FAUNA SAMPLING MANUAL

Guide to sampling techniques for wildlife research in Western Australia

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Conservation Council of Western Australia ccwa Look forward

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PURPOSE OF THIS DOCUMENT

This guide supports a mentor-based training program designed to provide participants with sufficient field experience in the most common capture and sampling methods to qualify for licences to take or mark fauna in Western Australia (currently Regulations 17 and 23 of the *Wildlife Conservation Regulations 1970*, under the *Wildlife Conservation Act 1950*), and comply with animal welfare requirements (*Animal Welfare Act 2002*).

There are existing Standard Operating Procedures (SOPs) for many of the techniques presented here, and publications such as Thompson and Thompson (2011) and Hyder *et al.* (2010) that describe the application of sampling techniques for environmental impact assessment. The combination of the current guide and training program, however, is intended to provide participants with an understanding of the basics, complexities and subtleties of sampling techniques whatever the circumstances under which they may be used. Such understanding goes beyond following a SOP, and enables participants to make the sorts of ongoing decisions that inevitably have to be made when sampling for fauna in changeable natural environments.

The aim of the guide and the training program is to capture and share the considerable experience that lies behind the implementation of sampling techniques, and therefore to foster the effective and ethical use of traps and other sampling techniques so that maximum conservation value is gained and minimum harm done to fauna. The program does not cover activities that involve the collecting of protected fauna for laboratory study, translocation, or the use of lethal methods or highly intrusive techniques in fauna investigations.

This document is structured in three main parts.

'Planning and Principles of Fauna Surveys' covers administrative requirements. Some discussion on sampling design is provided as an introduction to this extensive topic.

'Sampling Methods for Vertebrates' and 'Sampling Methods for Invertebrates' review the range of methods that can be used for sampling vertebrates and invertebrates, including trapping and observational techniques. The intention is to explain how the techniques are used and the section includes advice on animal handling. Information is provided on the setting and management of traps (including checking and minimising mortality), and approaches to the handling of key animal groups likely to be encountered during sampling, including guidance on handling techniques specific to particular methods. Methods and handling are summarised in tables of competencies that provide checklists for the assessment of skills.

'Making Observations Count' examines identification, measurements and observations that can be made to determine age, sex and reproductive condition, and how fauna can be marked. It also includes advice on the non-destructive collection of tissue samples for DNA analysis. Advice is also provided on recording standards and the maintenance of survey data.

As a companion to this document, a guide to survey and sampling techniques for investigations into fauna of conservation significance is under development but is not yet available. This considers techniques as they apply to the individual species, many of which, because of their significance, are hard to locate and have to be sampled with great care.

Regulation of Fauna Survey and Research in Western Australia

PROTECTED FAUNA

Field studies on fauna may require a licence if the activities are considered to involve taking a protected species. In the current State legislation (*Wildlife Conservation Act 1950*), 'take' includes to capture, disturb, molest, hunt or kill any fauna, to attempt to do so or to assist another party in doing so. 'Fauna' includes any part of an animal whether dead or alive or attached to the animal (the carcass, skin, plumage or fur) and specifically includes eggs, larvae and semen. 'Material' (eg. fur or bone) collected from scats or pellets, moulted feathers and sub-fossil deposits is still regarded as being fauna in respect to the Act, and it may thus be necessary to prove the origin of the material. It is therefore prudent to obtain a licence to take fauna if one intends to take possession of such materials. Fossilised fauna are minerals and their extraction is subject to the Mining Act (*Mineral Resources Act 1989*).

Where protected fauna is legally able to be taken under other legislation which has precedence over the Wildlife Conservation Act, such as the taking of fish under the *Fish Resources Management Act 1994*, a further licence to take fauna is not required under the Wildlife Conservation Act.

Western Australia's fauna protection laws are administered by the Department of Parks and Wildlife (DPaW; formerly the Department of Environment and Conservation) and currently prescribed in the *Wildlife Conservation Act 1950* and its regulations. This legislation protects all animals that have been defined as fauna according to the definition in the Act, and excludes any animal that has been specifically declared 'not protected'. Protected fauna includes any animal indigenous to any State or Territory of the Commonwealth, or the territorial waters of the Commonwealth, including visiting migratory species.

SPECIALLY PROTECTED & COMMONWEALTH LISTED FAUNA

Fauna may be declared by the Minister for Environment under Section 14(4) of the *Wildlife Conservation Act 1950* as 'specially protected' and categorised as rare or likely to become extinct, or otherwise in need of special protection. Such fauna are regarded as being threatened fauna and fauna otherwise in need of special protection, and also include migratory bird species listed under certain international migratory bird agreements. The State list of specially protected fauna is periodically revised and published as a Notice in the Government Gazette. The most recent revision was published as the *Wildlife Conservation (Specially Protected Fauna) Notice 2012 (2)* and is available on the DPaW and State Law Publisher's websites.

Nationally threatened fauna, and migratory and marine species, may also be listed under Federal legislation (*Environment Protection and Biodiversity Conservation Act 1999*). The State's specially protected fauna list is being harmonised with the EPBC Act lists of threatened and migratory species.

The targeted 'taking' of threatened or otherwise specially protected fauna requires the specific endorsement of the fauna species on the Wildlife Conservation Act licence, and any assessment of the approval is likely to be subject to great scrutiny. Research projects involving species listed as threatened, migratory or marine under the EPBC Act require a licence from the Department of Sustainability, Environment, Water, Population and Communities (DSEWPaC) if they are to be carried out on Commonwealth lands within the State or in Commonwealth Waters.

WHAT LICENCES ARE REQUIRED?

Licences required for any fauna survey or research project conducted in Western Australia may include: a licence to take fauna for scientific purposes (Regulation 17); a licence to permanently mark fauna (Regulation 23, if issued for bird and/or bat banding activities, dependent upon the applicant also holding an Australian Bird and Band Banding Scheme Authority); a Regulation 15 Licence (replaces the Regulation 17 licence if solely for the relocation of fauna during clearing or for the removal of fauna from infrastructure); and a DSEWPaC licence for activities on Commonwealth lands or waters.

If the land on which the activity is to take place is managed by the DPaW, a Regulation 4 licence (under the Conservation and Land Management (CALM) Regulations 2002) is required to carry out research activities. This applies to State Forest, National Park, Nature Reserve, Conservation Park and Unallocated Crown Land. This licence is also used to authorise camping or the deployment of any reference markers or fixed field equipment. If the land is not managed by the DPaW, written permission of the land owner, holder or manager of the land on which the activity is to occur is required. Land-holders or managers might include other State agencies, local governments (for shire reserves), private property owners, pastoral lease owners and traditional owners. Permission may also be needed from the owners of operating mining leases but not exploration licences.

BIRD AND BAT BANDING

The Australian Bird & Bat Banding Scheme (ABBBS) coordinates and licenses the capture of birds and bats for marking with centrally issued and data-based bird and bat bands. Banders are required to train under the mentorship of experienced licensed operators until they have sufficient experience to develop independent projects (for ABBBS authorisations see www.environment.gov.au/biodiversity/science/abbbs/). This activity, including the approval to use restricted traps (eg. mist-nets, cannon-nets), is regulated at the State level, usually with Regulation 23 licences to take and permanently mark fauna for research purposes. Fauna marking other than bird and bat banding is generally handled using Regulation 17 licences to take fauna.

A DECISION KEY FOR FAUNA LICENSING

1. Does the survey/research activity or method constitute 'taking' fauna?

   If **No** – no licence to take or mark fauna is required

   If **Yes** – go to question 2

2. Does the survey/research activity or method require the use of ABBBS bird or bat bands or the use of mist-nets?

   If **No** – go to question 3

   If **Yes** – Register as an ABBBS trainee under an A Class bander or register a new project if qualified. If you are a start-up bander you will need to obtain a DPaW Regulation 23 licence, and continue to question 3

3. Will the survey / sampling take place on DPaW managed land or marine waters, or Commonwealth lands or waters?

   If **No**, obtain written permission (may be an email) from the land-owner / manager to carry out the project on the land then go to question 4

   If **Yes**, go to question 4

4. Does your survey/research plan consider objectives, sampling design, sampling methods, data management, predictable interactions with listed species, animal welfare issues, environmental management and biosecurity?

   If **No**, revise project plan and outline how you will deal with these issues.

   If **Yes**, proceed to Regulation 17 (or Regulation 15 if project involves relocation of fauna) application for a licence to take fauna for scientific purposes (include targeted listed fauna species in the application if applicable).

   If on DPaW managed land include application for Regulation 4 permit.

   If involving listed species on Commonwealth land or waters, apply to DSEWPaC for permit under Part 13 of the EPBC Act.

LICENCE RENEWAL

State fauna licences are usually granted for a period up to 12 months and require renewal if a project is to continue for longer. Regulation 17 licences are issued on a per-project basis, and the application for these licences should specify the overall duration of the project to facilitate a full assessment of the project at the time of initial licence issue.

If a renewal is required it must be requested and is conditional upon digital reporting of fauna handled under the current licence. Expiry notices may be sent to holders of Regulation 17 licences and indicate the option for licence renewal, but no notices are sent for Regulation 4 licences. Regulation 23 licences are renewed annually following notification by ABBBS that the holders have complied with reporting requirements and have had their authorities renewed.

EPBC Act licences may be granted for longer periods of time. Reporting requirements for Part 13 EPBC Act licences are specified in the conditions of grant.

The Ethics of Fauna Capture

Licences to take fauna for scientific purposes will not be granted for activities that might put animal populations at risk, however all wild-capture methods will involve some risk to the survival or long term fitness of individual animals. The conditions and hazards prevailing in the external environment, and particularly in natural ecosystems, can be predicted with varying degrees of certainty but cannot be controlled as is the case with laboratory research.

As research involving the capture of fauna always involves some risk to the subjects, the work must be justified. Put another way, the benefits to fauna conservation, ecosystem management, environmental decision-making or the body of human knowledge must outweigh any potential harm or distress the capture of animals might cause.

Any authorisation for the capture of fauna for survey or research purposes comes therefore with the ethical and scientific responsibilities to share and disseminate the results, either through common, widely accessible databases or preferably by publication in the scientific literature. This not only ensures that the risk to individual animals, however minor, is justified, but also that future survey or research activity are efficient in their 'use' of animals, both key objectives of the *WA Animal Welfare Act 2002*.

This guide does not deal specifically with the requirements of the *WA Animal Welfare Act 2002*. It is hoped, however, that the fauna survey methodologies covered in this guide will adequately address issues of animal ethics in the manner that fauna are treated, and may consequently be accepted as 'Standard Operating Procedures' by the various Animal Ethics Committees, and the DPaW, for the approvals process with respect to animal ethics considerations.

Legislation aside, good animal welfare practice should be front and centre in any fauna survey or research program. The specific issues that relate to the selection and operation of capture methodologies will be discussed later in relation to those techniques, however some general considerations are:

1. Animal welfare outcomes should not be compromised by cost considerations. Field management intensity must match the animal welfare risks associated with the operating conditions (eg. trap checking frequency, triggers for trap closure).
2. Operators should be appropriately trained and skilled and trainees / students need to be supervised in the field by experienced licensed practitioners until they have demonstrated sufficient competence.
3. Selection of capture methods should consider not only the sampling of the research subjects but also minimising the by-catch of non-target species and / or the collection of usable data (eg. identity, measurements) from these species.
4. Fauna should be retained for the shortest period possible to complete the sampling / marking / recording protocol, unless release at another time would enhance the probability of subsequent survival (eg. in torpid animals or nocturnal species that might be better held until nightfall).

5. Adverse outcomes (eg. trap morbidities or mortalities) should be recorded and reported. Trap-mortalities should be retained as voucher specimens or provided to other relevant experts where possible.
6. Appropriate equipment / anaesthetics must be on hand to euthanise any injured or moribund animals.

Wildlife Disturbance

Wildlife disturbance occurs when the presence of people leads to a change in behaviour that is potentially detrimental to the survival of fauna. In the general fauna survey context, where animals are dispersed over expansive territories or home ranges, wildlife disturbance is unlikely to be significant (nests or burrows near regularly visited traplines might be a minor exception). Disturbance by researchers is a more significant factor where high concentrations of wildlife occur, such as in seabird, water-bird and sea-lion breeding colonies, on mudflats and roosting sites used by concentrations of shorebirds, and bat colonies in caves.

Procedures developed for research on a seabird colony provide a good example of the management of disturbance impacts and are given in Box 1, which explains the sampling protocol developed for band, release and recapture activities at a Common Noddy colony on Lancelin Island over an eighteen year period, during which time the colony expanded from 16 to about 1800 pairs. The key to this protocol is the understanding of the species' biology, its response to disturbance and the management of habituation (the dampening of flight response over time as the birds become familiar with and accept a certain level and type of benign activity). Different seabirds and situations may require other approaches to managing disturbance at breeding colonies, but the protocol for Common Noddies provides a guide.

Roosting and foraging shorebirds are sensitive to disturbance because, like seabird breeding colonies, the birds are concentrated in a small area that meets their requirements. Foraging shorebirds may have a limited window of time to forage when the tide is low and mudflats exposed, making them vulnerable to lost foraging opportunities if disturbed, while roosting shorebirds expend unnecessary energy if forced to take flight. As these are often migratory species that need to accumulate fat reserves in preparation for long-distance movements, disturbance at any time can interfere with critical energy budgets. Roosting and foraging shorebirds can be disturbed during ground and particularly aerial surveys, while the capture of birds for banding can be very disruptive of large numbers of individuals. Banding can also be very useful for research and conservation and should only be carried out by personnel with appropriate approvals from the Australian Bird and Bat Banding Scheme.

Visits to bat colonies in caves can result in the movement of bats into less favourable micro-climates (outside the roosting cave) or to the dislodgement of young from maternity roosts. Frequent or prolonged stays, particularly in cold caves (winter roosts), can force changes in micro-climates. As with seabirds, longer-term or repeated sampling investigations of cave bats need to develop protocols to maintain disturbance impacts within acceptable limits.

Eastern wheatbelt remnant (photo T. Gamblin)

BOX 1

Disturbance management at a common Noddy colony on Lancelin Island

In order to investigate the demographic structure of a recently established population, Common Noddies were captured by hand, or using long-handled nets, while nesting in a densely-packed colony on Lancelin Island (Figure 1). Most nests consisted of a platform lined with seaweed built on crowns of Nitre bushes. The start of egg-laying varied from late September to mid-December depending on the availability of the preferred prey, which in turn was controlled by oceanographic conditions along the edge of the continental shelf.

The following practices were adopted to ensure that research disturbance was not a significant factor in the dynamics of the Common Noddy colony.

1. No attempt was made to capture, handle or census Noddies until at least 2 weeks after the first eggs were laid.
2. Mark, release and recapture operations on nesting adults occurred (wherever possible) late in the incubation period for the laying peak, when site-tenacity was at its highest (and thus the birds were least likely to abandon nests).
3. No attempt was made to capture Noddies if site-tenacity and attentiveness were reduced by low food availability.
4. Banding operations generally took place in the first two to three hours after sunrise to avoid high temperatures and interactions with the public.
5. Operations in the colony were avoided in the late afternoon when nest change-overs occurred to avoid food regurgitation (except when it was necessary to obtain data on the prey being taken).
6. Banding and associated operations were limited to 90 minutes on any field day.
7. The cohort of chicks was banded in the early nestling stage when the young will not leave their nests during colony disturbance.
8. The colony was not entered when large (down & quill) chicks were present to avoid stimulating chick movement across adjoining territories. Breeding success was only broadly assessed by observation of the numbers of fledglings in colony and beach roosts at the end of the season.

This sampling protocol reflects a number of guidelines that can be used to minimise disturbance and maximise the value of habituation. Disturbance should be:

- **regular** – occurs at sufficient frequency to become familiar,
- **consistent** – use an established route or pattern of movement,
- **moderated** –researcher activity should be quiet, passive and sensitive to seabird flight responses and predator behaviour, and
- **time limited** – duration of operations should be rationed to reduce any disruption to normal behaviour. eg. brooding or chick provisioning.

Figure 1. Common Noddies in flight over Lancelin Island (photo J.N. Dunlop).

Biosecurity

Ecologists, along with their vehicles and field equipment, come into close contact with a variety of natural environments and plant and animal species, often moving between landscapes and biological regions between one survey or research project and the next. As such we are potentially a more significant bio-security risk than many other members of the community. Plant diseases, weeds, parasites, pathogens and native species from other regions may be inadvertently translocated from place to place in the absence of appropriate biosecurity practices. Island ecosystems are particularly vulnerable to the introduction of terrestrial plants and animals from the mainland or from other islands.

A biosecurity assessment of most field operations will reveal potential risk factors requiring management.

Vehicles – Off road vehicles used for fauna surveys may accumulate soil and organic matter harbouring plant pathogens, such as Jarrah Dieback. Tyres may also collect the burrs from weed species. Vehicles should be washed and / or brushed down and inspected before leaving a study site.

Tools – Digging implements and other tools may have soil of vegetative material adhering to surfaces. They should also be inspected and cleaned if required before being packed-up.

Camping equipment (eg. tents & swags) – As for vehicles.

