth to reduce the danger of injury. Amphibians must be anaesthetised prior to handling of this kind. For snakes a method of "passive manipulation", by which food in the stomach is gently massaged out of the stomach and up the oesophagus (Shine 1986) is more commonly used than stomach flushing. If you are considering using these very useful techniques you should seek the advice of an experienced vet beforehand.

2.13 Looking at disease

Parasites and diseases probably play an important role in the regulation of most animal populations. They are only rarely the subject of field studies, partly because identification of pathogens is difficult. Make a note of any animals that appear ill and consider preserving any that are obviously close to death. Faecal samples may contain evidence of internal parasites and other pathogens. Preservation is described above.

External parasites occur on most reptiles and are a neglected area of study. They can be collected from captured animals by dabbing them with oil/alcohol and removing them with forceps, taking care not to leave jaw parts embedded in the host animal. Make a note of the position of each parasite and preserve specimens of each type in alcohol.

2.14 Marking animals

The main advantages of marking animals are to stop you counting them twice and to allow individuals to be recognised. Sometimes natural markings can be used to identify individuals (e.g. McDonald *et al.* 1996) but more usually it is necessary to give individual animals a distinguishing mark. This will almost inevitably affect the animal adversely, and so it should only be done when identifying animals caught previously is essential. For studies of reptiles or tortoises lasting only a few days or weeks, blobs of enamel paint are the simplest marks to use. Loses due to skin shedding will be few if you apply the paint to fresh skin. If none of it looks very fresh, clean patches with a little ethanol and wait for it to dry before applying the paint. I use a variety of colours and use two blobs of paint at different positions. It's very important to work out a simple systematic method in advance. Applying the paint over distances (without catching the animals) is an interesting option for animals that are easy to see but difficult to catch. Firing oil based dyes from a paint pellet gun would do the trick. The necessary equipment costs less than £100 (addresses in appendix). Simon & Bissinger (1983) suggest that the colour of the paint used to mark reptiles does not influence survivorship. Obviously there is some conflict between the need to make animals recognisable from a distance and the need to retain as much of their natural cryptic pattern and colouration as possible. Behavioural changes will be kept to a minimum by not painting over the pineal gland (on the top of the head) and using as little paint as possible.

For longer studies a mutilation method is necessary unless you can afford implantable tags. Passive integrated transponders (PIT Tags) are still too expensive to be used for most projects. They consist of a chip sealed in glass which is implanted in animals using a simple syringe gun and can be read with a barcode scanner. Germano & Williams (1993) report that some tag loss occurs even when the chips are implanted into the body cavity. Other workers recommend implanting the tags into the lymphatic system to reduce losses and possible damage to the animals.

Unfortunately, cutting off pieces of toes is still the commonest way to mark limbed reptiles and amphibians. Schemes for toe clipping programmes are given by Hero (1989) and Waiddman 1992. However, there are serious objections to toe clipping based on the fact that removing toes is likely to adversely affect animals' behaviour and prospects for future survival. Wherever possible a less damaging method should be employed.

Many lizards have a crest running along the tail into which permanent nicks can be cut without drawing blood. Using two nicks of different lengths a large number of individuals can be marked. This method is not suitable for round-tailed species. Beads sewn into the tail crest with monofilament wire can provide identification from a distance, where paint marks are not long-lived enough or the animals are too small for individual marking by tail notching. Using ten colours and three beads on each animal allows up to 1000 individuals to be marked for at least 18 months (Hudnell 1982, Fisher & Muth 1989).

Soft-shelled turtles can be marked very simply by making holes in the carapace with a paper punch (Doody & Tamplin 1992). Tortoises can be marked by sawing small notches into the edge of the shell.

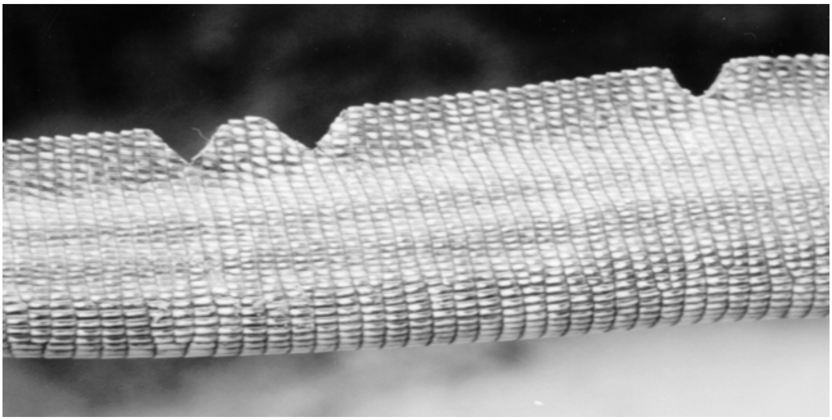


Figure 2.4: Tail notching is a cheap and harmless marking method

Figure 2.5: Mark recapture studies of tortoise can last many decades.

2.15 Following animals

It would be very interesting to know about the movement patterns of individual animals, but this is only rarely possible during survey work. The main methods used are telemetry, spool & line methods, use of fluorescent markers and individual footprint recognition.

Telemetry requires a larger budget than I have ever had, but it is an ideal method to use when the biology of particular species is under investigation. As well as indicating the location of an animal, transmitters can be adapted to give information on body temperatures, activity and even orientation. Kenward (1987) provides a good introduction to the subject. If the main purpose of the study is to survey entire communities it is very unlikely that either time or money will be available to make telemetry a serious option.

Spool & line methods are cheap and ingenious, but only if you can get them to work. A spool of thread is attached to an animal which plays out a line showing exactly where the animal has been (Thompson 1992). The main problems are attaching the device to the animal and keeping it small enough to be unobtrusive and not influence the animal’s behaviour. Data obtained is only useful if it can be convincingly demonstrated that the apparatus is unlikely to influence the animals movement patterns. The unfortunate frog depicted in Heyer *et al.* (1994: p.154) looks unlikely to get very far and it is difficult to imagine that having such a contraption strapped to your back would not influence behaviour to some extent. Similarly Scott & Dobie's (1980) turtle is unlikely to behave normally with its cumbersome load. Wilson (1994) recommended the use of cocoon bobbins, where thread is fed out from the centre of the spool rather than from around the outside.

Trails of fluorescent dyes or powders, detectable with ultra-violet light, can be used to track animals (Fellers & Drost 1989). These methods can provide more accurate information about movement than spool and line tracking. The dye reservoir is created by simply dipping animals in fluorescent powder (e.g. Butler & Graham 1993), using leaky pouches of dye attached to animals (Blankenship *et al.* 1990), impregnating a piece of rabbit fur with powder that is fixed to the animal (Keller 1993) or applying fluorescent pigments to yarn tags which are fixed to the animal and drag along the ground (Windmiller 1996).

Windmiller (1996) describes the use of small capsules filled with a long lasting luminous chemical such as Cyalume (see appendix for suppliers) which are attached to animals and glow for about 12 hours, allowing movement to be monitored over a night (see also Clark & Gillingham 1984)

The final method is by far the most exciting. In places with large expanses of soft sand or mud, footprints can provide very accurate data on movement patterns. Normally it is very difficult to distinguish the footprints of one animal from another but Tsellarius & Cherlin (1991) describe a method that allows individual recognition and precise records of movement of large lizards, which deserves wider use. A distinctive shape is cut from 2mm belt leather that is stuck onto a slightly larger piece of 2mm thick chamois leather. This is stuck to the bottom of the hindfoot with superglue (having first cleaned the skin with ethanol) before the animal is released and if the creature's footprints are encountered again over the next few weeks or months they will bear the unique impression of the footmark. When the animal sheds its skin the mark will be lost.

2.16 Taking specimens

You should only take specimens if you have good reasons for doing so and the consent of the relevant authorities. In general, the only reason for taking specimens is because identification is uncertain. If you are lucky you will come across a dead animal in good condition that will serve as a specimen. Otherwise, preserve an adult animal as soon as possible after capture. Kill it with an overdose of anaesthetic before taking measurements. Amazingly, some people still favour formaldehyde as a preservative. This evil substance is a potent carcinogen and thoroughly noxious and harmful in all respects. Its only conceivable use on expeditions is to destroy the rancid smell of some members' feet. We use ethanol to fix and preserve animals, at 70% strength. It can be bought from chemical suppliers in most cities at about 95% and diluted with water. Absolute ethanol is much more expensive and is used only to preserve DNA samples. In remote areas, ethanol can often be purchased from illicit local stills. Test the strength by setting light to a few drops. It is unlikely that rural distillers will be able to supply ethanol by the litre and so adequate supplies should be taken to the field whenever possible. Where ethanol is unavailable isopropyl "rubbing" alcohol can be used instead. Transfer the specimen to ethanol at the first opportunity.

Label the animals with a tag that will never come off. The safest option is to buy labels specially made for the purpose, despite their expense. The label should carry the date, collection location and field/specimen number of the animal, written in Indian ink. Fix specimens by arranging them with the limbs and tail in the desired position, cover them with paper soaked in alcohol sealed in a plastic bag for a few hours. Very small animals can just be dropped into alcohol when they are fixed, larger ones should have alcohol injected into the body cavity and into any large muscle masses. Keep a separate notebook to list specimens in, recording all the relevant data for each animal. Make multiple copies of this book as soon as possible. Specimens are best kept in plastic, shockproof jars with very close fitting lids. It is often necessary to seal up the lids with tape for transportation. Check the specimens regularly. If they begin to look a bit shrivelled add more water to the preserving fluid. If they begin to smell add some stronger alcohol. Specimens belong to the country they have come from, and unless exceptional arrangements have been made they should be lodged in the collection of a local institution. If they have to be identified elsewhere you should make sure that the specimens are eventually returned to their rightful owners. If you take specimens the animal must be anaesthetised before being killed. All field biologists say they do this, but an alarming number just drop living animals into preservatives.

Schueler (1981) describes a method of storing amphibian skins that are dried on waxed paper then and glued to a small board. This provides a very useful identification aid that is lightweight, takes up very little space and is easy to prepare.

2.17 Photography

2.17.1 Cameras and flashguns

It's a very good idea to take photographs of all the habitats you work in and the species you find there. Careful photography can provide a great deal of information that is difficult to collect from live animals in the field such as scale characteristics. It is usually impossible to get a positive identification of an animal from photographs alone, unless they show all important detail; ventral, lateral and dorsal views including details of scalation round the eyes and on the underside of the feet. Such photographs should always include a size scale.

The drawbacks of photography on expeditions are the expense and the very real danger of destroying camera equipment with dirt and water. In the field many good photo opportunities are missed because the mud encrusted photographer is unwilling to soil his expensive camera. Fancy electronic, autofocus, autowind cameras are not suited to expedition work. Much better is a second hand mechanical SLR camera that will still work when the batteries are taken out. Pentax, Nikon, Minolta or Olympus models cost £80-£200, take pictures as good as those from modern machines and are much more likely to keep working after months of dampness, dirt and impact. To focus down to 10cm (necessary for small lizards and frogs) use screw-on filters or extension tubes. Alternatively special micro or macro lenses can be used. These are more expensive but give better results. Close-up photography has two basic problems; pictures need a lot of light and the depth of field (i.e. how much of the picture is in focus) is small. Good pictures depend on keeping the aperture size small and the shutter speed short. Extra light is usually provided by a small flash-gun mounted on top of the camera or above/to the side of the subject. Older cameras have flash lead sockets that allow the flash-gun to be used in either position. The more expensive option is to use a ringflash that fits on the end of the lens and provides a more even light source.

2.17.2 Film

Slide film is preferred because colours are better, images can be projected and they give better quality reproduction in publications. Modern scanners, printers and software have eliminated the problems of making good quality prints from slides without enormous expense.

I use Kodachrome film because it produces very high quality slides that keep their colours for years. Only Kodak can process the films and if bought in the UK the price includes developing. The film can stand a bit of heat and is available in 25, 64, 200 and 400ASA speeds. Also it produces good results over a relatively wide range of exposures and does not enhance colours the way other films do. Finally it's inexpensive. 1998 price is about £6 per film (including developing). Cheap sources for camera film of all types (and specialist batteries) can be found in popular photography magazines.

In the tropics pictures come out best in the early morning or late afternoon. In the middle of the day the light is too harsh and the glare from the sun makes photographs very contrasty and difficult to expose properly.