Field clothing – Soil and plant seeds may adhere to clothing, particularly boots, socks and trouser cuffs. A clothing inspection is particularly recommended before landing on islands, and between sites.

Calico bags & other holding facilities – Cloth bags that have held animals could act as a vehicle for the transmission of parasites between bio-regions and pathogens between resistant and naïve populations. Bags used to hold mammals, reptiles, frogs or birds in one bio-region should be washed and disinfected after trapping sessions. Where biosecurity is rated as a high risk (eg. islands, remote locations), the safest way is to use new, clean bags, allocate a set of bags / containers to the specific project and leave them on-site for future use, or dispose of them at the study site when the project is completed. Note - cloth bags used for holding animals, even for very short periods, should be checked for loose threads that may entangle specimens.

Traps – Traps and bags can collect soil and seeds. They may also be contaminated by trapped animals and thus need to be washed and disinfected if moved between sites. Cleaning traps is discussed in the sections on each trap type.

Specimens – Live fauna should only be transported in appropriate and secure holding equipment. Dead specimens should be transported frozen or in preservative (5% formalin, 70% or 100% ethanol). In some cases, such as where an autopsy of a specimen of high conservation significance is required, the specimen should be chilled, sealed in a plastic container and transported to veterinary facilities as quickly as possible. Specialist advice should be sought for the wet preservation of specimens as the chemicals involved are hazardous and often require specific permits for their use, transport and storage.

Personal hygiene – Fauna are a potential source of zoonoses and therefore personal hygiene is important. Avoid hand to mouth contact when handling animals, hand-washing facilities should be available and protective equipment, such as safety glasses with eye-shields, may be needed under some circumstances. Personnel should ensure that their tetanus vaccination is current.

Protective clothing – Appropriate clothing should be worn during fieldwork. Long pants, hiking boots, gaiters, long-sleeved shirts, wide-brimmed hats, sunglasses and sunscreen are required for many surveys. Some fauna studies are carried out in conjunction with mining projects and there may be an expectation that personnel will meet mine-site requirements such as steel-capped boots, hard hats and high-visibility clothing. These may not be necessary or appropriate for the sort of work being undertaken. Steel-capped boots are not suitable footwear for walking long distances, while high-visibility clothing may bias bird survey results and hard hats make difficult the checking of traps, measuring and handling animals, and operating binoculars. The issue of protective clothing suited to the nature of work being conducted should be discussed before fieldwork commences.

Sampling Design

Fauna surveys are carried out for a range of reasons; the approach to sampling needs to be appropriate for the purpose and the expectations of the results need to be realistic. For example, few surveys conducted for the purposes of environmental impact assessment in Western Australia (and meeting the Level 2 requirements of EPA Guidance 56) would generate sufficient data to provide for statistical analysis (eg. to test for patterns in habitat use or inter-seasonal or annual variations in abundance). Equally, short term surveys involving one or two seasons in a given year are unlikely to provide a comprehensive inventory of the diversity in any fauna group. Inter-annual variations in fauna abundance are significant, particularly in the sub-humid and arid parts of the State, and these changes strongly influence the ability to detect many species.

Despite these technical limitations, fauna surveys should be designed to allow the baseline work to be extended where possible. For example, baseline surveys could become the foundations of future ecological monitoring programs. The sampling design should therefore ensure that:

- study sites are representative of the range of local environments,
- the sampling of environments is replicated,
- there is enough field time to apply sufficient and comparable survey effort at each site,
- sampling is timed to take place during optimal periods, and
- the survey as a whole could be repeated by other practitioners at a later date.

STUDY SITES REPRESENTATIVE OF LOCAL ENVIRONMENTS

The first stage in any fauna survey is a broad brush assessment of the range of environments present within a project area (eg. Level 1 Survey in EPA Guidance 56). This preliminary work should then guide the sampling design which can either be targeted (aimed at detecting particular species of interest eg. threatened species) or comprehensive (to provide an overall faunistic interpretation of the environment).

Targeted surveys will focus on the environments normally occupied by the species of interest. Faunistic surveys will involve sampling in all the environments characteristic of the project area.

LAYOUT OF SAMPLING POINTS

Whatever sampling is being carried out (such as pitfalls, cage traps, bird censusing), a range of sampling layouts is possible. There is no "right" layout of trapping or censusing sites, and capture/recording rates are so variable with season and daily weather conditions, and with factors such as vegetation structure and even the species being sampled, that there is no benefit to a universal approach across all projects. The layout is best tailored to the project, environment and anticipated analyses.

Pitfalls in a tight grid or along a common fence provide almost point data that can be compared between sites, while pitfalls in a long transect sample a larger area but there may be variation in the environment along the transect. Where the aim is to maximise the number of species recorded, pitfalls deployed in long transects will probably be most effective as they sample more of the environment than point sampling. Transects have also been found to provide more consistent data between replicates in the same vegetation type than traps placed in quadrats (Reed *et al.* 1988). This appears to be because the distribution and abundance of fauna at the local level (that is, over a few hectares or even over hundreds of square metres) is variable, and therefore clumped traps will be more subject to this variation than dispersed traps.

A common approach is to place sampling sites within large, contiguous areas of fairly uniform vegetation and thus avoid boundaries or transitional zones; this can simplify subsequent analyses by allowing results to be compared between sites within and between environments. However, this approach effectively biases the data collected by imposing environmental categories recognised by scientists upon the fauna sampling results. Boundaries and transitional zones may be important ecological features of the landscape but would be deliberately avoided using this approach. Birdwatchers know that the best places for birdwatching are often at the transition between two vegetation types, and other fauna groups may behave similarly. Because of this concern with sampling bias, the alternative approach of catenary sampling can be used. A catena is the sequence of soils from high to low in the landscape, and thus catenary sampling typically consists of a long transect of regularly spaced sampling points that passes through the different soils and vegetation types of a catena. Sampling points that are in the same vegetation type are grouped for analysis after the sites are laid out, and not based upon some prior assessment of what constitutes a vegetation type. Similarity analyses can be used to group sampling points based upon the sorts of fauna recorded, the vegetation/soil present, or a combination of these. A weakness of catenary sampling is that each sampling point, such as a single pitfall or bird census point, is effectively a site, and therefore a lot of effort may be required to get sufficient records from each site to support analyses. This can be partly addressed by having several traps at each point along the transect.

Sampling layouts based upon prior selection of representative environments versus a catenary approach have pros and cons, but both should be considered in terms of objectives and practicality.

REPLICATION

Relationships between fauna and habitats cannot be determined statistically without sufficient repeat sampling of the same habitat type at different locations (replication). Generally, multiple replicates are not feasible in short term biological surveys carried out for environmental impact assessment purposes (Level 2 in EPA Guidance 56), but the twinning of sites in the same environment may be sufficient for Similarity analyses (eg.Twinspan) or Ordinations (eg. Principal Axis Ordination) that reveal pattern preferences. Data from catenary sampling require a different analytical approach, including non-parametric statistics, but there can be replication within a transect, as well as the option of replicating the entire transect.

SURVEY EFFORT

The amount of effort (trap-days, observation hours) necessary to meet fauna survey objectives varies widely depending on the environmental context and the seasonal, weather and moon conditions prevailing at the time.

Generally the bulk of common and widespread species will be recorded early in the survey after the expenditure of only limited effort, but less common (but potentially more significant) species will only be detected after considerable effort over a longer period of time. As a result, species accumulation curves typically show a rapid accumulation of species with initial sampling, then a gradual and slowing accumulation of further species with additional effort (Figure 2). A faunistic survey may be considered adequate when the asymptote of this curve is reached, but it should be recognised that the survey is then only complete at that time and for that sampling site. Fauna assemblages can change over time and they can also vary over short distances even in what appears to be a uniform environment (Figure 3). Large inter-annual variations in population densities may prevent even some common species from being detected for several years.

Species accumulation curve for reptile species at the Learmonth Air Weapons Range

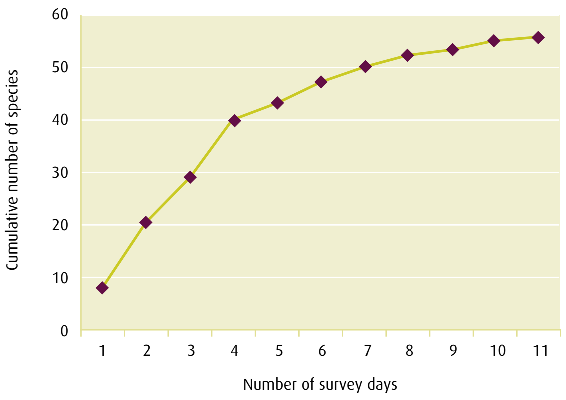

Figure 2. Species accumulation curve from October 2004 (days 1 to 6) and March 2005 (days 7 to 11) combined. Data based on observation as well as trapping results. The second survey period of five days added only five species compared with the 50 species recorded in the first survey period of six days. Within the first survey period, 40 species were recorded up to the fourth day, with only 8 species added on the last two days. Data from HLA Envirosciences Pty Ltd and Ecoscape Australia Pty Ltd (2005).

Accumulation of frog, reptile and small mammal species in Banksia Low Woodland over a 16 year sampling period

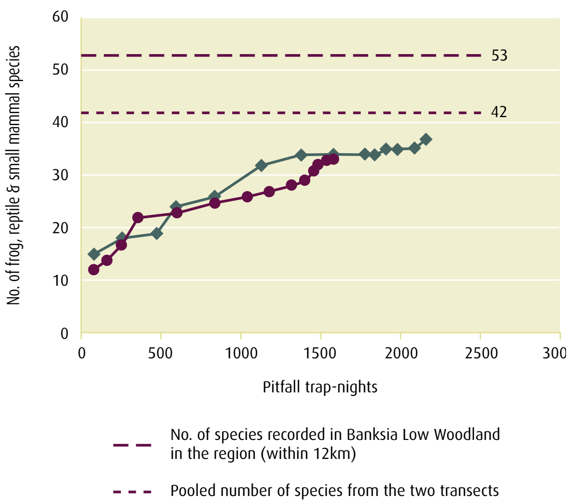

Figure 3. Number of frog, reptile and small mammal species (pooled, vertical axis) plotted against pitfall trapnights (horizontal axis) along two transects 400m apart, both in Banksia Low Woodland. The total number of species recorded in Banksia Low Woodland in the region (within 12km) was 53 (heavy broken line), and the pooled number of species from the two transects was 42 (fine broken line). The two transects had 31 species (74%) in common. These data were collected over a period of 16 years with a low intensity of trapping each year. They demonstrate that the species accumulation curve does not asymptote over long periods of time, due to the dynamics of fauna assemblages, and that the fauna assemblage can differ substantially across short distances, even over long periods of time (Source: Bamford Consulting Ecologists and Tronox, unpubl. data).

TIMING OF FAUNA SURVEYS

Surveys should be conducted during several seasonal periods. At least one of these (the core period) should be in the months following the rainy season, ie. spring in the temperate Mediterranean south, autumn in the tropical north, spring or autumn in the arid mid-west and central regions. At some locations, optimal seasonal periods may have to be excluded in order to avoid breding seasons, or the onset of high temperatures which may compromise animal welfare, however, this situation can often be managed as discussed in trap management sections of this guide (see pages 10-29).

Nocturnal fauna, particularly small mammals (including bats), are more active and more easily captured on the dark nights around the new moon. These monthly periods should be given priority when organising the timing of fauna surveys.

DURATION OF FAUNA SURVEYS

Recommendations for length of survey period vary in the literature from four to seven nights (Hyder *et al.* 2010), with some practitioners using periods as long as 10 nights. The effectiveness of pitfall traps in particular usually declines after three to four nights, but animals will continue to be caught, and a longer period increases the chance of intersecting a weather change which can lead to an increase in captures. The duration of a survey depends upon many factors. In a study targeting frogs that are active only after rain, established pitfalls are opened for just two or three nights to coincide with rain (Everard and Bamford 2013), but it is more usual to run traps for four to five nights to cover the period of optimal capture rates. If a site requires great effort to get to and is unlikely to be revisited, a longer sampling period may be appropriate. Longer trapping periods increase the likelihood of retrapping animals, which can provide more information but can also increase stress on animals / mortality.

ENSURING FAUNA SURVEYS ARE REPEATABLE

Fauna surveys need to be documented in a way that ensures that they can be repeated by other practitioners at a later date to confirm the results, locate taxa of uncertain identity, to extend the survey effort, seasonal or annual range, or to provide a monitoring baseline.

Each regular fauna site should be given a unique survey number and the following information recorded:

1. Coordinates (taken with a hand-held GPS). The WA Museum and DPaW databases use latitude and longitude, but UTM coordinates (Eastings and Northings) are more widely used by industry. These can be converted. If using UTMs, note the zone and the map datum
2. The landscape context
3. The soil type and depth
4. The dominant plant species in each stratum
5. The height / density / life form classes in each stratum
6. The percentage cover (projected foliage density) of each stratum
7. The fire / flooding history to the extent known

SAMPLING METHODS FOR VERTEBRATES

Capture Techniques

Sampling methods addressed in this section include various types of traps and ways of searching for fauna. All ultimately involve the capture and handling of fauna. Some of the techniques require specialist training and equipment (eg. mist nets). Information on each sampling technique is presented under headings as follows:

- Description of technique (eg. materials, styles, providers where applicable)
- How to deploy the traps (eg. trap layout)
- Management of technique (eg. how to set and check the traps, and how to minimise the risk of injury, stress or mortality)
- Handling of animals (eg. method-specific guide to handling animals).

STANDARD PROCEDURES WHEN OPERATING TRAPS

While a range of trap types can be used and require specific details for their operation as outlined in the following sections, there are some standard procedures recommended when operating traps. These are summarised very simply below.

Locations of traps should be clearly marked and the location given some sort of code. This can be as simple as 1, 2, 3..., or could combine a site code with a trap number, and even include information about trap types present. Coloured flagging tape is commonly used and can be written on with a permanent marker pen. Locations that are going to be used for a long period should be marked with a metal fence-dropper or similar. Aluminium tags can be used to label such long-term locations. Where available, the coordinates of locations should be recorded with a GPS unit. Be sure to make note of the datum and coordinate system used. Latitude and longitude are universal; UTM is widely recognised but note needs to be made of the datum and the zone.

Where a site consists of many traps, the details need to be recorded as the site is being set. This is especially important if different people may be checking (and/or closing) the site later.

When traps are being closed and/or taken up, it is very important to keep a record so that all traps have been closed or removed. Personnel need to know how many traps they are checking, and the number closed and/or collected needs to be checked before leaving the site.

The number, types and layout of traps used in a project will depend upon many variables: the questions being asked, site characteristics, the species likely to be encountered, time likely to be taken to check all traps, number (and experience) of personnel available, information to be collected from trapped animals and weather expected during a field trip. Ultimately, the number of traps deployed is limited by the need to minimise stress and mortality, while typically during a fauna survey the aim is to record as many species and individuals as possible.

HAND CAPTURE

Active searching for the hand-capture of reptiles (and frogs to some extent) is widely used and with an experienced team can record more species than the major trapping techniques. Active searching can include simply walking (or driving) around until an animal is seen, turning over rocks, breaking open dead trees and raking through leaf-litter, but can extend to digging out burrows. There are also capture techniques such as the use of a noose to catch fleet-footed lizards such as agamids. McDiarmid *et al.* (2012) provide a review of the many techniques that can be described as hand capture; the following section discusses only active searching techniques widely-used in Western Australia, and some of the ethical and welfare considerations associated with them. All these approaches result in some environmental disturbance which needs to be considered and assessed against the value of the records gained. Note that the reptiles found by searching are often active and can therefore be difficult to catch. There is some evidence that searching is best done in the cooler months not simply because animals are colder and therefore slower, but because they shelter in more conspicuous locations. For example, in winter small reptiles can readily be found close to the surface under leaf-litter, but in summer they appear to shelter at a greater depth. Species can include venomous snakes as well as invertebrates such as scorpions and centipedes. Care therefore needs to be taken.

Searching for reptiles requires skill in recognising where animals might be hiding. Approaches commonly used in WA are:

Turning over rocks and logs

Very effective in a range of circumstances, particularly on rock outcrops where reptiles cannot shelter in burrows. Care should be taken when turning over rocks that reptiles beneath them are not crushed, and rocks and logs should be returned to their position when searching is complete. Repeated disturbance of rocks may make them unsuitable for shelter. This approach should be avoided if the habitat of rocks and logs is very limited and is probably not appropriate for nature reserves and national parks, but is suitable where the habitat is not limited. The approach may be essential where the habitat is threatened by clearing (for example, ahead of a development project) and the survey includes the removal and relocation of fauna. Turning over rocks and logs is usually done by hand, but a jemmy (small crowbar) can be helpful.

Dead trees

These provide shelter under dead bark and in crevices, but searching through dead trees results in destruction of the habitat. Where the habitat is abundant, or is threatened by development, however, it can be a very fruitful technique for finding geckoes, some skinks and small varanids. This sort of searching is usually done by hand, but a jemmy (small crowbar) can be helpful.