Animals are best photographed *in situ* wherever possible. This is more often possible with creatures such as frogs and geckoes that are found at night. Otherwise it is often necessary to catch the animal and manipulate it to get decent pictures. A small box with scenery such as stones and branches can be used to stage photographs. Alternatively lizards can be tethered by one leg using a piece of dental floss and the picture taken in such a way that the tether cannot be seen. For very close up photography it may be necessary to anaesthetise the animal. If you are taking pictures of many similar species it's a good idea to include some method of identification (e.g. field number) on each picture. To avoid confusion, I always write a number in marker pen on each film and record the counter setting for each picture taken.

2.18 Safety and ethics

All members of the team have a duty to do as little damage to the study site and its flora and fauna as possible (with the exception of blackflies and mosquitoes). Damage should be minimised by careful attention to the amount of trampling that occurs and by disturbing animals as little as possible. However, the only way not to harm the animals you are studying is to leave them alone. Chasing and catching animals is stressful and the more handling the animal receives the less it is likely to survive the experience. In this respect reptiles have great advantages over other vertebrates. They tolerate biologists very well. Still they have to be treated with care and sensitivity. The basic rules are to keep handling to a minimum, keep captive animals in the dark and release them where they were caught as soon as possible.

Obviously safety is of paramount importance. Reptile and amphibians projects require no more than the usual precautions for working in remote areas. From my experience the most important of these is to work in groups of at least three people, watch where you step and where you put your hands, carry plenty of water, don't hesitate when grabbing animals and if you are bitten keep calm and don’t try to pull or shake yourself free. Clean wounds

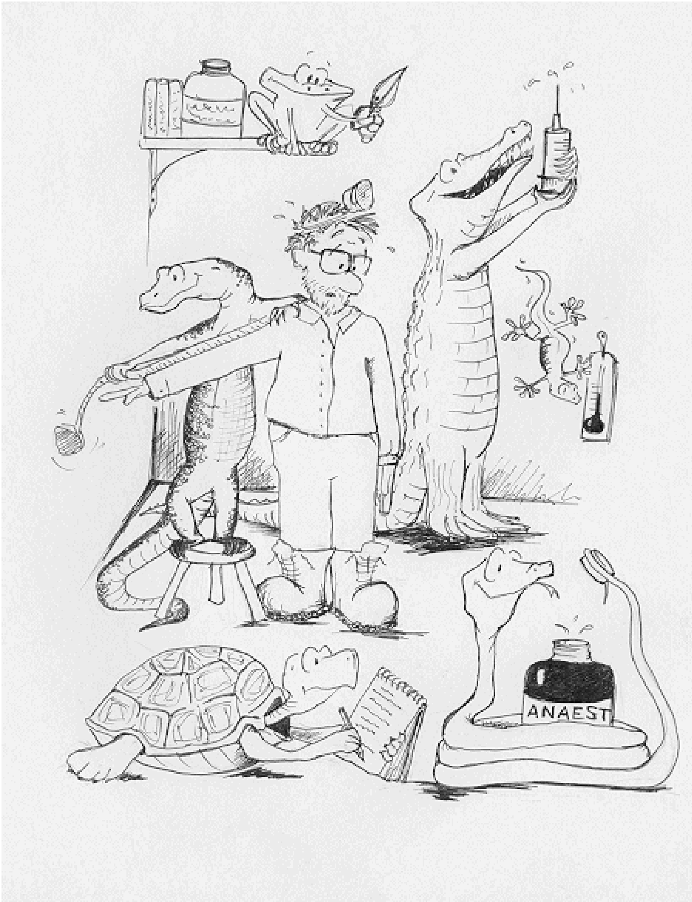


Fig 2.6. Treat study animals with the care and compassion you would like them to show you……

with iodine immediately. Never handle animals after drinking alcohol and remember that many animals react to handling differently according to the time of day. Some nocturnal snakes are much less disposed to bite during the day, but may defend themselves ferociously during the night. Lizards are generally harmless creatures and only the largest species can injure a person. An unfortunate error in Jermy and Chapman (1993) states that Asian lizards have no teeth. Alas, if only it were true. All lizards have teeth and in general the amount of damage they can inflict is proportional to their size. Some Asian lizards are the largest in the world. Finally, animals of the same species can have very different temperaments. Even if most individuals are placid there will inevitably be some that react to biologists with a bit more spirit.

2.18.1 Snake bites

Snakebites are extremely rare among sober people with their boots on. The vast majority of terrestrial snakes are harmless and most bites from "venomous" species inject no venom and complete recovery is usual. But envenomated bites do require medical treatment which is rarely available in remote areas. Envenomated bites are treated with an antivenom usually made from horse antibodies. Because it carries a risk of anaphylactic shock it should only be administered by trained medical personnel. The recommended treatment for snakebites changes from time to time. For up to date information on recommended treatments and antivenoms contact the Snake Venom Unit in Liverpool (address in appendix). Dry antivenoms do not require refrigeration and you may decide that you want to take some with you. If so you must also take drugs necessary to treat anaphylaxis. If you have no medical doctors on the team make contact with the nearest doctors or hospitals to the study site before you start work, leave the treatments with them **and make sure they are acquainted with modern treatment methods**.

Section 3

SURVEY TECHNIQUES

3.1 Asking people

People act as important predators of many reptiles and a few amphibians. Crocodiles and large lizards are often killed for their meat or skins, snakes are often killed whenever they are seen. Documenting patterns of human use of animals is an important component of conservation based studies in threatened areas, but it needs to be carried out with care and sensitivity. Around protected areas local people often fear persecution from the authorities for poaching and they may be understandably reluctant to admit to hunting anything other than rats. Ideally you should attempt to document the methods used to catch animals, how many are taken, the best and worst times of the year to find them, whether particular age classes or sexes are targeted and whether the primary purpose is for meat or leather. If you can co-operate with hunters and persuade them to work within the confines of scientific methodology your survey will be very fruitful. If you are searching for a particular animal, without success, markets in towns of all sizes might be worth checking. Sometimes it is the only source for elusive species. Reptiles are usually sold for meat, leather or medicine. The larger the market the less likely the vendor will be able to supply accurate locality data.

Figure 3.1. Markets can provide gruesome evidence of elusive species.

Where people eat reptiles it may be worthwhile requesting the intact alimentary canals and reproductive organs from the chef, which should be labelled and stored in ethanol immediately. Try to measure the animals before they are cooked.

If possible, it is a good idea to take along a set of photographs of the animals you are interested in, so that you can show them to local people when asking for help. You might learn about animals that occurred in the area within living memory but are now absent, or come across animals that locals consider very common but you have found to be absent or rare. To guard against the over imaginative, I always include a few pictures taken on another continent.

3.2 Searching for reptiles

Fig 3.2. Whole communities of reptiles and amphibians can be found in abandoned termite mounds, but finding them can be arduous and dangerous.

A reptile hunt is a journey into wonderland. Daytime strolls through the wilderness rarely encounter mammals and it is virtually impossible to catch them harmlessly. In contrast reptiles are much easier to find and offer better prospects of capture. The result is very pleasant strolls through the countryside, punctuated by phases of exquisite excitement and vigorous exertion. The methods used during searches depend on the animals you are looking for. To do a thorough survey of an area it is necessary it look everywhere; on the ground, below the ground, in trees and bushes, under stones etc. Unless you can identify all the animals in the study site by sight you will need to catch them.

Systematic searching can be done on quadrats or transects. The only practical difference between them is that transects tend to be less square. I use transects when I want to sample a variety of different habitat types in a patchy environment or along riverbanks. If the animals of interest are likely to be active during the survey, transects will result in a higher encounter rate because there is less prior disturbance when walking in a single direction. In practise, I have found transects to be very difficult to define in terms of area, especially when search teams are large. As well as the difficulties of ensuring that everyone walks in the same direction and keeps the correct distances apart, many interesting things are invariably spotted just beyond the study boundary. The temptation to investigate is too much for all but the driest mind, and as a result the area of the transect cannot be calculated with accuracy, although quantitative measures of length or search hours are possible. I have found that I finish walking transects unconvinced that my measure of area has any basis in reality. The more effort to keep the measurement of area right, the less fun the work becomes. My favourite transects are long convoluted ramblings that get home just in time for supper. In contrast quadrats are much easier to get right. They are not much good when the animals are encountered during their activity, because all the animals in the quadrat are alerted before even a small area of the sample site has been searched. They are best used when the search is for signs of the animals' presence such as burrows or tree refuges, which are not affected by disturbance. It is much easier to work within the quadrat boundary than the narrow confines of a transect, but in both cases the study sites should be measured and marked out at least a day before fieldwork to ensure accuracy.

As well as the environmental and climatic data discussed in Section 2, some other data must be recorded; the exact position of each quadrat and the start and end points of transects (ideally by GPS), the number of people carrying out the survey and the start and finish times.

3.3 Capture techniques

3.3.1 Hand capture

Some lizards will freeze when they are spotted by biologists, but most will run for shelter at the first opportunity. In some open areas it is worthwhile trying to outrun animals and grab them before they can escape into impenetrable thickets, but more usually you have to creep up on them.

Grabbing fast moving animals without hesitation is a very important skill for field zoologists. The best way to learn is with a patient teacher and an endless supply of lizards, or a suitable substitute. Wise students spend a lot of time practising; grabbing locusts or cockroaches and creeping up on frogs and dragonflies and immobilising them in a swift and harmless embrace.

The best way to catch any reptile that cannot retaliate with a savage bite is to pin it to the ground with the flat of the palm. Once it is immobilised this way it can be picked up at leisure. Animals should never be restrained by their tails because they will turn round and bite you or they will drop their tails and leave you with a wriggling, unidentifiable tip. The vast majority of lizards can be caught with the flat of the palm, the exceptions are larger species; tegus, iguanas, gilas and goannas. These animals are capable of inflicting severe bites and they must be caught by immobilising the head, usually by grabbing the creature just behind the neck and, a fraction of a second later, grabbing the pelvic girdle or tail base. Hesitation is the cause of most escapes and injuries.

Many animals will seek refuge in thickets of vegetation. Durden *et al.* (1995) increased their capture rate by laying out artificial burrows made of cardboard or plastic tubes and setting them around bushes known to contain reptiles. With luck, when the animals are frightened out of their hiding place they will make straight for the “burrows”, where they are easily caught.

Most snakes are encountered moving along the ground or basking close to their shelters. Snakes on the ground should be immobilised with firm pressure across the top of the head rather than the neck. Many snakes shelter in burrows or old termitaria. Snakes cornered in holes usually make a rush for freedom and can be immobilised by keeping a hook just above the burrow entrance during digging. Tree snakes are most easily captured by grabbing them with pilstrom tongs as close to the neck as possible and then unwrapping them from their branches tail first. Don't try to pull the animal off the branch because you will damage it. One person keeps the head immobilised whilst the other unwraps the body and takes its weight. If the animal is really tangled it is best to let it go, tail first. Do not try to catch tree snakes with fishing nets, nor from canoes. Water snakes are sometimes caught in basket traps intended for fish or drowned in fishermens' nets. From boats the best way to catch them is by grabbing them with hooks or pilstrom tongs as they swim past. Water snakes tend to be very fast and agile. Lutterschmidt & Schaefer (1996) describe the use of inexpensive polypropylene mistnets set across streams and small rivers to capture swimming snakes. The nets can be set singly or arranged to form a baited enclosure. Seigel *et al.* (1987) provide useful information on snake catching techniques.

3.3.2 Catapults

For fast moving lizards, or those in otherwise inaccessible places, catapults can be an ideal capture method. A heavy duty rubber bands fired at a lizard can stun it for a short period, after which they usually make a full recovery (Brain 1959). A scientist with an international reputation for accuracy with this method recommends B.F. Goodrich size 107 rubber bands which can stun reptiles of up to 40g over an effective range of 5-7m. Normally you should aim for the pectoral region or tail base to stun animals and only aim at the head if you intend to kill it. It is often best to work in pairs when hunting with this method, so that one person can quickly grab momentarily stunned animals before they recover. We adapted the technique for slightly larger (up to 70g) lizards by firing 0.75g seeds from black widow catapults. Mortality rate was 8% and usually occurred when animals were hit on the head or limbs rather than the base of the tail.

3.3.3 Noosing

Many ground dwelling and arboreal lizards can be caught with a simple noose on the end of a stick. For small lizards waxed dental floss is the preferred material. It is stiff enough to stay open and smooth enough to give a rapid response when tightening. Small daft lizards are often attracted to the noose and try to eat it. It is a simple task to slip the noose over its head. I have never found this method destructive. Skinks and other tubular creatures whose heads are no wider than their bodies can be difficult to noose. The best way is to work the noose over at least one of the front limbs before tightening.

Lizards that are cornered on the ground but are too dangerous to pick up, or those that sleep on branches, can often be caught with a rope noose attached to a long pole. The most convenient pole to use is a telescopic fishing rod or pole. Animals sleeping in trees can sometimes be snared without disturbing them. Again the small daft ones will respond by jumping off the branch into captivity, but larger ones will hang on like Joseph and have to be teased off by shaking the branch, prodding the creature with a stick, or by climbing up and unwrapping the animal by hand. Simply tugging at the noose will have no positive effect at all, for the lizard would rather choke to death than let go of its safe perch. Most of my attempts to noose lizards in trees have failed because the animals jump from the trees before they can be noosed. Recently I have greatly increased my success rate by holding a seine net under the tree and enticing the lizard to drop into it. The method is particularly invaluable when the lizards rest on branches overhanging water.

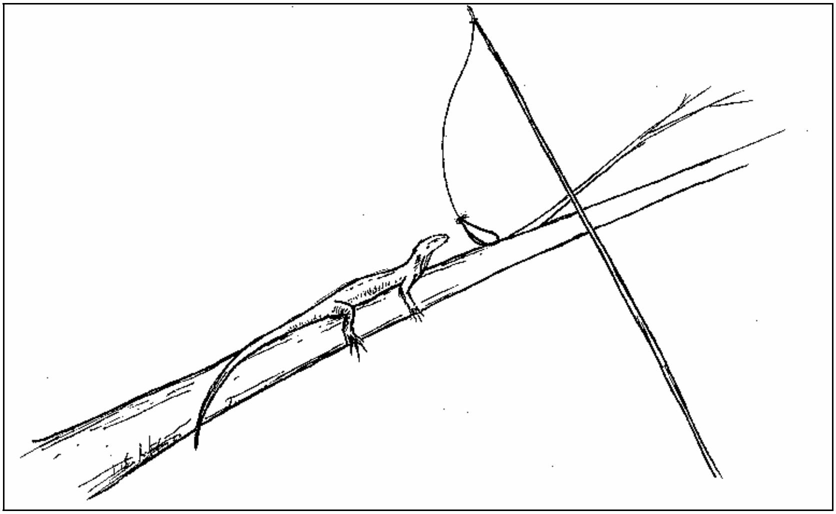


Fig 3.3. Noosing an arboreal lizard

Lizards that cannot be noosed may be persuaded to bite at an insect or worm on the end of a piece of line (dental floss or fishing line) tied to a pole. When the lizard bites the bait the pole is jerked, and the animal is either launched into the air and is caught before it hits the ground or else it refuses to let go of its prize and remains fixed to the end of the line (Strong *et al.* 1993, Durden *et al.* 1995). Witz (1996) describes the conversion of a standard bolt retriever into an ingenious device to grab smaller lizards without hurting them (Fig.3.4). Durtsche (1996) describes the use of a pole with a sticky pad attached which can be used to catch animals that are otherwise inaccessible.

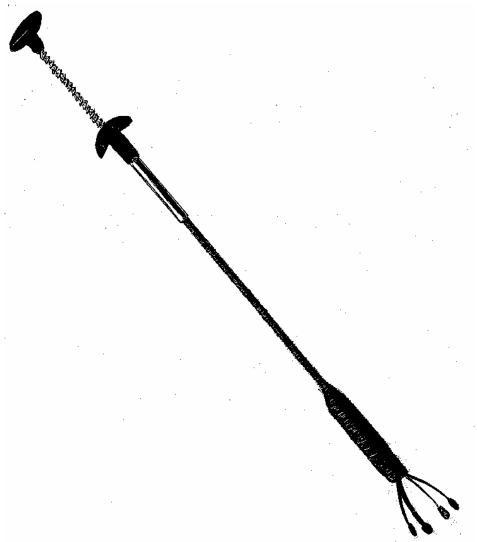


Fig 3.4 Lizard grabber, after Witz (1996).

3.3.4 Other methods

The most delightful technique, for catching small shy lizards that live on tree trunks, was published just before we went to press. Paterson (1998) ties bridal veils around branches, above the lizards that are to be caught. The veil is tied so that an overhang is created, which lizards either refuse to cross or get stuck in. Having cut off its escape routes the animal can then be caught by hand much more easily. Mosquito netting could be used instead, but the work will not be as romantic.

3.3.5 Looking in leaf litter

Many small skinks are found in leaf litter. Ideally they are collected by sifting through leaf litter by hand, but, if there is a danger that venomous animals are lurking, they can be found by turning over rotting vegetation with a rake on the end of a pole. Brattstrom (1996) describes a “skink scooper” which consists of a plastic box or square bucket into which leaf litter and its occupants are swept with a hand. Using a box in this way greatly increases the capture rate of small litter dwelling reptiles.

Figure 3.5. A tiny leaf dwelling skink, only found after intensive searching.

When looking under stones and logs the object should be levered up on a long pole and another person be ready to dislodge the animals with a stick (not a limb) and grab them. Always replace rocks and logs exactly as they were. Soule & Lindberg (1994) describe the use of a device for rolling logs, called a peony to lift large (up to 450kg) rocks in water up to 1m deep.

3.3.6 Animals in holes

Most reptiles use holes in trees or burrows as shelters. In order to produce a full inventory of the reptiles in an area it is essential to identify the occupants of holes. I have little experience of lizards in tree holes, but my greatest teacher was an expert in the art. When a lizard head was sighted poking out of a tree hole and there were no firearms to hand, a Landcruiser with a large winch was employed, the tree felled and the branch of interest sawn off and taken back to camp. In many cases deforestation is not an option and unless the hole is very small it may be impossible to get the occupant out. Patient biologists may wait quietly for the animal to emerge from the hole in the hope of catching it with a noose or catapult, but lizards like to take their time.

Reptiles can never be smoked out of holes in the way that mammals are caught. They will suffocate to death rather than leave the hole. Trapping at tree holes can be accomplished with sticky traps (I have had no success with Sherman traps placed in trees). Another method uses a flexible wire hook which can be used to manipulate lizards into a position where they can be grabbed with a pair of forceps (Bedford *et al.* 1995). The hook must be perfectly smooth to avoid damaging the animal, which is hooked around the tail base, with great care taken to avoid hooking the cloaca. For this reason it is important to have enough light to see the animal with. A torch or mirror will usually suffice. Zani & Vitt (1995) reported 96% success rate catching lizards in trees by identifying the holes containing animals and covering the entrances with minnow traps, filling in the gaps (and other holes in the branch) with tape or cloth.

I have heard reports that using a short acting noxious solvent gets snakes (and presumably lizards) out of holes, but the few times I have tried this method have been unsuccessful. The method is employed by pest control companies who should be consulted for advice. It is worth trying. A net over the entrance will catch any animals that bolt and bear in mind that there may be more than one creature in the hole!

Burrows are more accessible than tree holes. They can be dug up, and this is the usual and most certain method of catching lizards, but digging destroys resources that might be a limiting factor for animals in the area. Destroying the lizard's home will force it to move elsewhere and may effect other animals as well. Unless a short acting solvent can be found (see above), attempts can be made to trap animals as they emerge from burrows using sticky traps, nooses or baited box traps.

In an ideal world, we would examine burrows using a fibrescope or videoscope. These flexible endoscopes can be pushed to the end of the most tortuously winding burrows and the animals inside recognised with the minimum of disturbance. Unfortunately they are very expensive, but if you are working on an endangered species and expect to find it only in holes, this is the method you should aim to use.

Figure 3.6. Large lizard well hidden in a burrow (photo: Nicky Green).

3.4 Night-time techniques

Many reptiles, and nearly all amphibians, are easier to find at night than during the days. Methods used to survey animals at night are not very different from those used for diurnal studies. Repeating quadrat or transect work at night is necessary to get a better picture of species richness and diversity at any site.

Good head torches are essential for night-time work. They cannot be bought in most countries so you should take enough to allow the whole team to work at night. In the past I have made the mistake of trying to economise by buying cheap headtorches, but they are not reliable enough. Large teams working at night get through massive amounts of batteries. Each member of the team should carry spare bulbs and batteries at all times. In general, it’s no fun being lost in the jungle in the dark. Consult a specialist company for advice about using rechargeable batteries. I have never found it feasible, even with very large teams.

Looking for eyeshine is a very good way of finding crocodilians and many geckoes at night. By keeping the torchlight fixed on the eyeshine animals can often be induced to remain still until you can approach close enough to identify or catch them. It can take a little time to learn to distinguish the eyeshine of geckoes and spiders from a distance.

Some diurnal reptiles are easier to survey at night. For monitor lizard studies I often identify burrows during the day, mark them and return at night to see if the hole is occupied. During the day only 10% of holes are occupied, looking at night raises the encounter rate to about 45%.

Sites that will be surveyed at night-time should always be visited in daylight first. Mark the route and borders of the site with some reflective material and make such you know the location of anything potentially dangerous like swamps, cliffs or minefields. Obviously great caution is needed when conducting night-time work in areas occupied by large dangerous animals such as elephants and hippos that take objection to having lights shone in their eyes. It may be necessary to sacrifice a lot of work to avoid encounters with such creatures and it is usually best to pick study sites that you know are not frequented by these animals.

If you have access to electricity or vehicles mega-bright, rechargeable torches are also an option. Unfortunately they are either too heavy to carry or else they hold little charge that they go out after about 30 minutes and require days of recharging. They are so bright that most smaller reptiles and amphibians will take cover from the beam and they are best suited to long range work such as crocodile surveys.

The phase of the moon and a qualitative measure of moonlight must be included in the environmental data collected for each site surveyed at night. Lunar phases are known to influence the activity patterns of many reptiles and amphibians.

3.5 Trapping methods

Setting traps that will catch animals for you can yield very exciting results and this is the only way many species in the community can be sampled. Simply searching for animals is very unlikely to reveal all the species present in their correct abundancies. There are few quantitative measurements of the differences between searching and trapping methods for reptile surveys. Fitch (1992) showed that trapping methods (funnel traps and artificial shelters) were more efficient than searching for all but the largest snakes in a community. Different trap types catch different species (or age classes) of reptiles and so a variety of search and trapping methods must be employed in order to get a representative sample of the community.

However traps must be designed and set with care and checked regularly otherwise some animals will almost certainly die.

Tips for minimising mortality during trapping:

1. check traps very regularly;
2. prevent trapped reptiles from predation (especially ants);
3. prevent trapped reptiles from desiccation and overheating;
4. try to design traps that do not catch unwanted species;
5. release animals at the exact point of capture.

When we have been working in large teams, I have found it essential to display a large board showing the rota for trap checking. With many different trapping sites being studied at one time it is easy for some traps to get overlooked for a day or so, by which time some of its occupants will have perished.

3.5.1 Theory of trapping

Passive trapping is a very energy efficient way of collecting large numbers of specimens, but surveys that rely solely on passive methods overlook many species. Many reptiles avoid traps, either because they are not interested in bait or because they are too wary. However trapping does allow effort to be reproducible, which is impossible in surveys that rely on individuals' ability to spot and catch animals. Comparison of trapping data between sites is only meaningful if all samples are subject to the same conditions and effort. Effort means how many traps are set and for how long. The conditions are the types of traps used, their orientation in space and the times of day at which they are checked at. Useful discussions of trapping theory can be found in Heyer *et al.* (1994) and Wilson *et al.* (1996).

It is difficult to produce densities from trap data. Usually there is no way of estimating the area that has been sampled by the trap. This is particularly true of baited traps, that might attract animals from different distances depending on the smell of the bait and how far it travels. They may attract animals into microhabitats they do not usually visit. Trap data is usually expressed most simply as a function of trapping effort or by a measurement of trap spacing. Traps also provide a useful measure of activity because inactive animals never get caught in traps. Thus by checking traps at defined intervals (e.g. every two hours) comparisons can be made between relative abundancies of different species with time of day or weather conditions.

As for quadrat and transect surveys, at least two replicate samples should be taken from each area. In each replicate (and in each habitat) effort and conditions are kept as similar as possible. Satisfying these conditions is very difficult and it is usually possible to think of many plausible reasons why trapping results will differ between samples. It is well worth the trouble of getting trapping effort and conditions as standardised as possible. This means keeping orientation of traps similar and paying close attention to local topography. It is obvious that there is conflict between the need to standardise methodology and use randomly selected sites.

I have classified trapping methods as non destructive (ND) or semi- lethal (SL). The latter always present some danger of mortality and should only be used where this is acceptable and justifiable.