Raking through leaf-litter

This is a very effective approach in many areas, and is best done in winter when small reptiles are cool and therefore slow-moving, and when they shelter close to the surface. Raking through leaf-litter is typically done with a three-prong cultivator (often called but technically not a hoe), and raking is best done systematically using short, deep strokes. It is possible that more reptiles are found in litter on the sunny side of trees rather than in cool, damp areas. Raking is not as destructive as other searching techniques as the litter can be pushed back into place and will settle after a few weeks and with further leaf-fall.

Standardising effort

Unlike trapping approaches that are standardised by trapping effort, active searching is not readily standardised as it is skill-dependent, however effort can be timed. Furthermore, rather than searching over a large area by selecting locations where reptiles may shelter, searching can be concentrated into a total removal plot in which every reptile in a measured plot is found through total searching just within that area. Rodda *et al.* (2001) used this total removal plot approach to estimate absolute densities of reptiles in Guam. Bamford and Calver (in prep.) have used a similar technique in sandplain heath north of Perth. Total lizard density was over 400/ha north of Perth, and much higher in the tropical and three-dimensional rainforest of Guam. Although destructive of the environment, the heath at the site north of Perth had regenerated within five years.

DRY PITFALL TRAPS

Pitfall traps are very widely used in fauna studies for the capture of small vertebrates (and invertebrates; see page 40).

MATERIALS

Almost any container that can be sunk into the ground can work as a pitfall trap. Commonly used are 20 litre plastic buckets (40cm deep and c. 28cm in diameter); but varying lengths and diameters of PVC pipe are also used (Figure 4). The merits of different designs of pitfall traps can be debated but the principles of managing them are the same. The buckets are convenient for transportation and storage, come with sturdy, UV stabilized lids (essential and must be UV stabilized), but PVC pipe can be cut to greater lengths if some mammal species (eg. *Notomys* and some *Sminthopsis* spp.) are being targeted. 15cm diameter PVC is commonly used where piping is used for pitfalls, but wide (25–30cm) and deep (>60cm) PVC pipe pitfalls can be effective under some circumstances (such as when targeting mammals that can leap out of 40cm deep pitfalls, including hopping-mice *Notomys* spp.). An alternative to deeper pitfalls to retain mammals that leap high is having a funnel inserted into the top of the pitfall; this may also be cooler as the funnel keeps the bottom of the pitfall in shade. Figure 5 illustrates a pitfall with a funnel and a removable chamber for easy extraction of specimens; Figure 6 illustrates a pitfall and funnel *in situ*. Note that if a removable chamber is used, it must have drainage.

Whatever materials are used as pitfalls, the design should allow for adequate drainage. Metal flywire can be glued across the base of PVC pipes. Drainage in buckets is best achieved by drilling out a c. 5cm diameter hole with a hole-saw and gluing flywire over this hole. Selley's Quik-Grip is effective and long-lasting in attaching metal flywire to plastics. Note that drilling small holes into the base of bucket pitfalls for drainage is rarely adequate, and any hole more than about 4mm diameter will allow for the escape of small lizards.

Pitfalls can be used with or without drift-fences. A drift-fence is a low (25 to 30cm high) barrier of a material such as flywire, gutter guard, dampcourse material, custom-made, lightweight frames of mild steel and flywire, and even strips of cardboard or similar that act to guide small animals towards a pitfall trap. Metal flywire is probably most common as it is light, flexible, cheap and fairly durable. The mesh is also fine enough to stop even small lizards. Opaque or mesh materials seem not to differ in their effectiveness (Bancroft and Bamford in prep.). Drift-fences can increase the effectiveness of a pitfall trap by 100% (that is double the number of captures), but it should not be automatically assumed that pitfalls should be used with drift-fences. Drift-fences can be deployed in different ways as discussed below. Note that when no fence is available, simply digging a trench leading up to a pitfall can have a similar effect upon capture rate as having a fence installed. Animals appear to be attracted to disturbed ground and may then run along the trench and into the pitfall; a worthwhile area of study would be to investigate the effect of trench versus fence.

Figure 4. Lengths of PVC pipe (in this case 40cm) and 20 litre buckets used to create pitfall traps. Note drainage hole, covered with metal flywire, on base of pitfalls (photo M. Bamford).

Figure 5. Schematic diagram of a pitfall trap (with drift-fence), with a funnel and a lower chamber that can be removed for extraction of specimens (illustration M. Bamford).

Figure 6. Pitfall (in this case a 20 litre bucket) with a broad funnel to stop animals from leaping out, at the end of a single drift-fence. Note the pitfall lid propped up with a small branch to place the pitfall in shade around midday (photo T. Gamblin).

DEPLOYMENT OF PITFALL TRAPS

Pitfall traps (and drift-fences) can be deployed in many different ways depending on circumstances. Many factors need to be considered, and a universal approach is neither practical nor necessarily desirable. Some variables in the deployment of pitfall traps are discussed below.

To fence or not to fence

Pitfall traps can be surprisingly effective without drift-fences. Fences take time to set up and maintain, so where digging is easy (sandy soil), simply installing more pitfalls may be viable, and has the advantage that with the same spacing, sampling covers a larger area. At least in principle, 10 pitfalls, each 20m apart and each with a fence, compared with 20 pitfalls, 20m apart but without fences, might catch the same number of animals but the unfenced pits, because they sample a larger area, could record more species. This (and other aspects of trap deployment) could usefully be investigated. Fences can also bias results if comparisons are being made between open and dense vegetation, as fences can attract animals in open environments (the fence may appear to provide shelter, or may accumulate leaves along its length that provide shelter). In very dense heathland, drift-fences appear to have little effect upon capture rates and are difficult (and destructive of vegetation) to install. Studies on the length of the drift-fence (or length between pitfalls along a shared fence) suggest that the effect of fence length on numbers of captures is complex. Longer fences are better up to a point, and this length may be specific to species and factors such as vegetation structure. It seems that animals will only travel a certain distance along a fence before turning away, but that this distance is variable.

Despite these considerations, drift-fences are usually to be recommended, particularly where a survey is to be carried out over a short period of time and numbers of captures need to be maximised.

Layout

Pitfall traps can be deployed in a variety of layouts with respect to other pitfalls and with respect to drift-fences. There is an extensive literature on this topic but common layouts include:

- A number of pitfalls (commonly 6 or 10) spread evenly along a common and linear drift-fence of 30m in length.
- A number of pitfalls along a complex, fenced array (such as star-shaped with 30m “wings”, a central pitfall and several pitfalls along each wing). This layout is recommended in a review by McDiarmid *et al.* (2012) for maximising captures at one location.
- Pitfalls spaced 20 to 50m apart in a transect and each with an individual fence (or unfenced). Where these individual pitfalls have drift-fences, the number and length of fences can vary. In this situation, a longer fence may not be useful as animals will travel only a certain distance along a fence before turning away. In dense heathland, fences only 1.2m long were found to be just as effective as fences 2.4m in length (Bamford and Bancroft in prep.), but in more open vegetation longer fences are helpful. In the same heathland study, three short fences (that is, each 1.2m long, and arranged like a Mercedes Benz symbol around the pitfall) were found to maximise capture rates (Figure 7).

- Pitfalls (fenced or unfenced) in a grid; this can be on a grand scale (effectively several transects side by side) or can be more modest (eg. 25 pitfalls in five lines of five, with only 5m between pits).

Figure 7. An assisted pitfall trap with a three-way drift-fence (photo M. Bamford).

There is no "right" layout of pitfalls and drift-fences, and capture rates are so variable with season and daily weather conditions, and with factors such as vegetation structure and even the species being sampled, that there is no benefit to a universal approach. The layout is best tailored to the project and environment. Pitfalls in a tight grid or along a common fence provide almost point data that can be compared between sites, while pitfalls in a long transect provide continuous sampling across gradually changing landscapes (such as a catena of soils and related vegetation from low to high in the landscape). Where the aim is to maximise the number of species recorded, pitfalls deployed in long transects will probably be most effective as they sample more of the environment than point sampling. Transects have also been found to provide more consistent data between replicates in the same vegetation type than traps placed in quadrats (Reed *et al.* 1988).

Timing the use of pitfalls

Pitfalls work at any time of the year for everything, but are better at some times for different groups. They don't have to be operating at maximum capture rates to be effective; you may get rare species you might not see otherwise at a time of year when capture rates are generally low. So don't get too hung up on timing. However, if you have a specific target, you need to understand the biology of the fauna you are studying to get the best out of pitfalls. In southern Western Australia, virtually all reptiles are most active (and therefore most likely to fall into pitfalls) in spring and sometimes through until early autumn (generally November to December (Figure 8) but a little earlier in warmer/drier areas). Many frogs are most active in spring but only following rain. Small mammals (eg. *Pseudomys*, *Sminthopsis* but not *Tarsipes*) tend to be spring breeders in southern Western Australia. With *Sminthopsis*, captures are high during mating (August around Gingin but July around Eneabba), low during spring when males are not very active and females are carrying pouch young, then high in late spring/early summer as juveniles emerge and start to run about.

In the Pilbara, deserts and Kimberley, pitfalls may be most efficient in late summer to autumn if there have been rains, but these are hard times for sampling. In the Kimberley, we have had excellent capture rates in May and June when conditions were moderately cool but there was still enough moisture to ensure fairly high levels of fauna activity.

Seasonal variation in capture rates of reptiles near Gingin (after Bamford 1986)

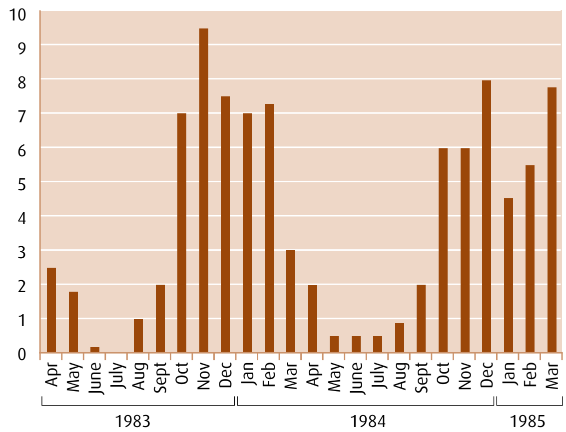

Figure 8. Values are the average number of captures per grid of 50 pitfalls over 5 nights from April 1983 to March 1985. Sampling in January and February 1985 coincided with unseasonally cool weather.

Trapping periods

The effectiveness of pitfall traps declines with time (Thompson *et al.* 2005). It is usual to talk of sampling effort in terms of trap-nights, and after three or four trap-nights, numbers of captures often fall off markedly. This appears to be because animals are attracted to disturbed soil around a newly set or opened trap. However, if there is a change in weather, such as rainfall or an increase in temperature, numbers of captures can increase towards the end of a trapping period. Trapping periods commonly used range from four nights to as much as ten nights, but as with trap layout there is no "best" trapping period. Leaving traps open for longer will catch more animals despite the decline in rate after four nights; but consideration can also be given to installing extra traps and running them for four or five nights, or even running the traps for two short trapping periods rather than one long period, with a fortnight break in-between. More than five nights trapping can result in a slight increase in mortality from small mammals being caught a second time within a few days, and there may be an increased chance of predators finding the open traps.

Numbers of pitfall traps

In theory and because of enhanced capture rates in the first three to four nights, it will be better to run more traps than to leave a smaller number of traps open for longer, but the number of traps that can be run will depend on many factors. Even under perfect conditions and high densities of animals, capture rates of greater than one animal per trap per night are very unusual; rates are often more like 0.1 animal/trapnight. Therefore, large numbers of traps are needed to record many species and to get sample sizes that may be useful for analyses. With good access, an experienced team and mild weather so traps can be checked as late as five or six hours after sunrise, as many as 400 pitfall traps have been operated at once, but most projects operate only about 100 pitfalls.

PITFALL TRAP MANAGEMENT

Setting

Pitfall traps are installed by digging a hole, which is easy in sand but can be very difficult in rocky landscapes. However the creation of a hole is achieved (post-hole shovel, hand or hand-help power-auger, vehicle-mounted auger and even blasting), an important consideration is minimising the environmental impact of installation and trap placement. The use of heavy digging equipment in fragile vegetation is inappropriate. The combination of post-hole shovel, crowbar and a metal scoop (elliptical tins such as those used for some tinned fish, with one side flattened to make a hand-hold, are very effective in this role) is very efficient. When selecting trap locations, try to ensure minimal direct damage to vegetation and try to place the pitfall where it will receive shade from late morning to mid-afternoon. It should go without saying that the pitfall should be set with the lip level with or slightly below ground level. However if this is not possible, building a ramp of earth up to the pitfall lip will assist captures.

Always ensure there is some sand and leaf-litter in the bottom of a pitfall. This calms animals and offers them some protection from cold, heat and predators (see mortality management below). Other additions to a pitfall that may give captures some protection are cotton wool, egg cartons, cardboard tubes and stubby holders.

Checking

Should be done usually once a day in the morning (but see managing mortality below), although the frequency of checking pitfalls (and any traps) depends a lot on conditions and the sorts of animals being caught. Petit and Waudby (2013) recommend checking pitfalls before dawn (to remove mammals) and several times during daylight, but note that checking frequency is dependent upon the conditions that can be maintained in the pitfalls. Such a regime is not necessary unless weather conditions are extreme (hot or cold), or there are likely to be other mortality issues such as from animals interacting in pitfalls. Reptiles caught during the day should be able to shelter in sand and litter in the pitfall overnight, but an afternoon check and removal of such reptiles would be needed if there was risk to these reptiles from ants and even from mammals trapped the following night. Checking twice a day (or more often) increases trampling damage and disturbance around pitfalls.

Checking of traps should be complete by late morning so that animals are not confined for too long; especially in summer as heat can build up in traps. However, the time when traps should be checked is not simple. If many mammals and/or frogs are being caught overnight, and conditions during the day are hot and dry, then traps should be checked very early in the morning. However, if many reptiles are being caught and diurnal temperatures are high, most will be caught in mid-to-late morning, so it is better to check traps later in the morning (as reptiles caught mid-morning when traps are checked very early will be in the trap over the heat of the day). If both mammals and reptiles are being caught, and it is hot, then consider checking both early then late morning (but this is very demanding of personnel and means repeated disturbance of traps). Reducing the number of traps open (or increasing the number of personnel) so that all traps can be checked in a short window of time before about 10:30am may be an effective compromise to minimise mortality of small mammals and reptiles. Under extreme weather conditions, consider closing traps (or not opening them in the first place).

Remember to get around the traps quickly... don't go off birdwatching or spend too much time admiring your catch! Identification of the catch should be immediate (with the animals then being released) or animals should be retained for later examination.

Opening and closing pitfall traps

Buy the best quality, UV stabilized lids and be prepared to replace them when they begin to perish. Those designed for buckets click firmly into place and, if covered with of soil when the trap is closed, are still flexible and secure after two decades (and counting). They can (and should) be hard to remove; a pair of pliers helps. Levering the lid off with a shovel or screwdriver can break the lip of the pitfall. If pitfalls are left for long periods of time in a monitoring project, they need to be checked intermittently to ensure no lids have collapsed or been removed by wildlife or people. Once a year is adequate but immediately after a fire (even pits covered with soil can be damaged and the lid collapse). If pitfalls are to be left for many years unchecked, fill them with sand/rocks. When closing pitfalls, be absolutely sure that all have been closed.

If a pitfall sampling program is finished, pitfalls should be removed from the soil and the hole back-filled. Pitfalls should be cleaned as much as is practical in the field to avoid moving soil and plant material around; if using the buckets, cleaning also helps to ensure they do not stick together when they are stacked. Special care with cleaning of pitfalls (and of digging equipment) is needed if there is a risk of dieback contamination. Between field trips, pitfalls should be cleaned thoroughly in warm water and detergent.

Removing animals from pitfalls

When checking traps and removing animals; be gentle. Before attempting to remove animals from a pitfall, check very carefully for scorpions, spiders, centipedes, etc. Use a stick or pole to check under sand or objects before putting your hand in. Small mammals can be scooped up in a cloth bag inverted over the hand. Reptiles can usually be pinned to the floor or side of the pitfall, although venomous snakes require a different approach. See Animal Handling summary (page 24) for more information.

Mortality management

Once in a pitfall, the animal is trapped and your responsibility. Realistically, achieving mortality rates of <2% (that is 1 in 50 captures) is good; a mortality rate of <1% is better and achievable. However, it must be accepted that some mortality will occur during pitfall sampling and sometimes things are beyond your control. Once all the expense of setting up a field program has taken place, you can't easily close everything up and go home. Following are some guidelines for minimizing mortality.

- **Heat.** You can operate pitfalls at any time of the year BUT have to manage them accordingly. If it is very hot (regularly over 40°C), check them early and often, and consider closing them by mid-morning and re-opening them in the late afternoon. Better still, don't trap when it is that hot. Pitfalls should be placed so they will be in the shade through the middle of the day (for example, on the south side of a bush or tree). A leafy branch pushed into the ground on the north side of a pitfall will help. Shelter placed inside the pitfall may be of some assistance but will not keep the heat out of the pitfall. Such shelter can include soil, leaf-litter, sections of an egg-carton, or sections of cardboard tube. Some scientists use custom-made funnels that place the pitfall in the shade, or stand the lid on a pole above the pitfall (Figure 6); these do help to keep the pitfall temperature down.
- **Rain.** Consider closing the pitfalls. Drainage holes can only do so much. Lumps of wood or other pieces of floating material (cork and Styrofoam cups are suggested by Petit and Waudby 2013) in the pitfall can provide somewhere for captured animals to sit. It is the water that collects in the pitfall, rather than the rain itself, that causes the problem. We've had dry Honey Possums in pitfalls after 20mm of rain overnight; but these pitfalls had good drainage holes.
- **Cold.** This can be a killer for rodents. Soil and leaf-litter in the pitfall may help. If you have a site with rodents, avoid sampling in really cold conditions (minima <5°C); rodents killed by cold will usually be dead before dawn but if they are still alive, even just, they can be brought back by putting them inside your jumper. It is amazing what a little body warmth can do. Pieces of egg carton and sections of cardboard tubing may provide some protection from the cold for small mammals (Figure 9). Cold drink holders ("stubby-holders") are very effective in providing rodents with an insulated refuge in pitfalls. They also float in the event of flooding.
- **Ants and other invertebrates.** Close pitfalls where ants are a problem, and select locations with ants in mind. Ants can be very determined, and while powder/ant sand will discourage them, the impact of ant powder upon captures is unknown. Ants tend to be worse when it is warm to hot, so trapping when conditions might be slightly cooler than will give you maximum capture rates could be an option. There are other invertebrates that cause problems (centipedes, carab beetles, trapdoor spiders, scorpions); in general these are most active when it is warm to hot. For example, carab beetles are active and frequently caught on the northern coastal plain in November and December, so consider trapping in October/November (reptile captures will be quite good but you will get very few *Sminthopsis*) or even March/April (low reptile captures but some autumn active species, and you'll get juveniles as evidence of breeding, as well as more *Sminthopsis*).
- **Multiple captures.** Aiming for maximum numbers of captures per pitfall is not always desirable and can increase mortality. A third of deaths in one study (M. Bamford unpubl. data) were due to high capture rates with several animals in a pitfall at once. In this regard, rodents killing lizards can be a particular problem. Therefore, reducing trap efficiency (such as not using drift-fences but installing more unassisted pits) can be recommended.
- **Predators.** Predators will occasionally attempt to get at trapped animals. Foxes, cats, varanid lizards and large snakes (Figure 10) have all been reported removing trapped animals from pitfalls. This seems to happen rarely but is something that should be monitored. It may be more of a problem with extended trapping periods, repeated trapping at the same locations or having the traps close together rather than widely spaced. Motion-sensitive cameras can be set on traps where such interference is suspected.

Figure 9. A Honey Possum *Tarsipes rostratus* torpid and sheltering in an egg carton provided in a pitfall trap (photo T. Gamblin).

Figure 10. A Mulga Snake *Pseudechis australis* preying upon a Little Long-tailed Dunnart *Sminthopsis dolichura* in a pitfall trap (photo M. Bamford).

FUNNEL TRAPS

Funnel traps are a fairly recent (late 1990s) innovation in fauna sampling in Western Australia, but have been used in North America for a longer period (McDiarmid *et al.* 2012). They are more efficient than pitfalls at catching legless lizards, small snakes and some of the active skinks like *Ctenotus*, but they are less efficient at catching fossorial skinks and small mammals. They can be used on rocky surfaces and over waterlogged soil where pitfalls cannot. Animals are caught by entering the trap then being unable to find their way out (referred to as a confusion trap design, like a craypot).

MATERIALS

The only commercially available funnel traps are those developed and introduced by the late Jason Fraser. These consist of green shade-cloth over a wire frame, can be opened and folded up like a concertina, and have a zip the length of the trap (see Figure 11). They are usually used by being set along a drift-fence and with shadecloth and/or leafy branches placed over them to provide shelter. They are convenient to use but have a number of disadvantages. For example, there are mortality issues (see page 18 on managing mortality in funnel traps), they can be fiddly traps to set and check, and rodents (and large invertebrates) can chew holes in them. These aspects of using funnel traps are discussed below. Note that there may be other funnel traps designs available from overseas, or that could be developed in Western Australia, that may be better than the funnel traps in use.

Figure 11. Commonly-used design of funnel trap with two Mulga Snakes (photo M. Bamford).

DEPLOYMENT OF FUNNEL TRAPS

Funnel traps are almost always set along drift-fences (drift-fences are described and discussed under Pitfall Traps, page 12) although they will catch animals without a fence being installed. They are commonly used in conjunction with pitfall traps along drift-fences, but can be used without pitfalls, such as where pitfalls cannot be installed. However, the combination of pitfalls and funnels on a common drift-fence is very effective.

Layout

Funnel traps can be deployed in a variety of layouts but as they are often used as a supplement to pitfalls, the funnel layout is often determined by the pitfall layout. Two major types of funnel layout are:

- Funnels placed at intervals along a common drift-fence, either linear or in some other arrangement (with or without pitfalls).
- Funnels placed individually or in small numbers along short drift-fences. The number of such arrangements is limited only by imagination. For example, it is possible to simply have a drift-fence of 5m with a single or pair of funnels in the middle, such a drift-fence with funnels at each end, or in combination with a pitfall trap. A sampling unit we use successfully is to have a pitfall trap with a three-way fence, and to have one fence extended by several metres and a funnel in that fence (Figure 12). The sampling unit is then the combination of funnel and pitfall. Note there may be interference between the two trap types, with an animal getting caught in the pitfall before it encounters the funnel, or vice versa.

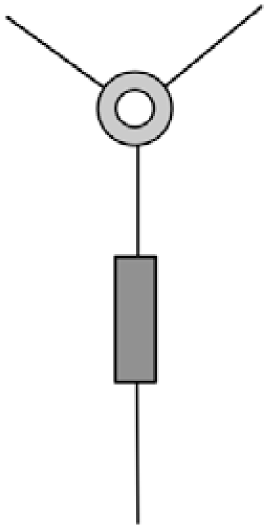

Figure 12. Combination of a pitfall, three-way drift-fence and funnel trap (illustration M. Bamford).

Timing the use of funnels

Funnels can be disappointing a lot of the time, and often seem to be effective only when reptile activity levels are high. Funnels are time-consuming to set and check, and so are best used only when they are going to contribute substantially to a survey. Therefore, use of funnels in southern Western Australia is limited to late spring through to autumn. They are probably effective for a greater proportion of the year the further north the site, and can be effective year-round in the Kimberley. Note there are issues with using funnels during hot weather (see 'Mortality management' on page 18).

Trapping periods

The effectiveness of funnel traps appears to decline with time as it does with pitfall traps, although this has not been well-documented. In reality, trapping periods with funnel traps will often be determined by trapping periods for the pitfalls.

Numbers of funnel traps

When funnel traps are effective, they can achieve capture rates in excess of 1 animal/trapnight. However, this falls off rapidly in cooler weather to rates as low as ca. 0.02 animal/trapnight even in moderately warm weather when pitfall traps are still very effective. Funnel traps are often used in the same numbers as pitfall traps.

FUNNEL TRAP MANAGEMENT

Setting

Funnel traps can be placed in two different ways along a drift-fence: against the fence or with the fence running into each entrance. When placed against the fence, they are often used in pairs (Figure 13), with one funnel on either side of the fence. This arrangement is easier to check and re-set than when the fence is pushed into the funnel entrances (Figure 14), and is the only way a funnel can be used when rigid fences are used. However, having a floppy flywire fence pushed into the funnel entrance appears to result in higher capture rates. When set, the funnel trap should be firmly against the fence or with the fence firmly pushed into the entrance, as this helps to guide animals into the trap. Funnels should always have shade placed over them: this can be a hessian bag, high density shadecloth (eg. 90%) or even vegetation. If at all possible, funnels should also be in a position where they get shade through the middle of the day from overhanging trees or bushes. When setting a funnel, check the zip is closed and check for holes. Holes can be temporarily fixed with gaffa tape (ensuring there is no sticky side where an animal might get trapped) and permanently fixed by being stitched up. Even with repairs, the life of funnel traps is usually short, largely due to rodents chewing their way out.

Bait is not used in funnels (they are an interception trap and bait will only attract ants). If large numbers of frogs are being caught, moist earth, cloth or paper-towel should be placed inside the funnel as frogs dehydrate very quickly in these traps.

Figure 13. Funnel traps set in an offset pair either side of a drift-fence. Note shade (photo A. Bamford).

Figure 14. Funnel trap set with fence pushed into entrance; this appears to result in higher capture rates per funnel trap than when the funnel is set against the fence as in Figure 13. Note sand ramped into entrance of funnel (photo T. Gamblin).

Checking

Should be done once a day, or twice a day under some circumstances (see Mortality management on page 18). Checking of funnels is usually linked to the checking of pitfall and possibly other traps, but they can require their own schedule. Reptiles are caught in funnels during the day and at night, while frogs will also be caught in funnels; generally at night. Therefore, checking in the morning should be adequate for the management of captures, but flexibility may be needed.

If frogs are being caught, funnels should be set appropriately with some moist material in the funnel and checked as early as possible. Because funnels enclose captured animals in a mesh cage, they are not able to bury themselves or shelter under material as readily as they can in a pitfall. If many frogs are being caught then it may be necessary to remove funnels from the trapping program. In contrast, if reptiles are being caught then it may be better to check funnels later in the morning; many reptiles will be caught a few hours after sunrise, and if the weather is hot it is important that these animals not be confined in the funnel through the hot part of the day. Such animals are likely to be caught after an early morning trap-round.

Checking funnels twice a day is necessary in hot weather; and it is often under such hot weather that funnels are most efficient. The procedure is to check the funnels in the morning before it gets hot (this will obviously vary, but within 4 hours of sunrise should be sufficient), to leave the zip open and the funnel on a bush or in some other position where it cannot accidentally entrap an animal, and then to return in the late afternoon (at least three hours after noon) and re-set the trap. This avoids the hot part of the day but means that the traps are still catching for the main period of reptile activity.

Funnel traps should be checked very carefully. Before picking up a funnel trap, make sure it does not contain a snake. Small lizards can hide in the folds of shadecloth, and funnels also catch scorpions, spiders and centipedes. It is usually necessary to pick up a funnel trap and examine it carefully, such as by holding it up so light shines through the shadecloth (Figure 15). As with other trapping, it is important to get around funnel traps quickly and to either identify (and then release) the catch immediately, or to retain animals for later examination.

Figure 15. Checking a funnel trap for small lizards that might be concealed in folds of shadecloth (photo T. Gamblin).

Opening and closing funnel traps

Check very carefully that no animals are concealed in each funnel as it is closed. Apart from vertebrates, even small beetles left in a funnel will chew a hole in the fabric to escape. As with other trap types, make sure that all funnels are collected. Funnels (and their shades) are notorious for picking up seeds, so ensure that seeds and other debris are shaken from the funnel before it is folded up and taken to another site.

Removing animals from funnels

When checking traps and removing animals; be gentle. Before attempting to remove animals from a funnel, check very carefully for scorpions, spiders, centipedes and other invertebrates. The approach to removing an animal from a funnel trap will depend upon what it is. Venomous snakes can usually be identified without being handled and therefore can be released by gently opening the zip with long forceps, wire or a stick and allowing the snake to find its own way out. Do not try to drag a snake out of a funnel either using a snake hook or similar device, or by the tail. Lizards can be difficult to remove from funnels as they are often very active and the funnel design limits access. Pinning a lizard down through the mesh with one hand even before opening the zip is recommended. See Handling and Releasing Invertebrates (page 24) for more information.

Mortality management

Once in a funnel trap, the animal is trapped and your responsibility. As noted under pitfall traps, some mortality is inevitable but it can be kept to a minimum. Following are some guidelines for minimizing mortality.

- **Heat.** Funnel traps are generally only effective for catching reptiles under warm to hot conditions, and it is under such conditions that mortality is likely to occur. While the official maximum may only be 35°C, the air temperature at ground level will be more than this, and the temperature in a funnel trap in the shade will be about the same as that at ground level. To make the situation worse for reptiles in funnel traps, they are unable to shed body heat by burying themselves into sand, as they can do in a pitfall trap. Heat-induced mortality of reptiles will begin to occur at air temperatures in the trap over 35°C. The best way around this is not to have traps set during hot conditions. This can be achieved by leaving the funnel trap open during the hot part of the day (as outlined above under trap checking), but under conditions where this is not possible, funnel traps should not be used.
- **Dehydration.** This is a concern for frogs. Placing moist soil or moist material (cloth, paper towel) in the funnel will help, but this can dry out quickly. Where large numbers of frogs are being caught and dehydration is a risk, use of funnel traps may not be appropriate unless only a small number of traps is being used and they can be checked within an hour or so of sunrise.
- **Rain.** Not generally a problem with funnel traps as mammals are rarely caught and drowning is not an issue. However, funnel traps should be set where they will not be impacted by surface runoff.
- **Cold.** Not generally a problem with funnel traps as mammals are rarely caught, and reptiles and frogs are tolerant of low temperatures. Freezing temperatures would be an issue but neither frogs nor reptiles are likely to be active under such conditions.
- **Ants and other invertebrates.** Funnels should be located to avoid obvious ant nests. Ants can be very determined, and while ant powder/ant sand will discourage them, the impact of ant powder upon captures is unknown. Unfortunately, funnel traps are most effective for catching reptiles under warm to hot conditions when ants and other invertebrates are also at peak activity.
- **Multiple captures.** Multiple captures in funnel traps can lead to mortality; some reptiles in the breeding season are aggressive towards each other, for example. Such deaths have been recorded with the skink *Ctenotus fallens* (a funnel trap with one female and two male specimens; one of the males dead and badly bitten by the other male), but this is probably unusual. Monitoring multiple captures on a field trip by field trip basis and removing traps if an issue is identified is probably the only course of action that can be taken. Avoiding peak activity periods usually results in very low capture rates with funnels.
- **Predators.** Predators will occasionally attempt to get at trapped animals. We have one record of a feral cat that killed a lizard (*Egernia depressa*) by biting it through the mesh of a funnel trap. This trap was not replaced in that trapping session and no further problems were encountered at that site.

BOX (EG. ELLIOTT / CAGE) TRAPS

There is a wide range of what are effectively box traps with a trigger mechanism of some kind. They are attraction traps that rely on the use of bait to lure annals into the trap, unlike pitfall and funnel traps that are interception traps that rely on animals entering the trap passively. Cage and Elliott traps are probably more widely-used than they should be, given that they are useful for only a few species of mammals (rodents, bandicoots, possums and some dasyurids) and are effective with a few reptiles. Furthermore, several of the species that can be caught with these traps are often more accurately and safely surveyed by using non-invasive approaches (see page 29). However, there are some occasions when they are useful, such as when animals actually need to be caught for the collection of biological data, or for translocation.

TYPES OF BOX TRAPS

Box traps come in a range of sizes and styles and they need to be appropriate for the species being sampled. Elliott traps (Figure 16) have solid aluminium sides, are collapsible and are named after the man who developed them. There are also traps made by other manufacturers in the same style and at various sizes, such as Sherman traps. A somewhat similar trap, the Longworth (Figure 17), is suitable for very small mammals but seems rarely to be used in Australia; the Elliott is simpler to assemble. These small aluminium traps usually come in a carry box that takes 10, 20 or 25 traps. Cage traps have entirely (or in some makes partly) wire mesh sides; some are rigid and some are collapsible (Figure 18). All these traps rely on a trip-plate that is linked to the trap-door, and use bait to lure animals into the trap.

Figure 16. Elliott traps (carrying box, trap folded and trap open). Note that when stored in the box, it is helpful to arrange them in sets of five for easy counting (photo M. Bamford).

Figure 17. Longworth trap, a small aluminium trap widely used in Britain (photo M. Bamford).

Figure 18. Cage trap (with Brush-tailed Possum). Note sack that has just been removed from over the trap (photo M. Bamford).

DEPLOYMENT OF BOX TRAPS

Box traps can be used in combination with pitfall and funnel traps or quite separately.

Layout

This can vary greatly depending upon whether or not other traps are being used, and on what animals are being targeted. For example, a sampling layout used in some Department of Environment and Conservation (now Department of Parks and Wildlife (DPaW)) studies uses a grid, referred to as the Kingston Layout, of assisted pitfalls (with drift-fences), cages and Elliott traps (Figure 19). Elliott traps can also be placed against the end of a drift-fence, in which case they will often catch small mammals and lizards that might not otherwise enter the trap. However, cage and Elliott traps are often used independently of pitfalls even when part of the same study. Where pitfalls are used in a transect at a spacing of 20m, an Elliott trap might be placed under a bush close to every second pitfall, and a cage trap under a bush close to every fifth pitfall. Spacing of cage and Elliott traps is important as often the target animals occur at low density, and thus placing the traps close together will limit the number of individuals that might be caught. A common layout for Elliott traps when targeting rodents such as *Notomys* spp., which are not usually caught in pitfalls, is to place the Elliott traps at 25m intervals in transects of 10, 20 or 25 traps (the number used may simply be determined by the number of traps in a box). Cage traps usually target larger species (bandicoots, possums, Chuditch) and therefore spacing can be greater. The DPaW uses a spacing of 200m for cage traps set for Chuditch. However, the Quenda (Southern Brown Bandicoot) tends to occur at high densities in patches of favourable habitat, so traps at 25-50m spacing is more appropriate, and often in a grid to cover the habitat patch. The important thing is to plan the layout to suit the species and the project.