3.5.2 Box, hoop and funnel traps (ND)

Sherman and other boxlike traps that work using a trigger-sprung door will catch a variety of lizards and the very occasional snake. They are available in a very wide range of sizes. Details of building extremely large traps can be found in Auffenberg (1981). One problem with opaque traps is that it's impossible to tell what sort of creature is inside before you open it. Shaking animals out of traps directly into sacks is the safest method against escapes or nasty surprises. Dean (1997) describes customised Sherman traps that allow contents to be viewed. Like all traps they must be positioned where animals cannot overheat. We usually bury traps so that only the entrance is exposed. They can be used on the ground or attached to tree branches at any height (note that Sherman traps will only worked when they are in a horizontal position). In all cases they work best when some attempt is made to disguise the trap by covering them with vegetation or other camouflage. Box traps are indiscriminate and tend to catch a lot of mammals. The number of traps used should be large enough so that no more than 20-25% of traps contain catches at any checking time. If this criterion is not fulfilled the traps become a limiting factor in the number of animals caught - animals are likely to visit previously sprung traps and escape as a result. Between sites traps should be cleaned thoroughly to reduce the risk of introducing novel pathogens.

Funnel traps are a classic way of collecting reptiles and one of the best ways of catching many snake species (Fitch 1951, Clark 1966). Commercially available traps tend to be both heavy and expensive and it is usually much more convenient to make your own. Feuer (1980) gives details of an inexpensive, double-mouthed hoop trap used to catch aquatic turtles (Fig 3.7)

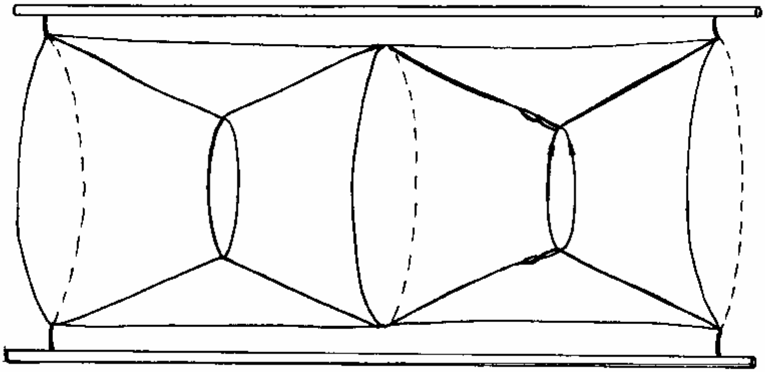


Fig 3.7. Hooptrap for catching turtles (after Feuer 1980)

Depending on the animals targeted, baits used in traps include meat, fish, fruit, peanut butter, eggs, rats, mice, snails tinned tomatoes or spaghetti. Traps can be rubbed with bait to enhance their appeal and disguise the smell of biologists. Durden *et al.* (1995) recommend baiting Sherman traps with two crickets tethered to cotton threads, one positioned near the entrance and another at the back of the trap. To catch males of some territorial species a rival male can be placed in, or close to, the trap, which will soon attract the indignant resident (Zani & Vitt 1995).

3.5.3 Sticky traps (ND/SL)

Glue or sticky traps have the potential to be a very useful tool for survey work (Bauer and Sadlier 1992; Rhodda *et al.* 1993; Whiting, 1998). Many forest based people will know of sticky resins that can be used to trap small animals, but the best approach may be to use commercial traps. Non toxic glue traps such as Catchmaster work very well and are available in a variety of sizes. The trap must be tied down and vegetable oil is used to remove glue from captured animals (either rubbed over the animals or by putting the gluey animal in a plastic bag with a film of vegetable oil). Sticky traps can be baited and used in the same way as spring door traps or they can be positioned at strategic sites such as burrow or tree hole entrances or along activity corridors. The great advantage of sticky traps is that they can be placed almost anywhere. Their main disadvantage is that they catch animals indiscriminately and traps must be checked very regularly to prevent animals becoming covered in glue. Insects that land on the trap may provide additional incentive for lizards to visit. There are no published data for mortality using sticky traps but they are unsuitable for non-destructive sampling of geckoes and other delicate reptiles. The adhesiveness of the trap declines with age and with wet weather. Zani & Vitt (1995) found that sticky traps had no adverse effects on the lizards they caught but note than many lizards will simply jump over glue boards.

Another, gentler, method for catching small lizards is described by Downes and Borges (1998) which uses strips of 50mm wide sellotape, rendered less adhesive by first sticking it to a piece of cotton, which is left sticky-side up to catch small (1.5g) skinks. The traps are checked more or less constantly and animals caught peeled off the tape head first with no ill effects. These traps can be baited, put along drift fences or left in strategic positions. The authors report success catching lizards of up to 40g although it is not clear how long larger animals would remain caught in the trap.

3.5.4. Drift fences and pitfall traps (ND)

Drift fences guide animals in the direction you want them to go, usually towards a trap. They can be used in conjunction with many types of traps including glue, funnel, pitfall and box types.

Ideally fences will be of a material that animals cannot climb over, but they must also be flexible enough to follow less than straight trenches, light enough to be carried in the field and inexpensive enough to be affordable. I much prefer rigid materials such as light plastic sheeting that do not require posts to support them and so consider fabric fences to be a waste of time unless they can be fixed to a heavy line stretched above the fence. Any obstacle along the drift fence will make it more likely that animals will wander away from the fence before they reach the trap. Good drift fence materials include light metal sheeting, rigid window screening and corrugated plastic sheeting (available from gardening shops). Enge (1997) advocates the use of silt fencing (used on building sites to control sediment run off) that allows water to pass through for sampling along streams and marshy areas. Vogt (1987) suggests using plastic window screening strung between two sets of pulleys to allow drift fences and funnel traps to be set between trees in the canopy, allowing the capture of snakes, lizards, frogs and salamanders.

Pitfall traps work in the following way. Animals moving along or under the ground encounter a fence and walk along it until they plunge into a cunningly hidden bucket from which they are unable to escape. Many people try pitfall traps once and are not impressed by them, but if they are set up correctly they work very well. Attention to detail is all important with pitfall traps. The buckets must be completely sunk into the ground. If small animals have to cross the rim they will hesitate. The fence must be buried throughout its length or animals will crawl underneath it and avoid the buckets and the ground on both sides of the fence should be clear of obstructions and swept free of debris. Most animals are caught when traps are laid at the borders of different vegetation types.

Figure 3.8. Pitfall line set in spinifex desert, Western Australia.

First pick an area and decide how to layout the traps. Give each bucket an easily seen, indelible number and dig holes and sink buckets. Then dig the trench for the drift fence. It need be only deep enough to support the fence, but deeper fences will traps many subterranean species such as blindsnakes and legless skinks. The critical part of the fence is that leading into the bucket. The fence should overlap the rim without affording any gaps that animals can sneak through. Because pitfall traps require little maintenance and they can be checked with a minimum of effort it makes sense to install as many as possible early in the project. This allows the use of replicates and makes achieving good sample sizes more likely. There is no denying that making pitfall lines is hard work. There is a lot to be said for hiring local assistance for some of the work. Farmers tend to be better at digging holes than students and it provides a way of involving local people in the project who might otherwise not benefit from your visit.

Three main disadvantages of pitfall traps are that some animals (especially frogs) can climb out of them, they are unsuitable for areas where it is difficult to dig deep holes, they get waterlogged in heavy rain and animals in pitfall traps are easy prey for predators. All these problems are neatly solved by the side flap pail trap (Nadorozny & Barr 1997). The bucket is sealed and animals travelling along the drift fence enter via a hinged, one-way door in the side of the bucket.

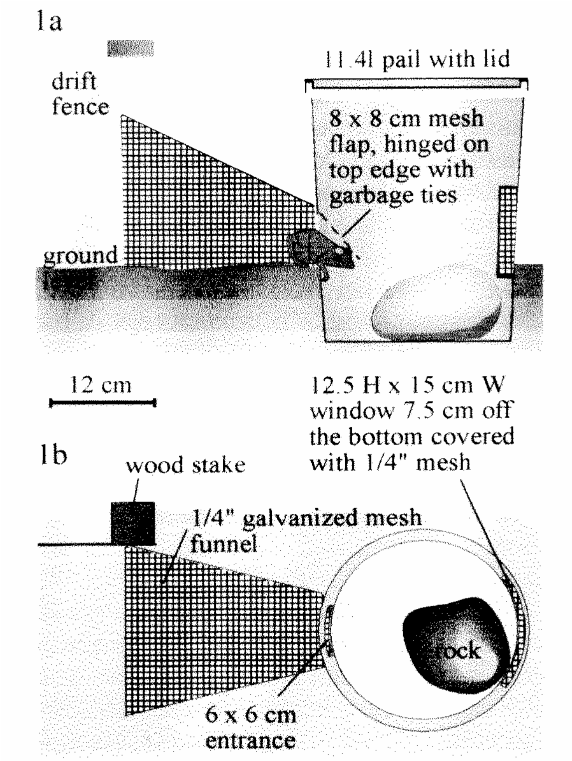


Figure 3.9. Side-Flap Pail Trap showing a cross-sectional side view (1a) and top view with lid removed (1b) (from Nadorozny & Barr, 1997, Herpetological Review 28(4):193, reproduced with permission)

Daoust (1991) recommends putting a piece of sponge soaked in water in each trap to prevent animals from dehydrating. This has definite advantages over simply putting water in traps because when small mammals (primarily insectivores) drop into "wet" traps they are much more likely to fall victim to hypothermia.

3.5.5 Refuge and pipe traps (ND)

Leaving items lying around that reptiles will shelter in or under is a very sneaky way of surveying shy and elusive species. Materials that can be used include tin sheets, old carpets and roofing felt (e.g. Riddell 1996). Moulton *et al.* (1996) describe the use of hollow poles (PVC pipes) that attract tree frogs. Here the diameter of the pipe seems to be crucial, with 2cm piping giving better results than larger sizes.

Pipe traps consist of a container (most easily made from window screening stapled together to form a cylinder) that is accessed via a narrower pipe (Lohoefener & Wolfe 1984). Animals enter the pipe and once they are in the main body of the trap they are unable to escape. These traps can be buried with the pipe flush to the ground or set along drift fences. Black coloured pipes are said to give best results.

Refuge and pipe traps may work best during hot weather and in areas where there are few natural shelters. They are a useful way of finding species for inventory work or other purposes but because they provide an important resource they are obviously not suitable for looking at aspects such as densities or microhabitat use.

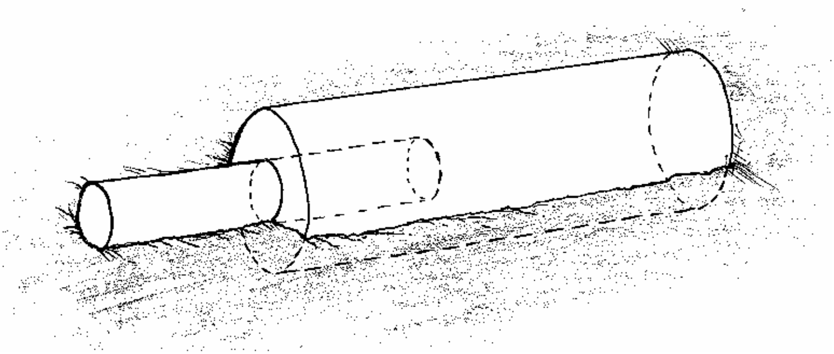


Figure 3.10 Pipe trap, after Lohoefener & Wolfe 1984).

3.5.6 Noose traps (ND/SL)

Noose traps work either by snaring animals as they walk past or by tempting animals onto a path that contains nooses. Some are simply snares that entangle animals, usually by the neck, and get tighter the more the victim struggles. My experience of these traps is that they catch a wide variety of animals, but rarely the right ones and sometimes with tragic consequences. More sophisticated noose traps are designed to snare animals by a single limb and are constructed to exclude as many unwanted species as possible. In theory noose traps can be used to catch animals of any size. In practise I have never been able to make sensitive enough triggers to catch lizards under about 50g.

The noose traps we have had best luck with are spring loaded. A trigger is pressed by the limb that is to be snared, releasing a bent-over sapling and tightening the noose (figure 3.11). To exclude unwanted prey such as cats and monkeys the bait is approached through a low tunnel. The trap should be set so that the noose is pulled tight, but not tight enough to pull the animal into the air and left to dangle, unless it is small, in which case it will be less vulnerable to attacks by conspecifics. Be very careful working around and setting up spring loaded traps. Triggers get sprung accidentally and fingers and eyes are at risk.