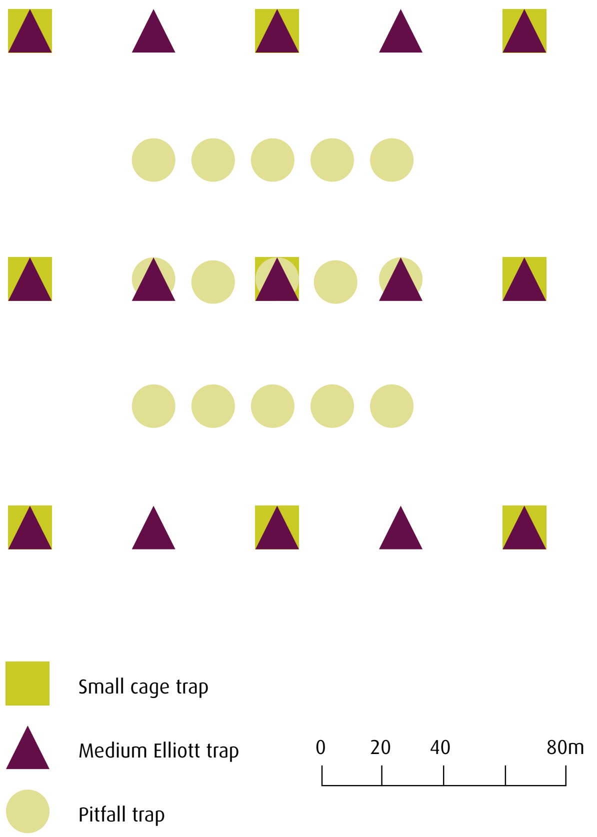

Figure 19. The Kingston Layout, an example of a grid layout that combines several different trap types. Funnel traps can be added to the drift-fences along the pitfall traps.

Timing the use of box traps

These traps are effective at any time of the year, although their effectiveness probably varies seasonally; for example, juvenile animals may be more willing to enter traps than adults and thus trapping after the breeding season may be most fruitful. There are limitations to using Elliott traps in particular during hot or cold conditions (see below), and trapping should be avoided at some times of the year if marsupial species inclined to drop their young may be encountered.

Trapping periods

Cage and/or Elliott traps are often used in conjunction with other trap types, and thus the trapping period may be determined by the performance of these other traps. Therefore, cage and Elliott traps are often run for periods of 4-7 nights. Pre-baiting (using baited traps with the door mechanism disabled or simply putting out bait before the trapping period) can be effective at increasing capture rates by "training" animals to accept the bait and the trap. Individual animals sometimes learn that traps mean food and get caught in successive nights and this may limit the trapping period; "trap-happy" animals can also interfere with the capture of more cautious species.

Numbers of cage and Elliott traps

This is dependent upon the project and the target animal. Capture rates are usually very low (often <5% or 1 animal per 20 trapnights) so many traps (100-200) may be needed, but the number of traps may be limited by the time required to get around them all, especially if conditions are warm to hot. Capture rate can occasionally be very high such as when targeting Quenda in ideal habitat, or in studies on island populations where high densities are present.

BOX TRAP MANAGEMENT

Setting

These traps should always be placed under cover such as dense vegetation. They must be in shade at all times. Cage traps should have a sack or something similar over them so trapped animals are sheltered and feel somewhat secure. Placing material inside the trap may interfere with the mechanism, but Longworth traps have a shelter compartment where nesting material can be placed. Beware of nearby ant nests. The use of sacks as cover or the use of nesting material inside traps raises the issue of trap hygiene (discussed below). The setting of traps for specific species can vary. While almost always set on the ground (in Western Australia), they can be set in trees if phascogales are being targeted (although both phascogale species are regularly caught in Elliott traps set on the ground). Setting traps in trees is very time-consuming, requiring a bracket installed in the tree, and a step-ladder or similar is needed when checking the traps.

When setting traps, ensure that the trigger mechanism is operating correctly. Collapsible cage traps in particular are prone to getting bent wires or having the trapdoor distorted so that it does not close properly. Careful packing of traps at the end of a field trip is important to avoid such damage. In Elliott traps and some styles of cage traps, the trapdoor is contained within the body of the traps and is sprung, but in many styles of cage traps the trapdoor falls across the entrance through the weight of gravity, and has a locking bar to ensure the door cannot be pushed open. Check that this works and that vegetation or stones will not interfere with the closing of the door.

As with other traps, cage and Elliott traps should be clearly marked and numbered in the field, such as with coloured flagging tape (or metal posts for long-term sites). Note that cage and Elliott traps are easy to remove from a site and members of the public regularly steal such traps if they find them. This can be a problem even in remote areas, but is a major issue close to towns and urban areas. In areas where interference from the public may occur, using a marking system such as having the flagging tape 10m on a particular compass bearing from the trap can be surprisingly effective. Interference can also come from corvids (ravens and crows) and magpies; these birds will learn to remove the pin of Elliott traps to access the bait. The pin can be taped into place but it may be necessary to open traps late in the afternoon to avoid such interference.

Bait

Cages and Elliott traps are attraction traps and are almost always baited (they will catch animals without bait but it is unclear if this is because the trap smells interesting or because animals move into the trap in search of shelter). Universal bait (peanut paste, rolled oats and sardines) is widely used and effective for a surprising range of species, but people have their own recipes. Some such recipes have been developed to be species-specific or to exclude "nuisance" species, such as lizards that may get caught during the day when the traps are needed at night for mammals, or common mammals that set off traps. Truffle oil soaked into paper towels can be effective for some mycophagous mammals, but we have found it not to increase captures of the Quenda. In some areas, it may be necessary to trial baits as high meat content may attract ants and often the mammals being targeted are attracted by peanut paste or fruit. Some scientists re-bait daily while others only renew bait when it has been lost from a trap. There is probably no advantage to daily rebaiting but the bait should be checked daily and refreshed as necessary.

Checking

Should be done once or twice a day, depending on conditions and the animals being caught. Checking traps once a day (morning) is suitable if the chance of catching animals during the day is low. If, however, animals of any kind (target mammals or possibly non-target reptiles) are likely to be caught during the day, then traps need to be checked in the late afternoon also. If conditions are hot, the best approach is to close after the morning check and then re-open them in the late afternoon. This doubles the amount of time spent checking traps but ensures animals are not inadvertently killed during the day, and that all the traps are open to catch mammals overnight. Very occasionally, it may be necessary to check traps through the night, such as when mammals are very abundant or a common species sets traps off so that a rare species cannot be caught. This can mean checking traps 4-5 hours after sunset or even several times during the night.

Collecting, packing and cleaning box traps

As with other traps, cages and Elliott traps should be collected and counted to ensure no traps are left in the field. If they are left out, they will eventually capture and kill an animal. Both cage and Elliott traps can be soiled with uneaten bait and excreta, and must therefore be cleaned. This can be difficult in the field, so dirty traps can be marked, such as with a piece of flagging tape, for later sorting and cleaning. Elliott traps can be unfolded by removing a long pin from one side to facilitate clearing. This is also useful when adjusting the trigger mechanism if it becomes bent. Sacks used to cover cage traps may also be soiled and there is some concern about the potential transfer of disease between sites as a result. Bags can be washed (such as in a weak (1%) bleach solution as suggested by Chapman *et al.* 2011) and sterilised by lying them in the sun. Clean bags should be used if going to isolated or remote areas, or those with highly significant populations.

Elliott traps pack away into custom-made boxes. When using and packing Elliott traps, mark any that are damaged or not operating well for later repair/adjustment. Collapsible cage traps should be neatly tied in bundles (usually five) and ensure that hooks are secured and the back door of each trap is tied down, otherwise it can unfold and get damaged.

Removing animals from box traps

The sorts of animals regularly caught in cage and Elliott traps include small and medium-sized mammals that can be very sensitive to noise, light and fast movements. A quiet approach is therefore required. When approaching a cage or Elliott trap that has been set off, confirm what animal (if any) has been caught. This is easy in cage traps but in Elliott traps with solid sides, this can be done by looking at the animal through crevices in the hinges. Note that snakes are often caught in these traps so identifying the type of animal present BEFORE opening the trap or trying to extract the animal is important. After confirming that the animal is not a snake, you can peek into the entrance of the trap to check what is there. Examples of approaches to removing different sorts of animals from these traps are outlined below.

Small mammal in Elliott trap and similar aluminium traps (also works for harmless lizards)

Carefully pick up trap and place a cloth bag over the end with the trapdoor. Gently tilt the trap onto its side so that the animal is on the side of the trap. Press open the trapdoor through the cloth bag until it engages with the trigger mechanism and clicks into the open position. The door will then stay open and the animal, because it is walking on the side of the trap, will not set the door off. Do not try to shake the animal into the bag but try to encourage the animal to enter the bag such as by gently tilting the trap down towards the bag. Gentle tapping on the opposite end of the trap and blowing into the opposite end of the trap can also work. Once the animal is in the bag, close off the top of the bag. Note that a sturdy, clear plastic bag can be used in place of a cloth bag; this allows the animal to be identified readily but gives it less security. Animals cannot be stored in plastic bags.

Small to medium-sized mammal in cage trap

The approach is similar to outlined above, but generally tilting the tap is ineffective as the mammal can grip the mesh sides of the trap, whereas in an Elliott it will tend to slide towards the bag. Remember that the animal is more exposed because of the mesh sides, and the door/trigger mechanism will differ. Larger mammals may also try to bite though the cloth bag. Medium-sized mammals can be very cautious about leaving a trap even to enter the apparent security of a bag. Give them time and make sure that other people are quiet and behind you so that by moving into the bag the animal is moving away from you and other people.

Snakes in Elliott traps

This happens surprisingly often; snakes are probably attracted by the mammal smells around well-used (even when well-cleaned) traps. Even non-venomous snakes are difficult to remove from Elliott traps as they can wrap themselves around the trigger mechanism. The safest approach is to place the Elliott on the ground close to cover and to remove the long pin that holds the trap together. This is hooked at one end but not on the other, so can be pulled out with pliers. Use a smooth action so that the trap stays in shape; you may have to step back and flick the top of the trap with a long stick to make it unfold. The snake will then almost always move into the nearby cover. Make sure onlookers are out of the way! Note that in some Elliott-type traps (not authentic Elliott traps but made in the same style as to be almost indistinguishable), the long pin is bent over at both ends and cannot be removed. These traps should be modified by cutting off one bent end before use.

Snakes also sometimes get caught in cage traps. Those styles of cage traps with a drop-down trapdoor that is not spring-loaded can be placed on one side and the door pulled open with a stick, then left for the snake to find its own way out. Cage traps with spring-loaded doors can be more difficult but often the spring system can be disabled so the door will stay open. Trying to remove snakes from traps forcibly will just anger and possibly injure the snake, and place the handler at risk.

Mortality management

Once in a cage or Elliott trap, the animal is trapped and your responsibility. As noted under other trap types, some mortality is inevitable but it can be kept to a minimum. Following are some guidelines for minimizing mortality.

- **Heat.** Animals in cage and Elliott traps may be exposed to extreme heat even when the trap is well-shaded. This can be a problem even when the official maximum temperature is <35°C. For example, the temperature in an Elliott trap can become higher than the surrounding air temperature due to the poor ventilation and heat generated by a captured mammal. Cage and Elliott traps can still be used when such maxima are experienced, but they should be closed after the morning trap-round and re-opened in the late afternoon. This also avoids by-catch of reptiles. If this is not practical then cage and Elliott traps should not be used. As these traps work at any time of the year, it is often possible to plan the use of these traps when weather conditions are most suitable.
- **Dehydration.** Not generally a problem with cage and Elliott traps as frogs are rarely caught.
- **Rain.** Mammals caught in Elliot traps have some protection from rain, as do mammals in cage traps that are covered with a sack or similar. Very heavy rain may be a concern. Run-off can also be a concern and if rain is expected (or occurs), the positioning of traps will need to consider this.
- **Cold.** This can be a problem for small mammals, especially in Elliott traps where the trapped animal is sitting on a sheet of metal. A piece of cardboard, cloth or cotton wool can be used in both cage and Elliott traps to provide shelter, but may interfere with the trigger mechanism and the traps take a lot more effort to clean at the end of the survey. However, such strategies may be needed if cold conditions are expected and small mammals are likely to be caught. Note that small rodents are especially susceptible to cold in Elliott traps.
- **Ants and other invertebrates.** Ants are occasionally attracted to the bait in cage and Elliott traps, and will attack small mammals. Traps should be set away from ant nests and if ants are attracted to a trap, move it and/or scatter ant deterrent around the trap whether or not any mammals have been caught. As cage and Elliott traps work for much of the year, hot seasons when ants are most active can sometimes be avoided.
- **Multiple captures.** Multiple captures of two or more adult animals in cage and Elliott traps are very unusual but do occur and can result in the injury or death of at least one of the trapped animals. This is probably unavoidable. Multiple captures involving pouch young of female marsupials present a separate case and can be avoided through timing of sampling periods. The aim is to avoid catching females with moderately large to large young which might then be ejected. This involves understanding the breeding season of the species that might be caught. In some circumstances, such as relocation before clearing of vegetation, there may be no choice in timing. In this case, having a prior arrangement with a wildlife carer means that if ejected young are encountered, they can quickly be taken to a wildlife carer.
- **Predators.** Predators will occasionally attempt to get at trapped animals. We have one record of a feral cat that caught its paw in the door of an Elliott trap while trying to reach in to grab a native rodent. In this case, the rodent was unscathed and the cat escaped. If predators become a problem around traps, it may be time to end the trapping session.

NETS AND OTHER TRAPS FOR BIRDS AND BATS

Birds and bats are very different groups of fauna but there is overlap in techniques used to trap them. There are existing procedures for the authorisation of people to use capture techniques for these groups through the Australian Bird and Bat Banding Scheme (ABBBS).

Unlike most fauna, birds are readily identified without the need to capture and handle them, but in some cases they need to be captured for research purposes. Mist-nets are widely used for this purpose, with other specialised capture techniques including hand-nets (often with spotlights at night), cannon nets, a range of nets that operate from springs or similar to be thrown over birds roosting on the ground (eg. clap-nets, flip-nets), traps placed over nests and the Bal Chatri trap (for birds of prey). Such capture techniques can be used effectively with very low rates of mortality; experienced bird-banders using mist-nets should lose fewer than one bird for 1000 handled. However, a lot of skill is required for the management of these nets.

Mist-nets are also used for the capture of bats, sometimes necessary to confirm identification of species detected through aural surveys (see page 32), and to collect biological data. Bats can also be caught with harp traps, with trip-lines set over water and even by hand (in caves). Bat capture techniques are reviewed by Churchill (2008). The capture of bats should only be carried out by experienced personnel and there are some health issues due to the possible transmission of bat viruses.

When used in conjunction with banding, operators are usually trained and authorised in the use of the different types of nets and traps through the ABBBS; they must also have a Regulation 23 licence to trap and mark fauna, which recognises their ABBBS certification. The ABBBS has an established system of classes of bird-banders and endorsements for different capture methods, as well as procedures for the training of people in the use of nets and bands. The training is done almost entirely by established A Class banders who mentor people as assistants in a banding project. Bird capture and banding should be carried out within the existing Standard Operating Procedures of the ABBBS, and the scheme provides a manual that details these procedures (Lowe 1989). Almost no bat-banding is currently carried out in Western Australia (due to injury issues with many bat species), but scientists using mist-nets to trap bats must also be experienced in the handling and removal of birds from mist-nets as these are a common non-target capture. The DPaW prefers all mist-net users to have trained under the ABBBS program and to hold a mist-net endorsement.

The use of nets and traps for the capture of birds and bats is highly specialised and training is best carried out through the existing ABBBS process. Some basic information about these techniques is presented below.

Mist nets consist of a net of very fine mesh with different gauges for different target bird groups. The nets come in differing lengths and heights, but all have taut lines running the length of the net referred to as shelf-strings. A bird (or bat) that flies into a mist net will drop into a fold or pocket of mesh behind a shelf string, and becomes entangled in this fold. When extracting a bird or bat from a mist net, the first step is to determine on which side of the net the animal is caught, so that extraction is not attempted through the mesh. The bird or bat needs to be reversed out of the net, and the standard approach to extraction is to first free the feet, then the tail, body, wings and head. Nets should be tensioned with a taut upper shelf string to minimise entanglement. The lower shelf string should be high enough to prevent the lower pocket touching the ground as birds in contact with the litter will become badly entangled and can be vulnerable to predators. Mist nets will not work effectively in strong winds and should not be unfurled if they are wet, or be used in the rain, as wet feathers are easily damaged in the abrasive mesh. When targeting birds, mist-nets should be checked every 10-20 minutes depending on the capture rate, and may even need to be monitored continuously under some circumstances.

Extracting birds and bats from mist-nets requires a lot of skill but less experienced people can help with setting nets and processing (measuring and recording) the specimens.

Animals caught in mist nets are very exposed to predators and the elements. Furthermore, bats will chew their way through the mesh. They may thus escape, but they can become more entangled or can escape with some mesh still around their body. Mist nets set for bats over water should be monitored continuously. Those set across flyways in dense vegetation should be checked every 20 minutes.

Net placement is important. For birds, nets are usually placed against background vegetation and where birds are expected to fly, often in the shade (where the mesh is less visible than in the sun), but for bats mist-nets are often placed over or adjacent to water. While mist-nets are designed to be used as a vertical barrier into which birds fly, they can be modified for the capture of ground-moving birds by having part of the mesh lying on the ground, and a system of ropes in place to pull the mesh up when a bird walks across it; this is not a recommended or SOP of the ABBBS. Mist-nets are not normally used on the ground as they get tangled in vegetation readily, and trapped birds on the ground can be attacked by predators.

Cannon nets, pull nets, flip nets and clap traps all pin birds onto the ground with a layer of mesh. They are commonly used for shorebirds and waterfowl, and occasionally for parrots that can be lured with grain. These nets can catch large numbers of birds at once, and cannon nets require a qualified shot-firer to load the cannons. Using these sorts of nets usually involves a large and experienced team to set the nets, encourage birds to enter the catching area, extract the birds quickly and then process all the birds in a reasonable time-frame. A team of 20 people may take most of a day to make a catch of 800 shorebirds, with processing taking 2-3 hours.

The Bal Chatri trap is one of several similar traps used to catch birds of prey. These use a lure to draw the predator onto the trap, where it is caught with either a spring-loaded net or with nooses attached over the lure.

Harp traps catch only bats and consist of fine monofilament line (fishing line) stretched vertically and tight across a frame. There may be two or three banks of these lines spaced a few centimetres apart. Bats are unable to detect the lines, hit them and then slide down into a holding area from which they can be collected in the morning. Injury rates are very low but bats do need to be removed early in the morning, with some species susceptible to exposure. Predators can also be a problem.

Figure 20. Demonstrating extraction of a Western Spinebill from a mist-net (photo T. Gamblin).

TRAPS FOR MISCELLANEOUS FAUNA

While main trapping techniques used in fauna investigations are outlined above, approaches to trapping may only be limited by imagination. For example, pitfall traps can be modified with the addition of a hinged trapdoor onto which bait can be attached; lizards walk onto the trapdoor and their weight causes the door to drop them into the pitfall. There are also some fauna groups that are not readily sampled by the main trapping techniques, and some approaches for these groups are described below.

FRESHWATER TORTOISES/TURTLES

The names tortoise and turtle are widely and interchangeably used in Australia for freshwater species, but they are not to be confused with the very large marine turtles. Several designs of freshwater tortoise trap are commercially available, often referred to as opera or cathedral traps because of their shape (some vaguely resemble the Sydney Opera House, see Figure 21). These are funnel entrance traps that can be baited (raw chicken is often recommended) and extend above the surface of the water. While freshwater tortoises can exist for long periods under water, this feature is important because other wildlife such as waterbirds and the Rakali (water-rat *Hydromys chrysogaster*) can also be caught. Simple funnel-entrance traps can be made out of wire mesh but these also need to be designed to ensure that trapped animals can breathe. When caught in such wire traps, tortoises may damage their heads by pushing against the wire, although this has not been observed in trapped *Chelodina oblonga*. The commercial traps use a soft, shade-cloth like material. Tortoise traps should be monitored closely, especially where by-catch may occur.

Note that freshwater tortoises can also be lured to within reach of a hand-held net using bait placed near the net.

Figure 21. Cathedral traps for freshwater tortoises/turtles. The entrance is at the base of the trap and the top section of the trap is held above water (photo T. Osborne of T and L Netmaking).

FRESHWATER FISH

There is a range of capture techniques available for freshwater fish. There are commercially available fish traps (usually sold for the capture of bait fish, but can be used to sample freshwater fish in small streams and lakes) that have a funnel entrance and can be baited with meat, dog pellets or even poultry pellets. Note that such funnel traps are an illegal capture method under the Fish Resources Management Act (1994) and their use to catch fish requires an exemption under that legislation. Funnel traps can sometimes catch freshwater crayfish (Gilgie, Marron, Koonac and the introduced Yabbie) that can prey upon trapped fish, and they therefore need to be monitored closely. Assorted nets are commercially available or can be manufactured to suit the need. A one-person seine net, with a mouth <1m wide, is easily made from suitable mesh and can be very effective in sampling large numbers of freshwater fish.

TADPOLES

While readily caught with small nets and in fish traps, a tadpole trap may be more effective in catching a wide range of species and in providing a standardised sample for comparative purposes. Tadpole traps can be made from plastic drink bottles, with the end cut off and inverted back into the body of the bottle to form a funnel-entrance. These can work passively to traps tadpoles that happen to swim into them, or can be baited.

HANDLING AND RELEASING VERTEBRATES

The aim of almost all catching of vertebrates is to be able to identify species present, collect biological data and obtain measures of abundance, with the animal being released once it has been 'processed'. While lethal methods of collection were widely used by scientists well into the 20th Century, such methods are now very rarely employed. Therefore, an important part of animal trapping is the safe handling of animals for both the specimen and the handler. Some method-specific notes on handling are provided above, mainly with respect to the approach to removal of animals from specific trap types. The following notes discuss general handling considerations for the major taxa, including approaches to euthanasia. Euthanasia on-site, while unpleasant, is almost always preferable to transporting an injured animal a long distance to a facility where euthanasia can be carried out by a vet. At all times, handlers should try to minimise distress to animals being handled by being quiet, gentle (but firm where necessary) and not exposing the animal to anything that may upset it further.

FROGS

Frogs do not bite or scratch and, unless dehydrated, are robust. Main points to consider when handling and releasing frogs are:

- The skin of frogs is a respiratory surface protected by oily secretions. Chemicals such as sunscreen and insect repellents may be absorbed through this skin and therefore need to be avoided.
- Frogs can be cradled in the hand and held firmly by the base of the legs (Figure 22). Avoid placing pressure on their throat or body.
- Frogs will often be caught in terrestrial environments and while they may be well-hydrated in a pitfall trap

several hours after sunrise, they will be released onto the surface while temperatures are rising. The sorts of frogs caught in such situations are usually burrowing species, and they should be released where they can burrow into soft, shaded soil, and they can even be placed into a small hole and covered in loose soil and/or leaf-litter. Moistening the soil in such a release hole is useful, especially if the frog is slightly dehydrated, but note that frogs will often hop away frantically when first released. A light covering of soil and litter will usually settle them and encourage them to dig further.

- Frogs that are slightly dehydrated can be released successfully with extra care such as moistening the soil in the release hole. Frogs that are badly dehydrated but still alive can be placed in moist soil in a cloth bag and retained for later release. Do not soak bag as this reduces air flow. If retaining frogs (or any animal) in a cloth bag, ensure the bag is placed in the shade, in a cool area and where it cannot be stepped or sat upon. The bag should be labelled so the specimen can be returned to the same location for release.
- **Euthanasia.** Frogs are occasionally victims of other animals in traps: beetles, ants, centipedes and small mammals will injure and kill frogs. If still alive they are best killed with a crushing blow to the back of the skull. A small screwdriver is effective for this. A frog with a damaged back leg is best euthanized, but a frog with a damaged front leg may well survive as frogs with a missing front leg are occasionally caught. Frogs, like reptiles, are 'ectotherms' and can thus be humanely killed by lowering their body temperature. If there is ready access to a fridge and a freezer, this should be done by first chilling the animal in the fridge until it is unconscious (body temperature of around 5°C), and then transferring the animal to the freezer. Animals should not be placed directly into a freezer as it is believed developing ice crystals in tissues may cause pain before the animal becomes unconscious.
- **Hygiene.** Some frogs exude toxins from their skin; members of the genus *Heleioporus* exude a sticky, milk-white substance when upset and this apparently can make people very sick if it makes contact with the eyes or mouth. However, a gently-handled *Heleioporus* will not usually exude this toxin. Fungal diseases can be spread between frog populations; therefore equipment used to handle/measure frogs should be cleaned between field trips and even between sites if the populations are isolated.

Figure 22. A comfortable and secure hold for a large frog. Pressure is placed on the thighs, not on the body (photo T. Gamblin).

REPTILES

Reptiles are a diverse group and thus a range of approaches to handling is needed. Some are so small and fragile they are easily crushed; others are large and can inflict serious injuries, while of course some snakes are highly venomous. Main points to consider when handling and releasing reptiles are:

- Small lizards can be restrained around the base of the neck by creating a 'collar' between the thumb and the first two fingers. They can also be restrained by pinning down one of the rear feet and having the body supported; but some species and individuals will thrash about and may injure themselves with this hold. Larger lizards can be grasped around the neck and the pelvis. Large varanids are best handled by keeping most of their bodies inside in a sturdy cloth bag as even when restrained so they cannot bite, they can still scratch with both the fore and hind feet. All geckoes (with just a few exceptions in the genus *Nephrurus*) and legless lizards, and most skinks, have fragile tails which they will readily drop if handled roughly. In contrast, agamids and varanids can be held by the base of the tail. Species and even individuals within a species will differ in their response to being handled. Lizards are very flexible so maintaining a firm hold with pressure on the pectoral and/or pelvic girdle is often necessary. Do not put pressure on the body (Figure 23).
- Snakes range from tiny, harmless species to large pythons, and dangerously venomous species may not be exceptional in size. If a reptile is a snake or thought to be a snake, it should only be handled if you are totally confident of its identification and have had appropriate training. Even small, very mildly venomous snakes will bite if roughly handled, and while the venom may be mild there may be an individual response to the venom that cannot be predicted. As a general rule, dangerously venomous snakes (genera *Acanthophis*, *Pseudonaja*, *Pseudechis*, *Notechis* and *Oxyuranus*) should not be handled or should only be handled enough to extract them from a trap. Blind snakes (*Ramphotyphlops* spp.) are non-venomous, do not bite but are extraordinarily wriggly and exude a pungent paste that may be irritant to some people. Small snakes such as *Simoseelaps*, *Neelaps*, *Brachyurophis* and *Vermicella* are mildly venomous, rarely even attempt to bite and can usually be handled safely; but they should be allowed to flow through your fingers and not be restrained. If restrained they may bite. Slightly larger species generally considered mildly venomous (*Suta*, *Parasuta*, *Denisonia* and even *Demansia* and *Echiopsis*) are often placid but individuals can vary. Some pythons can be cantankerous and their bite is unpleasant while not considered venomous, can become infected. If gently handled, pythons will not usually bite. Handling any snake is best done only with someone present who is experienced. For example, a python may be placid or very aggressive, and it takes some skill to determine which way an individual will go.
- Reptiles should be released in the shade and directed towards cover. A small lizard will die almost instantly if placed on hot sand. Lizards can also be taken by predators if released in the open. Species with specific shelter requirements, such as some geckoes, should be released so they can slip quickly into their preferred

micro-habitat. With species that live in colonies, such as some *Egernia* and allied genera, it is important that individuals are released into their colony's territory; this will usually be the nearest obvious habitat, such as a dead tree with many crevices, to the point of capture. All releases should take place close to the point of capture but make sure the reptile doesn't run straight back into the trap.

- If a reptile has to be retained for a while, such as for identification, it should be placed in a labelled cloth bag. Cloth bags used for holding any animal need to be free of loose threads to avoid entanglement (Figure 24). If retaining reptiles (or any animal) in a cloth bag, ensure the bag is placed in the shade, in a cool area and where it cannot be stepped or sat upon. Reptiles can safely be held in a cloth bag for 24 hours but there is a risk of the specimen getting overheated or accidentally injured. In warm environments, even if the specimen is held in an air-conditioned room, a small (fingernail-sized) piece of moistened tissue paper should be placed inside the cloth bag with it. Reptiles can dehydrate quite quickly inside a dry cloth bag in a dry atmosphere.
- **Euthanasia.** Reptiles are occasionally victims of other animals in traps: beetles, ants, centipedes and small mammals will injure and kill small reptiles. If still alive, they are best killed with a crushing blow to the back of the skull. A small screwdriver is effective for this. Reptiles can recover from quite substantial injuries and it is quite common to capture lizards with scars on the back, missing toes and even missing limbs. Therefore, if an injured reptile is still active and able to seek shelter, it may be better to let it go "to take its chances" than to euthanize it. Reptiles, like frogs, are ectotherms and can thus be humanely killed by lowering their body temperature. If there is ready access to a fridge and a freezer, this should be done by first chilling the animal in the fridge until it is unconscious (body temperature of around 5°C), and then transferring the animal to the freezer. Animals should not be placed directly into a freezer as developing ice crystals in tissues may cause pain before the animal becomes unconscious.
- **Hygiene.** Reptiles frequently defaecate when handled. During any animal handling, personnel should avoid hand to mouth contact and wash their hands when possible, particularly before eating.


A.


B.


C.

Figure 23. Examples of a range of reptiles being held in comfortable but secure holds. (A) Small dragon restrained with pressure on a rear leg, and the body supported. (B) Small varanid restrained by being held around the pectoral girdle and by the base of the tail. (C) Small skink restrained with three fingers around the pectoral girdle and supported (but not held) under the tail (photos A. Bamford).

Figure 24. A cloth bag with loose threads that could entangle specimens. Note this bag has also previously been used to hold a bird (feathers on left). (photo A. Bamford).

BIRDS

Handling of birds is a specialised area that needs training, usually gained as part of an Australian Bird and Bat Banding Scheme program. However, birds are occasionally caught in pitfall, funnel, Elliott and cage traps, and therefore some familiarity with the basics of handling birds is needed by people undertaking such trapping.

When trapped, birds are likely to panic and are prone to injury. They therefore need to be calmed down as quickly as possible, such as by covering the trap with a cloth. They also need to be removed as quickly as possible. However, hurrying the release of a bird from a trap can cause injury. For example, a bird hurriedly released from a cage trap could catch a leg in the trigger mechanism. Birds can also inflict injury on an unwary handler: even small parrots can bite very hard, the larger passerines such as wattlebirds and bowerbirds can cause puncture injuries with the large hind claw (hallux), and small birds of prey can inflict damage with their feet.

Before attempting to release a bird, make sure it has not injured itself in the trap; it is quite possible for a bird in the confines of a trap to break a leg or wing, but still to scurry out of a trap when the door is opened. Once the bird has been checked, if it can be released safely by simply opening the trap door, then this should be done. If possible, direct the fleeing bird towards trees/bushes rather than to open ground in case predators are watching. If the bird needs to be handled to remove it, it can be encouraged into a cloth bag (as with small mammals from Elliott and Cage traps), or in larger traps a cloth can be dropped over. If small birds need to be handled, they can be cradled in a 'cage' made of both hands, or the 'banders' hold' can be used (Figure 25). This is a very useful hold for examining birds; the head is placed between the first and second fingers and the body is caged inside the fingers and thumb, but with no pressure on the thorax. When releasing a held bird, place it on the ground and move the hands/cloth slowly away. Do not toss the bird into the air as it may be temporarily disoriented and not able to fly.

Occasionally chicks of species such as quails, button-quails, plovers and even the Emu get caught in pitfalls. There is a risk that the adult bird and other chicks will have moved on, but the best procedure is to release the chicks into cover and quickly move away. Trapped chicks are often very noisy and it is likely that the adults will be waiting nearby.

Birds caught in traps may be dead due to injuries or stress. Some species are also known to suffer a capture myopathy in which the major muscles of the wings and legs deteriorate rapidly. Unless there is ready access to wildlife carers, it may be necessary to euthanize birds with a sharp blow to the back of the skull (referred to as blunt force trauma). Small birds can also be euthanized by sternal compression, where a finger or thumb is pressed hard onto the sternum to prevent the bird from breathing. Both these methods are probably best demonstrated.

Figure 25. A male Variegated Fairy-wren held in the banders' hold. In this case, the feet are also being held with a second hand; note band on the bird's left leg (photo S. Smith).

MAMMALS

Mammals are a diverse group and thus a range of approaches to handling is needed, but a common feature is the need for quiet and darkness. Covering the head of any mammal, from a Dasykaluta to a Red Kangaroo, is surprisingly effective at ensuring the specimen lies still with minimal direct restraint. Handling of mammals is a broad topic and the larger species, such as wallabies and kangaroos, are unlikely ever to be handled in the course of regular fauna surveys. However, species as big as the Brush-tailed (Brush or Black-gloved) Wallaby *Macropus irma* have been found caught in standard 20 litre pitfall traps. Field workers may also occasionally encounter injured kangaroos and be expected to "deal with them". Main points to consider when handling and releasing mammals are:

- Small mammal species and even those as large as a possum should be retained inside a cloth bag, with measurements being taken and examinations done with the head covered as much as possible. This will keep the specimen calm to some degree, but be aware that almost all species can and will bite through the cloth if given a chance. Bandicoots will just kick with their extremely large hind claws and long nails, and therefore especially tough bags are needed when trapping them. Even when largely contained within a bag, mammals need to be restrained so that measurements can be taken, the pouch of female marsupials examined and the specimen can be marked. Small mammals (up to the size of a rat) can be restrained by pinning them down behind the head with the thumb and forefinger, with the rest of the hand lying firmly (but not pressing down) over the body. This can be done through the cloth. Be aware that if pressed down against your own leg, the animal may well bite your leg! Larger mammals (bandicoot, possum, Chuditch) require a whole hand over the shoulders and neck to pin them down, with the second hand possibly needed to control the rest of the body. Having two people to handle larger mammals is thus helpful. Even with such larger animals, it is important not to put pressure on the rib cage. Note that handling mammals requires a personal approach to some degree, and what works for some people might not work for others. Each species can also be different. Water-rats (Rakali) have loose skin and seem to be able to move inside it, making them very hard to pin down and requiring a lot of pressure to restrain them. Some bandicoot species (notably Quenda) will kick viciously with their back legs at the hand placed over their shoulders.