Sprung noose traps are a good choice in areas where it is difficult to move around; swamplands and thick forest for example, where actively searching for lizards is very difficult. They would also be good traps to use in swampland, but care must be taken to trap only above the high tide mark. Alternatively, traps can be set on rafts floating on the water. I have used noose traps to catch lizards living at low densities on steep slopes. Traps have to be spread over a large area and the terrain and distances involved make it impossible to check traps more often than once per day. Although many of the lizards noosed escape, those that remained were all released undamaged.

There are many variations on noose traps all over the world, but none of them are easily learned from a book. It is much more instructive and entertaining to learn the method from local people, but you should insist that they target animals' limbs rather than their necks. Otherwise, do not despair if early attempts fail, with practise techniques will be refined and eventually animals will be caught. Noose traps need to be used carefully and it takes time and practise to set them up correctly. They are also potentially dangerous and should only be used by people with responsible attitudes. I once heard tell that a zoology student, well versed in SAS survival literature, once tried to catch a wild pig with a noose trap and instead caught a very young shepherdess who luckily escaped uninjured. More than can be said of the zoology student after members of the child's family caught up with him.

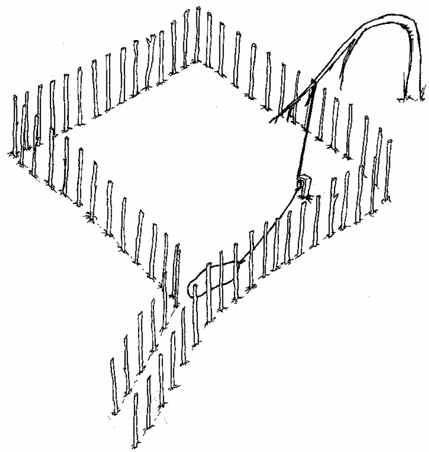


Figure 3.11 Noose trap.

3.5.7 Camera traps (ND)

Cameras triggered by a pressure switch will photograph animals emerging from or entering burrows (Guyer *et al.* 1997). The device costs around £110 assuming you can assemble the components yourself.

3.6 Surveying aquatic reptiles

3.6.1 Marine turtles

After birth, marine turtles spend all their lives in the sea except for visits to beaches by females to lay eggs. This makes them very difficult to survey meaningfully. The only practical way to do it is to try to estimate the how many mature females are present by surveying breeding sites at the right time of year. This is best done by walking along the water’s edge early in the morning, looking for tracks leading to nesting sites. Caution is needed because over half of the females may emerge more than once before laying eggs (Servan, 1975), and females of some species may lay up to four clutches of eggs in a year. Counting only tracks that end in nests is one way of reducing the error of the track count method, if it can be done without disturbing nesting females or their eggs. Students working independently should not attempt to tag turtles unless they have the approval and co-operation of a long term programme. Marking studies find very few recaptures, prompting suggestions that mortality among adults is very high or that females only breed every 2-5 years. If the latter is the case, estimates of seasonal nesting totals must be based on more than one season's surveying (Maylan, 1995; Hughes, 1995).

Figure 3.12 A turtle nest

One thing that is certain is that turtles nesting sites are dwindling world-wide. Development and other human activities are responsible for this. It has been calculated that nearly half a million eggs per year are required to maintain a stable population of less than 1,300 female loggerheaded turtles (Crouse *et al.* 1987). Turtle beaches are very sensitive to disturbance and keeping this to a minimum must be the first priority of any survey. Perhaps the most important rule is not to use torches around turtles. Eat plenty of carrots and avoid discotheques and within a few days you will be able to see like a cat. If you suspect you are close to a turtle turn your torch off, crawl along the beach and maintain absolute silence. In this way you should be able to approach females close enough to hear them breathing, but under no account should you touch them or otherwise disturb them in any way.

Surveys of predators of turtles nests and the hatching rates of babies can yield very useful data. Ten days after the last hatchlings have emerged (not before) the nest can be excavated and the hatching success rate can be determined. Record the total number of eggs and the numbers that have hatched, been destroyed, are infertile or contain dead/deformed hatchlings.

3.6.2 Freshwater turtles and tortoise

The comical tortoises are the easiest reptiles to catch but they can be fiendishly difficult to find. Usually their burrows are easier to find than the animals themselves. Bryan *et al.* (1991) describe the use of a trap set just outside the burrow entrance that will catch tortoise as they emerge. Woodbury & Hardy (1948) used a metal pipe with a hook attached to pull tortoise out of their burrows. Tapping the shell three or four times with a stick may induce some tortoises to leave their burrows. The holed-up animal is given a few gentle taps and the researcher retreats from the burrow entrance. After a few minutes the tortoise emerges and can be grabbed. Medica *et al.*(1986) report an 82% success rate using this method with *Gopherus agassizzii*. Possibly this daft reaction occurs because the tortoise associates the tapping with another (rival) animal. According to Medica *et al.*, sometimes tapping one tortoise causes several to leave the same burrow! This method is said to work best on warm animals and should therefore be employed only during the day or early evening.

At night and on cloudy days sleeping turtles can be observed or caught from boats. For capture a long handled net works very well. Turtles are much less wary when they are underwater and when visibility is good many animals can be netted in this way. A number of turtle traps have been devised, the most basic being a pivoted ramp over a cage (Legler, 1966). The turtle crawls up the ramp, attracted by bait, and tips into the trap. Feuer (1980) describes the construction of a lightweight hoop trap for catching turtles whilst Graham (1995) gives details of a D-shaped funnel trap. Traps are partially submerged in water and have bait inside. When turtles are not hungry (e.g. during cold weather) they can be lured with ingenious plaster turtles, painted to look like the targeted species. The decoy is placed in the trap which attracts turtles of both sexes (Plummer, 1977; Mansfield *et al.* 1998). Commercially made turtle traps are available (see appendix) but they will probably be too heavy and expensive for expedition use. Graham & George (1996) describe a method of keeping collapsible funnel traps open in deep water to catch turtles. They recommend that traps set without a "breathing space" should be checked at least every twenty minutes.

It may be possible to age tortoises of some species by counting the scute rings on the shell which are laid down yearly (Germano, 1988).

3.6.3 Crocodilians

Crocodilians are not a specious group and most can be identified on sight after a visit to a museum collection. Like goannas, crocodiles are extensively hunted for their flesh and skins and little is known of population sizes in most areas. Many crocodiles are thought to be endangered through habitat destruction and other human activities. Crocodiles present the dual challenge of being shy, amphibious creatures that are difficult to find on one hand, but also the only reptiles that might consider biologists good to eat.

Counting basking crocodiles during the day might be useful if you can find enough basking sites, but most crocodiles are elusive and surveys are best carried out at night from boats. Casting a powerful torchbeam (ideally 1,000,000 candlepower powered by a 12V car battery) over water at night reveals their orange/red eye shine which allows counts to be made. If animals can be approached closely enough their size and age can be estimated, otherwise they are recorded as "eyes only". The proportion of these animals in the population might indicate levels of predation by people. Replication (i.e. counting the same animal more than once) can a problem unless surveys are conducted quickly or individual animals can be distinguished. Spotlight counts are usually conducted during falling tides in estuarine areas or during the dry season elsewhere because this allows greatest visibility. Bayliss (1987) lists the main sources of visibility bias as being the density of vegetation, the width of the river, number of bends in the river and the position of the crocodiles and their wariness of mankind. To minimise the effect of immigration and emigration from the study area Bayliss recommends that observers with spotlights are positioned at either end of the study area to monitor movements. Because the numbers seen can vary enormously from night to night plenty of replicate samples and robust variance calculations are required (Bayliss, 1987; King *et al.* 1994). As well as the climatic data listed in Section 2, water depth and salinity should be recorded for crocodile surveys.

Crocodile surveys tend to concentrate on main river courses and neglect tributaries and swamps which are more difficult to travel through but tend to have the highest densities of animals. If you are going to survey away from the main river course it is essential that you become acquainted with the route during daylight first. It's very easy to get hopelessly lost! Whenever possible you should employ local people to man boats for surveys.

Recently much attention has been paid to aerial survey methods for crocodilians, but this is rarely feasible for expedition projects (e.g. Mourao *et al.* 1994). In the breeding season nest counts are used to determine the abundance of sexually mature females in the population (Hall 1991). This may be the only way to survey very shy species, but beware of getting too close to nests, which will guarded ferociously.

Mark recapture studies on crocodilians are possible, The usual method of marking is with a barbed number tag that is attached to animal's backs via a long stick, which makes it unnecessary to catch them.

Where plenty of footprints occur it may be possible to determine the size of crocodiles by measuring hind foot lengths (Wilkinson and Rice, 1996). Other methods for ageing crocodiles (e.g. Hutton, 1986) are beyond the scope of short, non intrusive studies. Details of trapping and restraining methods for crocodilians are given by Murphy & Fendley (1975), Mazzotti & Brandt (1988) and Jones & Hayes-Odum (1994) but such measures should not be considered unless you have very good reasons for catching animals and experienced help.

Obviously, large crocodilians are very dangerous animals and the normal precautions should be taken. Don't approach crocodiles any closer than is necessary and don't fall out of the boat!

3.7 Survey methods for amphibians

The damp skin, larval stage and communal breeding behaviour of most amphibians make them rather different subjects for fieldwork than reptiles. Detailed discussions of techniques can be found in Heyer *et al.* (1994) and Olson *et al.* (1997).

Amphibians tend to show highly clumped distribution during the breeding season and very dispersed patterns for the rest of the year. For survey purposes this has mixed blessings. Although breeding sites will contain large numbers of adult and larval amphibians, it is unlikely that the whole amphibian community will be breeding at one time or in one place and so targeting only obvious breeding sites will mean that some species are excluded from the survey. Breeding sites are not always obvious, and the breeding sites of many species are not aquatic. Away from breeding sites densities might be very low, making controlled surveying difficult. In general we use transects away from breeding congregations and in linear habitats (such as long the banks of lakes and pond). Around very densely populated breeding sites quadrats are usually suitable for sampling. High sided $1m^2$ quadrats are ideal for surveying small fast moving frogs such as microhylids (Figure 13.3). It is impossible to count frogs high in trees and the best that can be done is to estimate numbers from calls. A stick is helpful to flush animals out of dense or prickly undergrowth.

There are no set rules for how to find amphibians - but meticulous searching, luck and patience definitely help. The majority of amphibians are nocturnal and are therefore most active between dusk and midnight. A headtorch is essential allowing you free hands to capture specimens and make your way through difficult terrain. A halogen bulb isn’t necessary for amphibian searching on the ground and will run batteries down very quickly. A halogen bulb does allow you to scan higher in the trees and it is surprising how many frogs can be seen in a good torch beam.

3.7.1 Capturing and handling frogs

Frogs caught in the glare of a headtorch can often be picked off leaves and branches and rarely attempt to escape. Resist the temptation to run in and try to grab the animal. Move carefully and slowly. Keep your eyes (and your headlamp) fixed on the creature, approach it in such a way so as not to cast shadows and, if possible, try to block off any easy escapes into thick vegetation. It’s always easier to grab animals with both hands, but one hand is usually quicker. Try to get the hand(s) over the whole body and head. Without hesitating, simply flatten the animal against the ground. It can then be scooped up along with the accompanying vegetation

Large frogs are quite easy to grab but very difficult to hold on to. Use both hands and encircle the head and body leaving the hindlimbs free. If you restrain them the frog will gain purchase to jump out of your grip. Nets can also be used to catch large frogs out of water (small ones can crawl underneath). Catching frogs in the water is more difficult. It is not easy to grab them and usually a net will give best results. Most frogs turn and dive when they feel trouble coming, so best results are obtained by trying to slip the net under the frog. As soon as the frog has entered pull the net out of the water and flip it over to seal the mouth. The best nets to use are heavy duty sweep nets.

When working at the water's edge - DO NOT OVERREACH. Drowning is a major cause of death on student expeditions. It is wise to always visit amphibian sites during the day to determine water depths and which areas are particularly soft before starting field work.

Figure 13.3 High sided 1m^2 quadrat suitable for sampling small frogs around breeding sites

A number of the trapping methods described for reptiles work well for amphibians. Richter (1995) describes a simple funnel trap made out of two plastic drink bottle that is suitable for catching both adult and larval amphibians. If the trap is large enough to catch adults it must include a sufficiently large air pocket or the animals will suffocate. Pitfall traps (section 3.5.4) catch many species, but unless the buckets are covered most will simply jump out again. Murphy (1993) describes a modified drift fence that caught over 90% of the male treefrogs at a breeding site over 23 days. The animals climb the fence but are unable to cross the overhang and stay in the top crevice, where they are easily collected. Dodd (1991) quantifies the sampling bias of an amphibian community in Florida using drift fences.