- Some small mammals can be "scruffed" by holding them by the loose skin on the back of the neck, but while recommended in some SOPs (eg. Petit and Waudby 2013), this is probably stressful and best avoided. In addition, in some rodents the fur and even the skin will pull away in chunks. The tail can be used to restrain some mammals but in many native rodents the skin will strip from the tail like a stocking, leaving bare flesh and vertebrae. The tail will later die and fall off. In contrast, the tail makes an excellent handle when controlling a Honey Possum, which almost never bites (and they are too small to do damage if they do) and has no claws with which to scratch.
- If wallabies and/or kangaroos need to be handled, they can be grasped by the base of the tail; a large kangaroo can be "steered" in this way. Once held by the tail, a wallaby can be lifted from the ground with the other hand placed firmly on the animal's chest between its forelegs, and the animal's back pressed against the chest of the handler. This is best demonstrated before being attempted; the rear legs will lash out away from the handler, but therefore towards anyone standing nearby. Once so restrained, the wallaby will calm down immediately a hood is placed over its head.
- Female marsupials are known to drop their pouch young occasionally when in traps or when being handled. This can be very difficult to deal with and is most common with potorooids, bandicoots and wallabies/kangaroos. If targeting such species, consider trapping when large pouch young will not be present; or reassess the need to carry out trapping. If young are dropped, it is sometimes possible to get them back into the pouch. Some females will take the young back into the pouch if left quietly in a cloth bag for 20 minutes. In other cases, the young can be physically pushed back into the pouch and the pouch sealed preferably with surgical tape, but sticky labels have been used successfully. Unfortunately, some young will still fall from the pouch when the female is released. These must either be euthanized or, if available, taken to a wildlife carer.
- Mammals should be released in the shade and directed towards cover. While most are nocturnal, traps are usually checked in daylight and thus the release will take place in daylight. This makes it all the more important that the animal can go directly into cover. Petit and Waudby (2013) suggest that traps should be checked before sunrise if nocturnal mammals are being caught, but this is probably impractical under most circumstances. All releases should take place close to the point of capture but make sure the animal doesn't run straight back into the trap. Some small mammals appear to follow scent trails and have been seen to meander many metres back towards a pitfall.
- Mammals cannot be held for extended periods without food. They can be held during the day but will need to be released in the evening. If a mammal has to be retained for a while, such as for identification, it should be placed in a labelled cloth bag, placed in the shade, in a cool area where it cannot be stepped or sat upon. Holding mammals for more than 12 hours is difficult and small species (rodents, small dasyurids) will need to be fed. Rodents will take readily to rolled oats and apple if they do need to be fed.
- **Torpor.** Small marsupials (eg. small dasyurids, Honey Possum, Pygmy-possum) enter torpor naturally, and will do so when trapped under cool conditions (Figure 9). They allow their body temperature to drop to conserve energy, and they can be found curled up and barely responsive when handled. However, they are stiff to handle, almost rigid, unlike small mammals that are in uncontrolled hypothermia and near-death, which will be limp. Rodents generally do not enter torpor and can become hypothermic; they can be revived quickly with body warmth (see page 15). Small mammals in torpor are easy to handle but should be processed quickly and placed under cover when released, since they are barely able to move. When recovering from torpor, small mammals will shiver violently; this is perfectly normal.
- **Bats.** These are occasionally caught in pitfall traps (particularly the long-eared bats *Nyctophilus* spp. that forage close to the ground and will even enter pitfalls to prey on invertebrates and lizards), and may also be the subject of specialised studies. They are best handled only by experienced personnel. Where they have to be handled (such as a bat in a pitfall), cover them in a cloth bag to remove them, keep them largely in the cloth bag and release into a hollow tree or similar. There is some concern about the transfer of disease between bats and people and while this seems to be very unusual, personnel who regularly handle bats are recommended to have Rabies immunisation (the viruses from bats are related to the Rabies virus, and where Rabies occurs it is sometimes carried by bats).
- **Honey water.** This is occasionally used for nectivorous or partly nectivorous species if an individual seems unusually weak or to get it through to a nocturnal release. Honey Possums in particular will lap up a solution of honey water (Figure 26).
- **Euthanasia.** Mammals are occasionally victims of other animals in traps: beetles, ants, centipedes and other mammals will injure and kill small mammals. There are also occasions when pouch young may be dropped in a trap. It is usually impractical to care for such animals and therefore euthanasia may be necessary. A sharp blow (blunt force trauma) to the back of the skull is effective. Larger mammals (possums, bandicoots, kangaroos) can similarly be euthanized. When working in the field injured animals are occasionally found on the roadside, and there is often an expectation from other personnel that zoologists will know how to put such animals "out of their misery". A tyre lever, hammer or back of an axe may be needed to provide effective "blunt force trauma" to the back of the head of a large mammal. If delivered effectively, death appears to be instantaneous and this technique is accepted by the RSPCA.

- **Hygiene.** Mammals occasionally defaecate when handled. During any animal handling, personnel should avoid hand to mouth contact and wash their hands when possible, particularly before eating.

Figure 26. A Honey Possum being fed a solution of honey and water before release. (photo T. Gamblin).

Non-capture techniques

While fauna investigations commonly involve capture and handling of animals as outlined in the previous section, there is a growing field of non-capture techniques. These can complement trapping programs although in some cases, non-capture techniques may be more effective at detecting a species than capture techniques. For example, the use of ultra-sonic detection has revolutionised bat surveys, while some mammals (eg. Chuditch, Bilby) can be readily detected using motion-sensitive cameras and track-searching respectively, but are very time-consuming and often difficult to trap. Non-capture techniques have the advantage that the animal is not interfered with and that there may be no legal requirement for a permit (see page 4). However, non-capture techniques do not allow for the collection of biological data such as measurements and observations on reproductive status.

The use of non-capture techniques is a broad field and the following notes provide only an introduction to the main approaches currently in use.

FAUNA SIGNS: TRACKS, SCATS, NESTS AND BURROWS

Fauna signs are particularly useful for species of high conservation significance that can be difficult to trap, or for which trapping is not appropriate. Using fauna signs requires a lot of personal experience, but Triggs (2010) has collated information mainly on mammal tracks and scats, with a good array of photographs to aid identification. Bamford, Gamblin, Browne-Cooper and Turpin (in prep.) are developing a guide to non-capture techniques for detecting and surveying a range of significant species in Western Australia, and information is also provided by Thompson and Thompson (2011). Scatsabout is a small, Melbourne-based company that will attempt to identify scats for a small fee. A similar service is available through Murdoch University in Perth, including the extraction of DNA from scats (Australian Wildlife Forensic Services and Ancient DNA Lab, Murdoch University). Benshemesh and Shulz (2008) have developed a reliable survey technique for searching for marsupial moles (Figure 27), while the presence of three black-cockatoo species can be determined by examining foraging signs on Marri nuts (Figure 28). This is likely to be an area to experience enormous growth in the next decade as researchers develop a greater understanding of the biology and behaviour of significant fauna.

The key to using fauna signs in survey work is to become familiar with the signs of common species and to familiarise yourself with what the signs of significant species should look like. Learn to observe. Especially in sandy environments, it doesn't take long to learn to interpret tracks and to recognise those made by beetles as opposed to those made by lizards, to recognise tracks of small mammals and to tell in what direction a lizard is travelling (Figure 29). The drey of a Western Ringtail Possum is quite a different structure and placed in a different part of the tree from similar-sized nets of butcherbirds and magpies (Figure 30), but you need to be familiar with both to recognise the former. Part of the reason for becoming familiar with the signs of common species is so that the observer notices unusual signs (because they are different), and to avoid inundating experts with photographs and samples that could easily have been identified as common species in the field with just a little knowledge. It is very important to record observations so these can be checked by people with experience in this area. Tracks, scats and burrows should be photographed with a scale (preferably a ruler but a pencil will do) as in Figure 31; scats should be collected if possible so they can be sent to an expert for further examination. A description of the environment is also important as this may tell an expert what species might or might not be present.

Fauna signs can also be used as a monitoring technique for rare and common species. Western Ringtail Possums can be surveyed by counting their dreys, while the abundance of Western Grey Kangaroos has been monitored by counting the scats in 1m wide by 100m long transects, with a scat density of 10,000/ha being equivalent to a kangaroo density of 0.1/ha (Arnold and Maller 1987). A successful approach to counting kangaroo scats involves using a 100m length of strong cord to progressively mark out the transect in 100m units, with scats counted by walking along this string with a T-shaped frame having a 1m long horizontal section held close to the ground to define the 1m width. Each 100m length is 1% of a hectare, allowing for easy calculations of mean and standard deviation of scat density.

Counting footprints along sandy tracks can also be used to measure the abundance of a variety of species, especially if the sandy track can be smoothed at regular intervals. A variation on this is the use of sand pads, in which a clear area of sand is placed around a bait station and footprints in the sand checked regularly. Sand pads require regular maintenance and bait replacement, and are often used to monitor feral species such as Cats and Foxes. They are probably best used in conjunction with motion-sensitive cameras (page 31). Sand pads (especially with cameras) can confirm the presence of species but the relationship of records with actual abundance is probably unreliable.

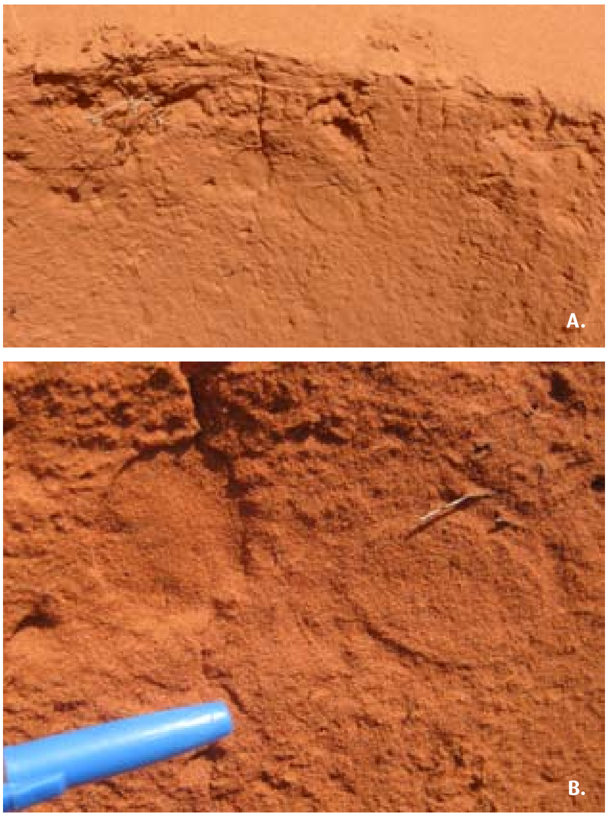

Figure 27. Marsupial mole burrows revealed by digging a trench on the side of a sand-dune in the Little Sandy Desert. One burrow is circular, the other elliptical because of the angle of the burrow, and both were back-filled with sand as the animal moved forwards (photo A: S. Smith, photo B: T. Gamblin).

Figure 28. Marri nuts showing the characteristic pattern of chew marks of Baudin's Black-Cockatoo (A) and the Forest Red-tailed Black-Cockatoo (B). (photos M. Bamford).

Figure 29. Track of Gould's Sand Goanna *Varanus gouldii*. The animal has travelled from left to right. The long, curved indentation is tail-drag, the prints of the front feet are on the right, with a hind foot print roughly central. The multiple narrow curved lines are the drag-marks of the claws of the hind feet as the animal swings its legs wide as it walks (photo M. Bamford).

Figure 30. Drey of a Western Ring-tailed Possum (photo S. Cherriman).

Figure 31. Scats of a Northern Quoll, with a pen used to give a scale (photo T. Gamblin).

MOTION-SENSITIVE CAMERAS

These are fairly new in wildlife research in Western Australia, and were developed mainly for the hunting industry in the USA, where the cameras are placed on game trails (hence sometimes called game cameras) so that hunters can determine what animals are around for the hunt. They are extremely effective for confirming the presence of mammal species detected by scats or tracks, such as Bilby, Northern Quoll and rock-wallabies. They can also be used to examine fauna coming into waterholes (Figure 32), to monitor activity levels and to determine the local distribution of species such as Cats and Foxes.

Several commercially-produced brands of motion-sensitive cameras are available. These have broadly similar features, such as data storage on a standard SD card, a choice of video or still photos, and variable settings for sensitivity and the number of images taken in a series. Most take colour photos using natural light in daylight, and black and white photos using infra-red flash at night. However, white-flash or incandescent-flash cameras are also popular with some fauna specialists as they have the ability to take clear, colour night-time photos of animals which may be useful for identification. Typically these cameras are limited to still photograph mode, at least during night operation. Box 2 contains some suggestions on what to look for when choosing motion-sensitive cameras.

Figure 32. A motion-sensitive camera (in this case a Bushnell Trophycam) set in a plant-pot to record birds visiting a garden birdbath (photo M. Bamford).

Some important points to note when using motion-sensitive cameras are:

- The camera should be stable, either tied to a large tree or rock, tied to a stake or set up on a tripod. If the camera is unstable (such as tied to a small tree), movement of the camera itself will cause photographs to be taken.
- It is best to face the camera south to avoid sun-flare on the lens and to avoid sun-generated false triggers with the rising sun, mid-day sun or setting sun.
- If possible, place a scale in the view of the camera. This can be a ruler, a pencil, the lid of a jar or just a branch of known length.
- Most cameras can be set in video or still mode, though some makes will record in 'hybrid' mode, recording both still photos and video with each trigger. Still mode allows for more photographs to be taken and it is easier to review still photographs than video, although video captures behaviour that might not otherwise be observed. Reviewing mixed still photos and video can take longer than either all photos or all video. Still photographs can be set to be taken in groups of three with only a few seconds between each image, and this is a useful feature when reviewing photographs.
- The light source of the cameras is designed for objects at a distance of up to about 20m (typically large game animals), whereas they are often used for much smaller animals at closer range. Masking some of the light source (such as with opaque or translucent tape) can reduce white-out (Figures 33 and 34). This can be a matter of trial and error and it is important not to cover the light sensor or arming lights as this may compromise operation of the camera. Several makes of cameras now offer variable LED settings, giving the choice of low, medium or high illumination, and this has the added advantage of not wasting battery power. For small fauna work at close range, the low and sometimes normal settings are generally most applicable. Reconyx cameras are some of the few that have minimal white-out without any masking or modification.
- Beware of moving branches, leaves and grass which will set off the camera repeatedly. This is especially a problem in hot, windy weather, and particular care should be taken to trim off problematic foliage etc. On some occasions, even the shadows of overhead branches moving in the wind can generate false triggers.
- Cameras can be set on burrows, trails, locations where animals have left scats, waterholes and can also be baited. If baited, beware that animals such as crows may turn their attention to the camera when the bait runs out! Burying some bait can ensure that animals stay within camera range for extended periods.
- Cameras can be left out for any time period and most will have the battery life and data storage capacity for several thousand images. In some projects, cameras have been left out for several months, with data downloads every fortnight and batteries replaced less frequently. However, the length of time a camera can be left out does depend on the activity it records. A camera set to photo mode with eight freshly-charged AA batteries and a 4GB memory card set up next to a well-used desert waterhole, for example, may only operate for several days before the card fills. The same camera set up next to a rock crevice or burrow with 'moderate' activity may last for one to two months without checking before the batteries run flat or the card fills.
- Rechargeable batteries are commonly used with motion-sensitive cameras, and the environmental and economic advantages quickly become apparent after many uses. If using rechargeable batteries, low self-discharge NiMH are recommended (brands include 'Eneloop' by Sanyo and 'Recyko' by GP) – these batteries are pre-charged in their packets when bought. Cheaper types of NiMH rechargeable batteries (non low self-discharge) are not recommended due to their tendency to lose charge when not in use.
- Keep a good record of where cameras have been left. Each camera should have a code number, this should also be written on the camera's SD card, and the code number should be recorded with the camera location and description of the site.

- Cameras should not be placed in locations that are conspicuous to the public, as they are likely to be stolen if found. Privacy concerns may also be raised.
- Check setting of cameras regularly: date, time and even compatibility with SD card – these can alter without warning! It is good practice to format SD cards regularly but especially when an SD card is being used with a camera for the first time; but be aware this will erase all files on the card.
- Reviewing photographs taken with motion-sensitive cameras is very time-consuming and needs to be done carefully. It seems inevitable that the most interesting animals are on the edge of the image or are facing away from the camera. When clicking through still images, watch for change from one image to another as this is easier to notice than actually recognising an animal. While most newer cameras have a small screen, it is preferable to review photographs on a computer. Remember to accurately catalogue photographs and it is worth storing all images in case you need to refer back to them.