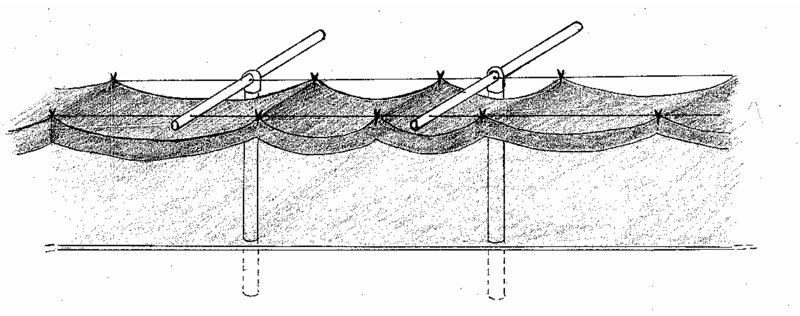


Figure 13.14. Modified drift fence for catching treefrogs after Murphy (1993)

Amphibians are kind to the biologist in that they often advertise their presence by calling. Don't ignore this - some species are impossible to find unless they are tracked down by their call, although this often requires a great deal of patience. Species can be overlooked if you don't make an effort to track down every type of call that is heard. If you can identify species by their call you can save a lot of time. Obviously you must be certain of the frog call - many insects are almost perfect mimics. Searching for frogs on the basis of their calls will greatly skew the apparent sex ratio and this should be recognised. I do not believe it is possible to measure densities from frog calls, but they can be useful for comparisons of numbers of frogs in different areas.

3.7.2 Recording amphibians

An important technique for studying amphibians is recording calls. Sound recordings act as a useful identification guide and they can help you familiarise yourself with the amphibian species of an area. Recording calls rapidly improves your skills in searching for vocal amphibians and will teach you a great deal about amphibian behaviour. Recordings provide a valuable permanent record that can be used for taxonomic research and acoustic behavioural research. Recording calls can be reliable evidence of species occurrence, even if the animals cannot be found. Amphibians that on first examination appear to be the same species sometimes have different calls and may therefore warrant treatment as different species. Recording itself is a very simple exercise. All that is needed is patience and the ability to stand in the same position for unduly long periods (and suffer the subsequent cramps and pins and needles) attempting to keep your microphone from wobbling while an amphibian refuses to call. Finding calling frogs requires great patience but it is always worthwhile in the end. Once a call is located most frogs can be approached to within a metre although your movement will undoubtedly quieten the animal for a while. It is often necessary to turn off your head torch whilst approaching the animal and recording its call.

Essential data that must accompany recordings are species, date and time, habitat, the temperature (water temperature as well as air if applicable) and details of any background noises. If an amphibian cannot be identified to species level the number of the voucher specimen should be given so that the call can be identified at a later date.

Recording equipment is simple and no expensive accessories are required. A simple, well made and robust portable cassette recorder such as a Sony Walkman “professional” model can be used (approximately £200). DAT recorders are likely to be damaged in the humid and swampy conditions that are favoured by amphibians. Short gun mikes are ideal for recording amphibians because they can be approached so closely. A good microphone such as a Sennheiser is likely to be the most expensive item of equipment (a shock mount is also necessary to prevent handling noise with such a good quality recorder). Recommended cheaper microphones are AKG D190 (£140) or Beyer M69 (£160). Connecting the microphone to the recorder can be a problem if the microphone is mono but the recorder is stereo, but with the right components a suitable lead can easily be soldered together. It is advisable to take a soldering iron and duplicate components so you can resolder any joints that get damaged or solder new connecting leads. These can usually be bought cheaply in the country of the expedition, but always bring any specialised components from home. Headphones have a tendency to get damaged at the joints so take a couple of pairs and some extra plugs. Take as many precautions as possible to avoid getting the equipment wet. Store equipment in silica gel when not in use. We routinely seal all cable connections with epoxy resin as a precaution against moisture. For certain frog calls, high frequencies may distort the recording therefore recording levels need to be set to counteract this. The ideal setting for recording varies from animal to animal and this is best learned from trial and error. Do not use Dolby, dbx noise reduction systems, peak limiters or automatic volume controls because they are not suitable for animal sounds. Connecting a set of speakers and using playback of previously recorded frog calls might be a very good way of attracting more frogs of that species. Small plug in units can be bought inexpensively. The batteries for some items of equipment (e.g. microphones) are impossible to obtain in other countries so they must be bought before departure.

For more advice on equipment and recording techniques contact the British Library of Wildlife Sounds (address in appendix). They hold training sessions for those wishing to learn more about wildlife recording techniques and may also provide tapes or even loan equipment.

3.7.3 Identifying larval amphibians

Uniquely amongst the air breathing vertebrates, amphibians have a free living larval stage. Visual identification of tadpoles is very difficult and the vast majority of tropical larvae are not well characterised. Well organised expedition teams working during the breeding season can make a significant contribution to knowledge of larvae with very little effort by collecting tadpoles and growing them to adulthood, taking specimens at each stage. In this way, over a couple of months the larvae of many of the amphibians in the community can be identified. If the duration of the field project is shorter than this it is still possible to collect the information by collecting pairs in amplexus and raising their spawn in captivity. In this way even if the tadpoles are not raised to maturity their identification is possible.

3.7.4 Raising tadpoles

I leave amplexus pairs collected in the wild in a large lightproof bucket which contains water, wet leaf litter and branches. Normally eggs are produced on the night of capture but after a maximum of two night the animals are released and the eggs left where they were laid until they hatch. Then I transfer up to six tadpoles into shallow plastic bowls about 15cm diameter with identifying numbers written in permanent marker. They are fed fish food (pellets or flakes) and the few species that do not accept this diet are given pieces of brittle-leafed water plants. Water is changed every day and the tadpoles kept out of direct sunlight. Note that although tadpoles will grow quicker under warmer conditions care must be taken not to let them overheat. The entire collection can be killed within minutes.

Every few days a single specimen of each species is preserved in ethanol. When the back limbs are half developed I cover the containers and give the froglets some land to climb on. By the time their front limbs have appeared and their tails are shrinking they can usually be identified and the excess animals released.

Expedition members with the necessary enthusiasm can draw and describe the morphology and metamorphosis of the tadpoles (Duellman and

Treub, 1994). Under magnification the mouthparts can be described. Otherwise the carefully labelled collection should be deposited with the rest of the collection.

3.7.5 Tadpoles in survey work

You should not use tadpole abundance or densities to make estimates of the relative abundance of adults in an area. The variable and vast numbers of eggs produced and very high mortality mean there is unlikely to be any relation between the diversity of larval amphibians and that of the adults. Similarly newly metamorphosed animals should not usually be counted in census work, which should target only sexually mature individuals.

Figure 13.15: Raising spawn from pairs of frogs found in amplexus provides a certain method of tadpole identification.

3.7.6 Marking amphibians

Because amphibians are delicate and have extremely sensitive skin marking them can be problematic (Ferner 1979). Most of my experience is with methods that do not work. PIT tags (Section 2.14) provide the least harmful method of marking amphibians, but the cost puts them beyond the reach of most projects, especially where large numbers of animals have to be marked.

Rubberbands

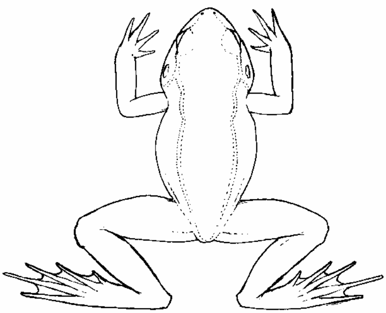
In theory tying rubber bands of different colours around frogs' limbs or waists provides a good short term marking system (Emlen 1968). The band is tight enough to stay on and not get snagged in vegetation, but not so tight as to injure the animals or influence its behaviour. In my experience frogs marked with rubberbands and kept in captivity seem to do all right, but wild frogs disappear from the study site immediately. Those that are subsequently recaught (very few) have injuries where the bands have rubbed on other parts of the body.

Fluorescent dye

Non toxic dyes have been used to mark many salamanders (e.g. Ireland, 1991) and in some cases have been showed to give higher recaptures than toe clipping (Nishikawa and Service, 1988). The method uses compressed air or nitrogen to blast particles of dye into the skin. The dye can be seen easily at night with a UV light and is also visible during the day. However for frogs Schlaepfer (1998) considered fluorescent marking to be inferior to toe clipping on the grounds of convenience, ease of use, cost and harmfulness.

Toe clipping

Toe clipping involves cutting bits of toes off in a systematic way that enables positive identification of individuals thereafter (Donnelly *et al.* 1994). The operation is usually performed with a very clean pair of surgical scissors. Objections to this method include the pain and subsequent danger of infection inflicted on animals and that important behaviours (running, jumping, climbing, digging) will be adversely effected. Although this method has been reported to increase mortality or alter behaviour in some species (Clark, 1972; Golay and Durrer, 1994), other studies have found no ill effects (Lemckert, 1996).



Capture and handling techniques

Pitfall traps can be considered for amphibians surveys, but in general traps are not much use for amphibians and most work must be done by hand and eye. Handle frogs as little and as gently as possible, using wet hands and beware of spreading diseases between amphibian populations. Recently transmission of fungal diseases through populations by researchers has been cited as a possible cause of decline in some amphibians communities. Keeping all equipment as clean as possible during field work, and sterilising it between study sites will reduce the risk considerably. The Declining Amphibian Population Taskforce Code of Practise is given in the appendix.

3.7.7 Anaesthetising amphibians

MS222 is a completely reversible anaesthetic for both fish and amphibians. It is used to make animals easier to handle and less susceptible to injury during examination and measurements. It is also a good way of getting very nervous specimens to pose for photographs (although good photographers reject this method unless everything else fails because of the accompanying changes in colour). Three stages of anaesthesia are distinguished, the most extreme being the surgical stage, at which animals turned upside down are unable to right themselves. This level of anaesthesia typically lasts 30-40 minutes and is followed by full recovery after 1-2 hours.

MS222 is a suitable anaesthetic for amphibians, but not for reptiles. It is sold as a white powder and 3-4g are dissolved in 1 litre of water. MS222 solution breaks down and becomes ineffective when exposed to light so it should be stored in a light-proof bottle. The anaesthetic should be stored in a wide-mouthed container that allows animals to be removed easily. Frogs are dropped into the solution and left for 30-300 seconds depending partly upon their size. Some species (particularly Microhylids) appear resistant to MS222. Initially it is best to check animals in the solution every 30 seconds to determine the level of anaesthesia.

Recovery is effected by placing the animal in a shallow layer of fresh water. Recovery is rapid and usually takes less than 20 minutes. The biggest risk with aqueous anaesthetics is that they will enter the lungs and drown specimens. I have found that adding a small float to the container allows animals to keep their nostrils clear of the liquid.

If the solution is kept cool and dark it will last for at least several months. However animals tend to defecate in response to anaesthesia and a fresh batch should be made up at least weekly or whenever the study site is changed. This will reduce the risk of disease transmission between populations.

3.7.8 Transporting and keeping amphibians

Newly caught amphibians are best transported in self-seal bags inflated with a little air. Usually only one animal is put into each bag. Opinions differ as to whether or not water should be added to the bag, with some sources maintaining that evaporative loss from the skin is sufficient to maintain humidity, but I always add just enough water to the bag to keep the skin moist and not enough for a traumatised animal to drown in. The only drawback of adding water to the bag is that it complicates weighing. The bags I use weigh 1.5 and 3.0g. The weight of a frog in a dry bag is therefore easy to calculate, but if water has been added the frog has to be transferred to a dry bag before it is weighed. During general fieldwork it is not practical to add a known amount of water to the bag (i.e. 1ml of water weighs 1g), or to weigh the animal immediately after capture. Thus two bags are used for each animal; a wet one to transport it in and a dry bag to weigh it. There is a possibility that the weight of the frog will change between capture and subsequent weighing. If this is likely to have important implications for your experiments the animals should be weighed immediately after capture, but the method is safe for general recording purposes.

The other threats to amphibians in bags are that they might overheat, be stepped on, or lie, forgotten, somewhere in the field station. Using polystyrene cool boxes to carry bagged frogs should be considered if there is a risk that the animals will be exposed to strong sunlight. In unexpected situations I use a damp flour sack (50lbs capacity) to keep bagged frogs cool. Note that this will not work for reptiles, which will suffocate in wet bags.

I have found that mortality rates are lowest when frogs are released within 12 hours of capture. Of course they should always be released exactly where they were caught. For examination of live specimens, deep trays are best. Particularly frisky individuals or those that require minute inspection should always be anaesthetised prior to examination.

Section 4

SHORT NOTES ON SOME REPTILES, HABITATS AND EQUIPMENT

This list does not cover all types of lizards and snakes, but just the ones I have had (enjoyable) experiences with.

4.1 Lizards

4.1.1 Geckoes

The loveliest of the lizards, the big eyed geckoes are generally nocturnal and best collected by torchlight searching. The eyes of some species shine bright red in the beam of a torch and many geckoes will remain frozen in the light and can be caught by hand. Geckoes should be picked up gently at the back of the head. They are delicate creatures and only the largest species are capable of inflicting a nasty bite. Not only are the tails fragile, but some species shed the skin from their bodies in the same way. One worker reports recurrent nightmares after seizing a gecko that wriggled out of his hand red raw naked, leaving him clenching the warm skin. Terrestrial geckoes often wander into pitfall traps. For tree dwelling species box or post traps or lines baited with fluttering prey may work. About 900 species of geckoes are found throughout the world.

4.1.2 Agamids

Agamids, or chisel-toothed lizards, occur throughout Africa, southern Asia and Australia, but not Madagascar. There are over 300 species and they tend to be ground dwelling or arboreal. The vast majority are active during the day. At night many arboreal species can be simply picked off branches as they sleep. Agamids tend to be very fast moving animals that rarely get caught in pitfall traps. Only the smallest species move slowly enough to be caught by hand. Most species can be caught with a noose of dental floss on

the end of a pole. Sometimes the fleeing animal must be chased into a refuge and then gently extracted. Agamids are the most common lizards seen during the day around human habitations. The males, who are often brighter than the females, defend territories and do a lot of head bobbing. Therefore an animal that escapes is likely to be found at the same spot on subsequent occasions. It is a good idea to practise noosing techniques on urban lizards before going into the field.

Figure 4.1: Ctenophorus inermis, a typically alert agamid.

Figure 4.2: A few agamids rely on camouflage rather then speed. This Australian Moloch is very difficult to spot among grass tussocks.

4.1.3 Skinks

Up to 1500 species of skink occur throughout the tropics and subtropics. They are primarily diurnal but a significant number are active during the night. Ground dwelling skinks often fall into pitfall traps, but their smooth scalation, short legs and small heads make them difficult, but not impossible to noose. Skinks occur everywhere, under the ground and in the tops of trees. They have very delicate tails which they will shed at the first opportunity. Although tail shedding is "harmless to the lizard" it is likely to result in reduced survival and fitness because of the energetic demands of growing a new tail and the increased danger of predation before the tail tip regenerates. For this reason it is important to grab skins as close to the head as possible. Small skinks can suffer heat stress just from being hand held for short periods, it often takes rather longer periods to key them out. Because skinks are a very speciose group they require careful identification, often by minute examination of very tiny scales. Many very similar species often occur in the same area and new species are always being discovered. Unless good keys exist or the skink community is very simple it may be necessary to take voucher specimens to get identifications correct. Sometimes this can be predicted prior to arrival in the field by finding the available literature and trying to identify museum specimens with it. If you are not going to take specimens high resolution photographs are a good alternative. It is a very good idea to take DNA samples of skinks in diverse communities.

4.1.4 Lacertids

Lacertids are small, diurnal lizards found in Europe, Africa and Asia. They can usually be caught with the methods described above for skinks.

4.1.5 Iguanids

Smaller iguanids can be treated like agamids. Iguanas are particularly foolish lizards, but extremely handsome. Larger specimens use the tail as a whip in defence and a have a powerful bite. Some can be attracted by meat or fruit bait and many can be noosed as they lie in trees.

4.1.6 Goannas

Goannas, or monitor lizards, occur in Africa, Asia and Australia. They are always the largest lizards wherever they occur. The family includes the Komodo dragon and at least 50 other species. In Australia and eastern Indonesia dwarf monitor lizards of 20-100cm occur.

In Africa and all parts of Asia except Indonesia monitor lizards are not a speciose group and can be identified on sight. This is fortunate, because they are strong lizards with powerful tails and very long and sharp teeth and claws. The safest way to catch them is with a noose and great caution. The only monitor lizards I have ever caught in pitfall traps are newly born ones. Some species can be attracted to baited noose traps. Goannas throughout

Africa and Asia are extensively hunted for their meat and skins. The skin trade accounts for at least 3 million skins per year and perhaps 30 million goannas are consumed annually. Despite this virtually nothing is known of the effects of exploitation on populations anywhere in the world. Estimates of population sizes in different areas are urgently needed (King and Green, 1993; Bennett, 1998).

4.1.7 Chameleons

The wonderful chameleons live in Africa, Madagascar and India. About 130 species are known. They are difficult to find, especially during the day when their cryptic colouration is most effective, but once located they are very easy to catch. Just grab the animal behind its head and unwrap the claws and tail from the branch. Many people are afraid of chameleons, but they are completely harmless. Chameleons are usually seen in bushes or trees, but some very small species live in leaf litter (Grismer, 1988).

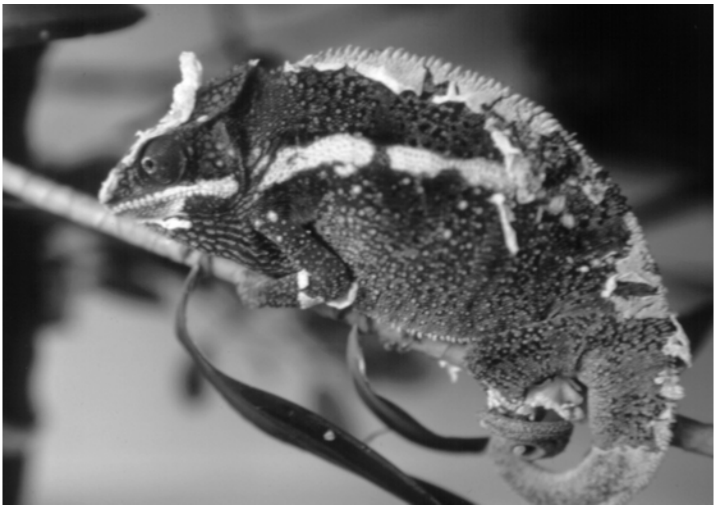


Figure 4.3. Chameleons grip branches very tightly using five limbs.

4.1.8 Pygopodids

A group of about 40 legless lizards that live in Australia and New Guinea. Very secretive and usually encountered during night-time searches. They are very difficult to catch. Searching leaf litter or lose sand by gentle raking may reveal specimens during the day which might be easier to catch (Kluge, 1974).

4.1.9 Teiids and heloderms

Large New World lizards handled in the same way as monitor lizards. The heloderms are slow moving animals, instantly recognisable. Their bite is venomous and their slow movements disguise great strength. Teiids are heavily exploited for meat and skins (Bogart and del Campo, 1956).

Figure 4.4. Heloderma suspectum – the gila monster is often persecuted by people.

4.1.10 Cordylids

About 75 species of plated lizards occur in Africa. They are not a well known group. Despite their robust appearance their tail tips are very delicate (Lang, 1990; 1991). I have only found them in burrows.

4.2 Small legless burrowers

This is not a real taxonomic group. It includes legless skinks such as the Australian Lerista, and the worm-like snakes Typhlops, Leptotyphlops and Uropeltids. These animals are rarely encountered unless you use pitfall traps or do a lot of very fast digging. Often they turn out to be the most abundant reptiles at a site, but they all tend to look the same and their taxonomy is a bit of a nightmare. It may not be worth collecting specimens of these groups unless you have a good key or know someone who is able (and willing) to identify them for you.

Figure 4.5 Plated lizards occur only in Africa

4.3 Snakes

People make a lot of fuss about snakes, but I have found that unless you go looking specifically for snakes you are unlikely to encounter them very often. Often when you go looking for snakes you find very few as well. If you have a surprise encounter with an unknown snake you don't like simply keep still, shut your eyes and count to thirty. If it hasn't disappeared back away from it slowly. There is very little possibility that you will encounter aggressive and dangerous snakes. More likely, your local guides will point out gorgeously marked vipers lying on the forest floor and then insist on bludgeoning them to death. Killing snakes that your family or neighbours might tread on is a social responsibility in many places. Treat the corpse with as much caution as you would afford to a living animal, for even severed heads are capable of biting some time after death and should be picked up with sticks. In many cases the snake is left to rot. It may be needed as a specimen but I make a habit, which I recommend to other biologists, of cooking the unfortunate creatures for supper, in a pot that I take especially for the purpose. Many people are surprised that I do not die from the meal, supposing that the venom is in the meat, but people who taste it often resolve not to let the meat go to waste in future. I often come across harmless snakes that have been killed along paths. I carefully stitch them back together and leave them in a jar of preservative in a prominent place with a label in various languages "I am not harmful - please don't hurt me!"

4.3.1 Boids

There are over 60 species of boids, muscular snakes that include the pythons and boa constrictors. None of them are venomous but they are all powerful animals with long teeth. Many species are heavily exploited by mankind for meat and skins and the status of most wild populations is unclear. Boids differ very much in temperament; some, like the West African royal python, are very gentle animals that never attempt to bite, whilst others are not.

Figure 4.6. Python regius never bites.

4.3.2 Vipers

Terrestrial or arboreal snakes. Although these beautiful snakes are all venomous they rarely bite unless provoked. Many vipers act sluggish but most species can move very fast when they want to. A few can jump from a static position. Many vipers are very well camouflaged and difficult to spot on the ground or in vegetation.

4.3.3 Elaphids

Fast moving venomous snakes that include the cobras, kraits, mambas, coral snakes and sea snakes. A few species show parental care in which both sexes guard the nest. Some people catch them with hooks but I prefer to use tongs.

Figure4.7: A large cyptic ground dwelling viper.

Figure 4.8. A more conspicuous arboreal viper.

4.3.4 Colubrids

About 75% of the World's snakes are colubrids. The group includes a mishmash of snakes found all over the world. Colubrid snakes include the ratsnakes, king snakes, racers, whipsnakes, grass snakes, egg eating snakes and snail eating snakes. A few species are venomous (including the rear–fanged boomslang) but most are not. It is impossible to generalise about this huge group.

4.4 Habitats

4.4.1 Deserts

Sandy deserts are wonderful places to find reptiles and are often home to a surprising number of amphibians as well. One nice thing about deserts is that they contain simple plant communities that can be characterised relatively easily, and most of the ground is clear sand. Pitfall traps and drift fences are easy to install in deserts, and there is plenty of scope for looking at footprints, especially in the spring, when most animals will be active. Deserts are very easy places to get lost in and because of the sometimes fierce heat, traps of all types need to be checked very regularly, especially during the hottest parts of the day.

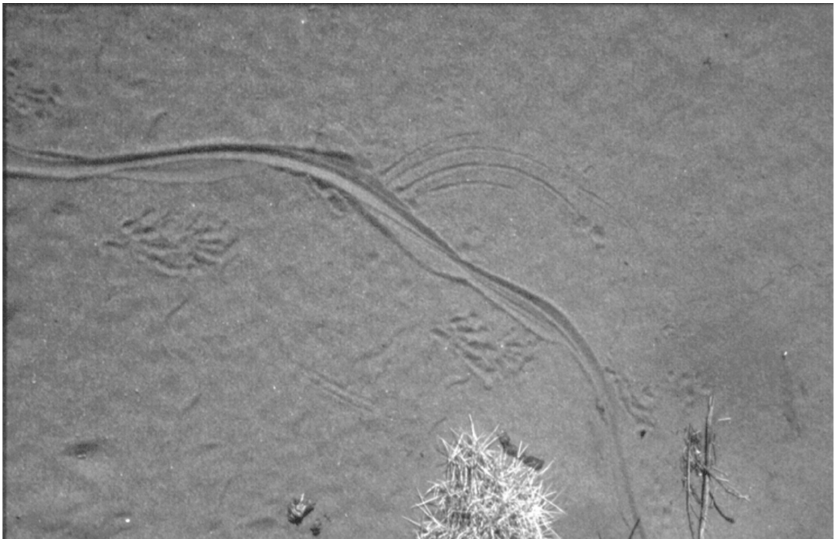


Figure 4.9: Footprints of Australia's largest lizard, Varanus giganteus, in the Great Victoria Desert. The long claw marks indicate that the animal is tired and literally "dragging its feet".

4.4.2 Forests

Forests provide a very rich source of different microhabitats for reptiles and amphibians. Because of the complexity of the plant communities they always require enthusiastic botanists to describe vegetation sites. Usually it is impossible to sample all habitat types equally. High canopy, in particular, is very difficult to survey. Methods that might be useful are sticky traps placed on branches within the canopy or even arboreal drift fences. For most short studies, there will be plenty to occupy teams closer to the ground. High canopy work is dangerous. If you try it you should concentrate on searching just a few trees thoroughly and always work with ropes, safety harnesses and people experienced in their use. Further reading on canopy access can be found in Perry (1978), Moffett (1993), Munn *et al.* (1995) and Moffet and Lowman (1995).

Forests can be very wet places and British university holidays tend to coincide with the rainy season throughout the Old World. This is ideal for many studies, because animals are more active at this time of year, but it can put enormous pressure on team members' morale and equipment unless adequate preparations are made. Notebooks rendered illegible, electronic equipment ruined, wet clothes, rotting feet etc. are all problems that can be greatly reduced with some foresight.

Figure 4.10:. Lizards that live over water can sometimes be noosed or caught in nets.

4.4.3 Swamps, rivers and riverbanks

Swamps, rivers and riverbanks are often rich in reptile and amphibian life. Searching riverbanks by cruising in canoes is a particularly profitable method. I have found that surveying in an upstream direction makes it much easier to stop and look at animals without disturbing them. Be careful to put any traps set on riverbanks above the high water mark. Muddy riverbanks can provide a rich source of footprints which may be used to make estimates of the densities of easily recognised species. Remember that water is dangerous and surveying aquatic habitats always provides plenty of opportunities for drowning.

4.5 Equipment

Airlines presume that their poorer passengers need no more than 20kg of luggage for their trips. For expedition work this can be a real problem. Most expedition members bring about 15kg of their own equipment (sometimes much more). Although most airlines do not take the 20kg limit too seriously, when they do excess baggage charges can be over £100 per item. Usually a smile and explanation that the equipment is for "conservation work" will do the trick. But if the aeroplane is very full or the check in staff don't like the look of you it can get very expensive. I find it best to send the most beautiful expedition members to the checkout desk first, with all the heavy items of luggage. Keep the weight of luggage down by limiting the amount of personal equipment that members bring and not taking anything that can be bought in the host country. This will probably include batteries, tarpaulins, animal bags, field clothes (except socks and boots), cooking equipment (except decent tea bags), tape measures, machetes, spades, ropes (except climbing ropes) and string. A surprising amount of necessary equipment can be purchased locally, saving some of your meagre luggage allowance and allowing you to spend more of your sponsors' money in the host country.

In my experience the things that **cannot** be purchased in many poorer countries are:

Decent boots. It is unlikely that many of the local people you work with (including students and forest rangers) will have a suitable pair of boots. Fatal snakebites nearly always have barefooted or sandled victims. A variety of sizes of army surplus boots from home will be very popular and useful. Used ones are much cheaper and take less breaking in.

Good torches (but not batteries which are always cheaper in tropical countries. If necessary weigh a single battery to check for fakes). The 4.5volt flatpack battery that powers most Petzl headtorches seems to unobtainable almost everywhere in the world.

Good scales. Only Pesola seem to make very accurate and durable spring balances in the 1-1000g range. But you pay through the backside for them, especially if you buy them in Britain. Much cheaper to buy a few sets mail order from the USA. As it states on the box, “Pesola scales are not children’s' toys”. The reaction of the uninitiated on seeing a Pesola for the first time is always to stretch the spring. Pesolas are durable, but they are too precise to withstand the rigours of dirty fieldwork. I prefer to keep them in the field station and bring animals back to weigh them. When I have to take them out I keep them as dry and clean as possible.

Waterproof paper is a great advantage in wet dirty field work. The photocopy paper is nowhere near as durable as the coated notebook sheets. Waterproof paper would also be much cheaper in north America were it not for the postage costs.

If you buy goods abroad for use abroad, any VAT paid is Britain should be refundable. Contact your local VAT office for details.

Section 5

SHARING RESULTS AND DISTRIBUTING REPORTS

5.1 Writing reports

Every member of the team and all academic supervisors and relevant organisations such as wildlife/forestry departments, conservation bodies and universities should have a paper copy of the raw data collected by the expedition. I like to keep a copy of all field data logged in one book that is photocopied and distributed as soon as field work finishes. At least two people should be responsible for putting raw data into a computer spreadsheet, that can then be distributed to everyone involved in writing the report.

If your home university uses state of the art software it may not be readable by older systems. If necessary input the data on an old software package that you know is compatible with the equipment of your collaborators. The older programmes can be read by the newer ones, but not vice versa.

In my opinion, complex statistical analyses are rarely an important aspect of expedition reports and most should be reserved for peer-reviewed publications. The format should be simple, with abstracts from each section of the report together at the front. Maps, diagrams, line drawings and black and white photographs should be used but limit the use of colour to the cover, if at all. Sponsors like to see their logos on the title page.

5.2 Distributing reports

The report will probably weigh at least 300g and costs several pounds to post abroad. Keep the costs down (and save paper) by printing on both sides of each sheet and consider shrinking the format from A4 to A5. If insufficient money exists, make richer students pay for their copies to subsidise the cost of copies set abroad and distribute it in electronic format to home institutions and sponsors. If they like it they may consider producing and distributing paper copies for you.

Copies must go to all organisations and individuals in the host country who co-operated with the work, and other local Universities and conservation organisations, even if you have had no previous contact with them. It's important to send reports directly to the people they are intended for. If you just send a box of reports to the head office of a large underfunded department there is no guarantee that they will ever be seen by anyone. Request acknowledgement from everyone you send the report to, especially in countries with unreliable postal systems.

The report is the purpose of the whole project and without it the expedition will be viewed as a complete waste of time and money by everyone else. There is a danger that some expedition members will loose enthusiasm when it comes to writing the report and in these cases a disproportionate amount of the work lands on a few shoulders. Ultimately it is the responsibility of the Expedition Leader to produce the report. They will therefore be wise to insist that enough money is left in the bank account to cover the costs of production and distribution.

5.3 Making the report effective

Very few people will have time to read the report from cover to cover, especially heads of national parks, government departments and the like who are ultimately responsible for approving any action taken on your recommendations. Ideally they should be briefed in person on the project’s finding and conclusions. This is most easily and effectively done by setting aside enough money for one of your local counterparts to deliver the reports in person and discuss the findings with the relevant authorities.

FURTHER INFORMATION

6.1 Useful contacts

Local herpetological societies will have members happy to teach you how t0 handle reptiles and amphibians. Remember that animals in the wild tend to be a bit more vigorous than those accustomed to captivity! Contact with your nearest groups can be made through the British Herpetological Society (address below). The BHS and some other societies have excellent libraries that might have books and journals missing of the larger institutions. Zoos may also co-operate with advice and training in animal handling. A comprehensive list of zoos with their interests together with some details of animals kept can be found in the International Zoo Yearbook.

The Society for the Study of Reptiles and Amphibians produces many useful publications, including Herpetological Review in which many workers share their, often ingenious, techniques for studying animals in the field. The society also publishes many important guide books and monographs.

British Library of Wildlife Sounds
The British Library, 96 Euston Road, London NW1 2DB
Tel: 0171 412 7440
Website: www.bl.uk/collections/sound-archive/wild

Department of Environment
Trade in Endangered Species,
Tollgate House, Houlton Street, Bristol BS2 9DJ
Website: www.defra.gov.uk

British Herpetological Society
C/O Zoological Society of London
Regents Park, London NW1 4RY
Tel 01258 857869
Website: www.thebhs.org

Society for the Study of Reptiles and Amphibians
St Louis University, 3507 Laclede, St Louis, MI 63103
Website: www.ssarherps.org

Snake Venom Research Unit
Liverpool School of Tropical Medicine
Pembroke Place, Liverpool L3 5QA
Website: www.liv.ac.uk/lstm/research/venom_research

6.2 Equipment suppliers

BioQuip
17803 La Salle Avenue, Gardena CA 90248 3602
Email: bioquip@aol.com
Website: www.bioquip.net
Wide range of surveying equipment

BCB International Ltd
Clydesmuir Industrial Estate, Cardiff CF2 2QS
Website: http://webshop.bcbin.eu/
Camping equipment

Swains Packaging
Brook Road, Buckhurst Hill, Essex IG15 5TU
Tel: 0181-5049151, Fax: 0181-5061892
Website: www.swainspack.co.uk
Plastic bags

GB Nets
45 Burnley Road, Todmorden, Lancs OL14 7BU
Hand nets and accessories

Miller & Weber Inc.
1637 George Street, Ridgewood Queens, NY 11385, USA
Tel: 718-8217116, Fax: 718-8211673
Website: www.millerweber.com
Cloacal thermometers
J.G.Natural History Books
17 Streatham Vale , London SW16 5SE
Tel: 0181-764 4669
Email: ReptileBooks@hotmail.com

Nelson Paint Co.
PO Box 2040, Kingsford, MI 49802, USA
Tel: 906-774-5566, Fax: 906-774-4264
Website: www.nelsonpaint.com
Paintball guns and pellets

Tomahawk Live Trap Co.
PO Box 323, Tomahawk, WI 54487, USA
Tel:715-453-3550, Fax: 715-453-4326
Website: www.livetrap.com
Animal traps

IUCN Publications Unit
219c Huntingdon Road, Cambridge CB3 0DL
Website: www.iucn.org/bookstore

BJ Herp Supplies
Purlands Farm, Bridport Road, Dorchester DT2 9DJ
Tel: 01305 261302, Fax: 01305 261446
Website: www.reptilekeeping.net
Tongs and snake hooks

Sherman Trap Co.
3731 Peddie Drive, Talahassee, FL 32303 USA
Tel: (00 1) 904 575-8727, Fax: (00 1) 904 575-4864
Website: www.shermantraps.com
Animal traps

Atlantic Paste & Glue Inc.
4-53rd Street, Brooklyn, NY 11232, USA
Tel: (00 1) 718 492 3648
Website: www.catchmaster.com
Sticky traps

Midwest Custom Products Inc.
8608 East 32nd Street, Kansas, MO 64129, USA
Tel: (00 1) 816-8613351, Fax: (00 1) 816-8614126
Website: www.tongs.com
Snake handling tools

Olympus Keymed
Stock Road, Southend on Sea SS2 5QH
Website: www.keymed.co.uk
Fibrescopes and videoscopes

Section 7

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Appendix 1

The Declining Amphibian Population Task Force Fieldwork Code of Practice

1. Remove mud, snails, algae and other debris from nets, traps, boots, vehicle tyres and all other surfaces. Rinse cleaned items with sterilized (eg. boiled or treated) water before leaving each study site.

2. Boots, nets, traps etc. should then be scrubbed with 70% ethanol solution and rinsed clean with sterilized water between study sites. Avoid cleaning equipment in the immediate vicinity of a pond or wetland.

3. In remote locations, clean all equipment as described above (or with a bleach solution) upon return to the lab or "base camp". Elsewhere, when washing-machine facilities are available, remove nets from poles and wash with bleach on a "delicates" cycle, contained in a protective mesh laundry bag.

4. When working at sites with known or suspected disease problems, or when sampling populations of rare or isolated species, wear disposable gloves and change them between handling each animal. Dedicate sets of nets, boots, traps and other equipment to each site being visited. Clean and store them separately at the end of each field day.

5. When amphibians are collected, ensure the separation of animals from different sites and take great care to avoid indirect contact between them (e.g. via handling, reuse of containers) or with other captive animals.

Isolation from unsterilized plants or soils which have been taken from other sites is also essential. Always use disinfected/disposable husbandry equipment.

6. Examine collected amphibians for the presence of diseases and parasites soon after capture. Prior to their release or the release of any progeny, amphibians should be quarantined for a period and thoroughly screened for the presence of any potential disease agents.

7. Used cleaning materials (liquids etc.) should be disposed of safely and if necessary taken back to the lab for proper disposal. Used disposable gloves should be retained for safe disposal in sealed bags.

The DAPTF Fieldwork Code of Practice has been produced by the DAPTF with valuable assistance from Begona Arano, Andrew Cunningham, Tom

Langton, Jamie Reaser and Stan Sessions. For further information on this Code, or on the DAPTF, contact John Wilkinson, Biology Department, The Open University, Walton Hall, Milton Keynes, MK7 6AA, UK. E-mail: DAPTF@open.ac.uk. Fax: +44 (0) 1908-654167