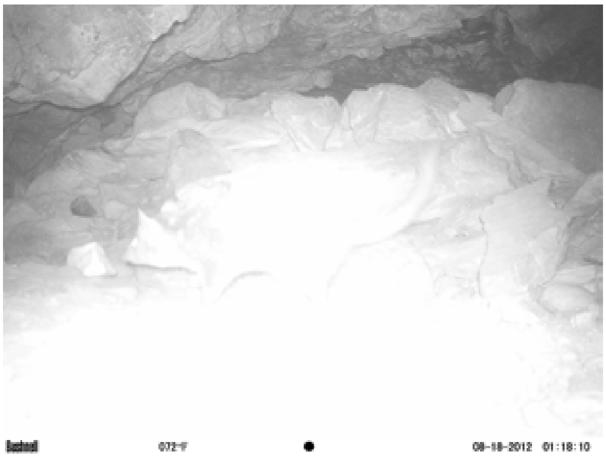

Figure 33. Photo taken at night showing white-out from too much illumination; light-source was inadequately masked. Despite this, the photograph was adequate to confirm the presence of Northern Quoll where the species had not previously been recorded (photo J Turpin, from Gamblin *et al.* 2012).

Figure 34. Photo taken at night with light-balance about right and a juvenile Bilby clearly recognisable (photo P. Smith).

ULTRASONIC DETECTORS FOR BATS

Insectivorous bats (micro-bats) are a substantial proportion of the mammal fauna in many areas, but are difficult to trap and were often over-looked in fauna studies until the development of (relatively) low-cost ultrasonic detection units made them readily accessible to fieldworkers in the 1990s. These detection techniques record the ultrasonic calls that the bats use to echo-locate, and because the calls are mostly distinctive in wavelength, frequency and general structure, the calls can be analysed to determine what bats have been flying around. For this analysis, a reference collection of recordings is needed, but these are now available for many species.

Within Australia, the Anabat unit (a frequency division system produced by Titley Electronics) has been the most popular; however continued development of technology within the field has produced a range of new units, many utilising several call analysis methods. The Songmaster 2 (SM2, a frequency-division system produced by Wildlife Acoustics) is increasingly being utilised by fieldworkers (Figure 35). All of the units available generally use one or more of the following recording methods.

Heterodyne detectors

Heterodyne detectors are tuneable to specific frequencies and are useful when conducting active surveys for species with an audibly distinguishable call. This requires the user to be familiar with the bats of the area and their characteristics of their calls.

Frequency division detectors

These frequency division divide call frequencies down by a set ratio, bringing them into the range of human hearing. These units record across all frequencies within the range of the microphone/s. This system is used in conjunction with Zero-Crossings Analysis (ZCA) by the Anabat system.

Time-expansion detectors

Generally used for more in-depth study of bat calls, this system records a call at a high sample rate and replays it at a slower speed, reducing the frequencies to audible levels eg. a 50kHz bat call slowed down by a factor of 10 will produce calls of 5kHz. The recorded calls can then be further analysed.

The frequency-division systems are the most suitable for passive monitoring; units can be deployed overnight and call recordings analysed at a later date. The following websites provide reviews of some of the detectors currently available.

- batdetecting.blogspot.com.au
- durhambats.org.uk/reviews.htmbatmanagement.com/Ordering/acoustic/acoustichelp/bdcompared1.html

BOX 2 *Suggestions for choosing cameras*

1. Heavy (when holding all batteries) and bulky cameras can be difficult to transport and set. There are now small cameras available with good battery life such as Bushnell Trophy Cam HD Max and Reconyx HC series. LtL Acorn is one of the smallest cameras on the market.
2. **Battery life:** It is hard to gauge battery life as this will vary with activity at a camera site as well as temperature, age of batteries, etc. Most makes operate on 6 volt systems but some operate on 12 volts, and these have half the battery life expectancy.
3. **Battery type:** Some cameras operate on C-cell batteries, some on D-cell and some even require both. However most operate on AA batteries, making it easy to buy good quality, low self-discharge batteries. It is difficult to find low self-discharge batteries in C and D-cells. It is also a disadvantage to have to supply two or more types of battery for each camera, such as C and D-cells in Leaf River cameras.
4. **Ease of use:** Many makes are intuitive to operate for the first time and virtually do not require the users' manual (however it is always recommended to read these). Bushnell, Ltl Acorn, DLC Covert, Scoutguard, and Reconyx are examples of easy to operate cameras; Leaf River cameras are not easy to operate and the instruction booklet is of limited help; some of the white-flash type cameras are also difficult to operate. Ease of use is particularly important when several people different skill levels may be using the cameras.
5. **Trigger speed:** Cameras should have a relatively fast trigger speed to maximise capture of animals; with a slow trigger speed the animal may have entered and left the field of view before the photograph is taken! 1.3 seconds is satisfactory (eg. Ltl Acorn, DLC Covert, Moultrie D-555i), 1 second or less is very good (eg. Leupold, at least two Moultrie models), 0.6 seconds is excellent (eg. Reconyx models, several Bushnell Trophy Cam models). Trigger speed can be partly or entirely compensated for by extra-wide detection capacity of specific cameras (see Moultrie Panoramic 150, Ltl Acorn – most models – with side sensors).
6. **Photo and video resolution:** Many camera makes now allow for up to 8 megapixel photos to be captured and 1280x720 High Definition video. High quality images are an advantage for identification.
7. **Remote controller:** Reliance on remote controllers is considered a disadvantage. Most makes of camera can be programmed on a special built-in panel on the camera itself. However some require a plug-in remote controller to program; this can be fiddly and time-consuming when done repeatedly with several cameras in the field. It can also be problematic with multiple camera operators – if the remote controllers are lost the cameras cannot be re-programmed. This can be particularly awkward if date and time settings are lost when batteries are flat or removed from the camera. Several Scoutguard cameras use remote controllers, as do Leupold cameras (However the Leupold remote controller doubles as a high quality card viewer).
8. **Photo/video viewer:** Some makes/models of motion-sensitive cameras have built-in viewers for on-the-spot viewing of captured photos/videos. This is useful in the field to gauge whether or not a camera is in a good position and is working correctly, but the viewers should never be used as the basis for deleting files from a memory card as it is very easy to miss animals.
9. **White-out:** Most camera makers strive to make their infra-red flashes (LEDs) as bright as possible for maximum illumination of North American deer, moose, elk etc. at considerable distances from the camera. If possible, choose a camera that has more balanced illumination (notably Reconyx) or a camera that offers a choice in LED settings, allowing the operator to select a lower setting.
10. **Timer function:** This can be very useful if the target species is mostly nocturnal. The camera can be programmed to stand by during set hours (eg. 0900 – 1700 hours) then start operating at a specified time (eg. 1700 or 1800 hours). Not all camera makes and models have this function.
11. **Low-glow infrared versus standard infrared flash:** The LEDs of low-glow or 'black flash' infrared (IR) cameras are more-or-less invisible to human and mammal eyes. The red-glowing LEDs of 'standard' IR cameras are quite visible on the other hand, even though the infrared light produced by both is invisible to humans and other mammals. Low-glow cameras are considered to have less impact on wildlife and are thus to be preferred. Low-glow cameras may also be useful in areas frequented by people – their IR flash units are virtually invisible to the human eye while videoing in the dark of night. Some standard red-glowing cameras have very shiny silvery LEDs that are very conspicuous during sunny daytime weather and are more likely to attract attention, eg. some models of Scoutguard. Low-glow 'black flash' do not have conspicuous silvery LEDs.
12. **Video format:** For reviewing purposes, it pays to know whether a camera's video format can be watched in 'fast-forward' while using the appropriate program (eg. Windows Media Player). This may be a matter of trial-and-error or learning from the experience of others. Many AVI and ASF videos allow fast forwarding but many MOV videos do not. Capacity to review video in fast-forward allows for much faster checking of memory cards and can be a worthwhile advantage if many hundreds of video clips are captured from a single camera. If cameras are used mostly in photo mode, this is of little consequence.

Camera set on a Bilby burrow (photo T. Gamblin)

Figure 35. SM2 recording unit set up on a fence post (photo N. Dunlop).

ANALYSIS OF CALL RECORDINGS

Analysis of bat calls is generally best left to an expert who should have an extensive call library from species occurring in the area where calls were recorded. A range of characteristics of each call recording will be analysed and compared to reference calls. Some of the call characteristics used for analysis include:

Fmin Average minimum frequency of pulses within a call sequence

Fmax Average maximum frequency of pulses within a call sequence

Dur Average duration of pulses within a call sequence.

It is unlikely that all calls recorded will be of a quality suitable for analysis, and even high quality calls may not be identified; in some areas, several species may produce indistinguishable calls. For example, within parts of the Kimberley there are three vespertilionid species that have similar calls, *Scotorepens greyii*, *S. sanborni* and *Chalinolobus nigrogriseus*.

Some of the variety of bat calls from within an area of the western Pilbara is shown in Figure 36. In this example, each call is relatively easy to distinguish from the others based on its call shape and its minimum and maximum frequencies.

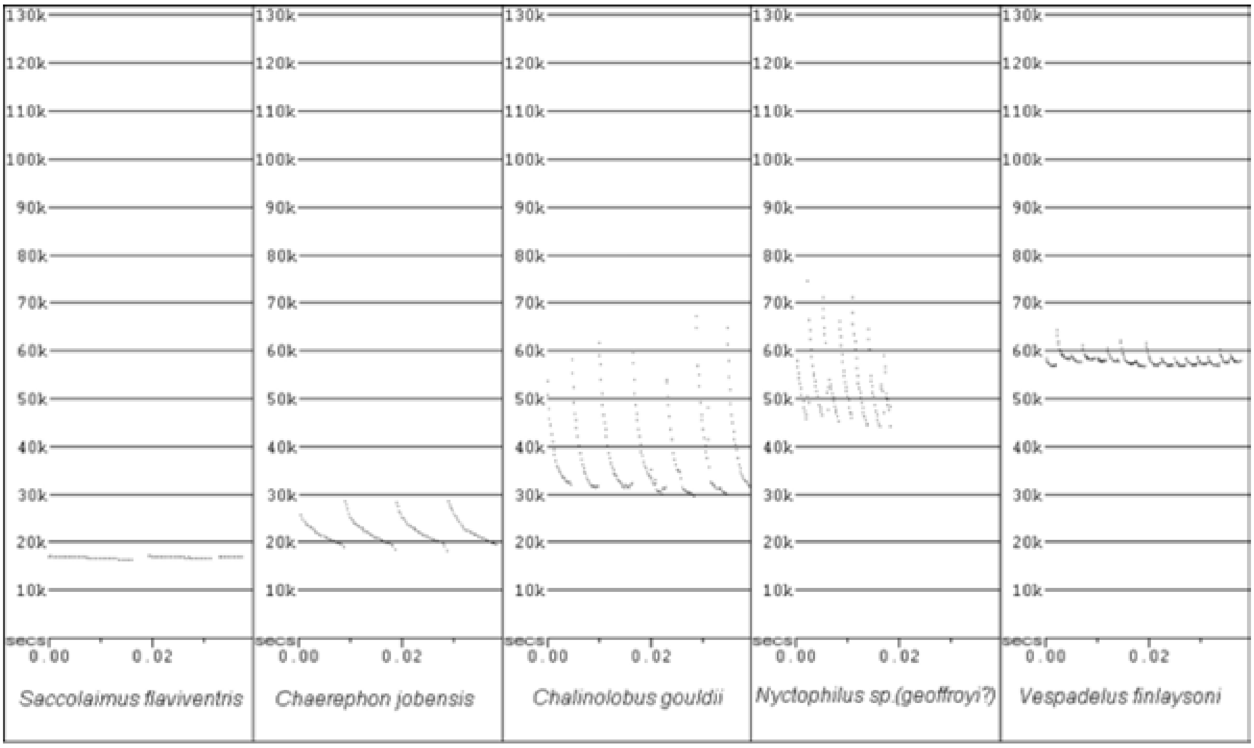

Figure 36. Variety of bat calls from within an area of the western Pilbara (source: B. Metcalf)

TIPS FOR RECORDING

The Australasian Bat Society (ABS) provides a range of recommendations on conducting surveys for bats (ABS, undated) including:

- Conducting trapping in conjunction with detector sampling, so as to ensure as many species recorded as possible (including some that may be difficult to identify)
- Detectors should be deployed for a minimum of three nights in each major habitat type of a study area
- Surveys should be conducted during warmer months of the year and in good weather conditions.

Some of the best places to record bats include around waterholes, along watercourses, at the entrances to caves, along forest tracks or other flyways; where a change in vegetation forms an edge, bats will often fly along this edge. Where a large number of bats congregate, it may be better to move detectors away from the main congregation, otherwise calls may be difficult to separate.

AURAL SURVEYS FOR FROGS

Frogs are a remarkable group of fauna and, along with birds, are highly vocal. Frogs can be sampled directly as both tadpoles and adults (many species are readily caught in pitfalls), but their loud and species-specific calls make them ideal candidates for aural surveys. Aural surveys for frogs are quick, virtually no equipment is required (although a simple recorder can be used to record unfamiliar species), and calls of most species are widely available so it is easy for a surveyor to become familiar with them. Recordings of the calls of Western Australian frogs can be found at: http://museum.wa.gov.au/research/collections/terrestrial-zoology/herpetology-reptiles-and-frogs-collection/frog-calls

The presence of frogs can quickly be confirmed through an aural survey approach, although information is needed on the season when frogs call as the calling season for some species is very narrow (Figure 37). At Whiteman Park, surveys would be needed in May and July to detect all species, but a third survey would be required as two other species known from the area and not detected by Bancroft and Bamford (2008) call in late spring.

Aural surveys for frogs are an ideal community activity because they are easy to do, and seasonal presence/absence abundance data can readily be gathered. The number of frogs calling can also provide a measure of abundance if limits to the sampling site are readily-defined, and particularly if the survey effort can be repeated (to allow for variations in number calling due to weather, such as rainfall or cold conditions). Surveys should be carried out around dusk, and because only a period of less than five minutes is required at each site, 20 sites can readily be checked in a couple of hours. We have conducted two seasonal frog surveys over an area of 60 by 20 km in four evenings with four people, visiting about 40 sites each evening.

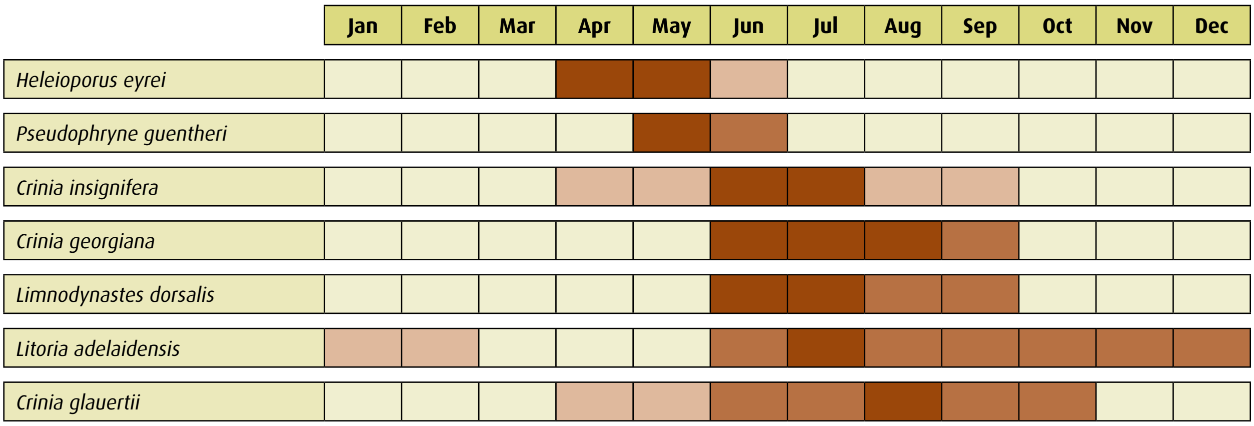

Figure 37. Summary of frog calling phenology at Whiteman Park as an example of differing calling periods (from Bancroft and Bamford 2008).

HAIR TUBES

Many mammals have hairs that are distinctive in microscopic detail, which means that their presence can be confirmed in an area if hair samples can be collected. Hair tubes allow this. Hair tubes can be as simple as a length of PVC pipe with double-sided sticky tape around the interior diameter at each end, and a piece of bait in the middle. More complex hair tubes have only one entrance and the bait is contained in a small mesh cage, but the use of tape is the same.

The diameter of the hair tube needs to be suitable for the mammal being targeted; the animal should be able to easily move through the tube and its hair just brush the tape. Hair tubes should be set where mammals are likely to move; either on the ground or in trees (if phascogales are being targeted). To some extent, motion-sensitive cameras perform the same function as hair tubes.

Hair samples need to be sent to an expert for identification (see page 29). Depending on the weather, hair tubes can only be left for a few days before dust or rain render the sticky surface ineffective, and there may be issues with insects and even small vertebrates (eg. lizards) being caught.

BIRDS; OBSERVATION, CENSUSING AND COUNTING

Birds are unusual in that most are readily observed and identified without being captured. While bird-watching is a popular hobby, it is also an important skill to develop as a field biologist. Half the bird assemblage of an area can be confirmed in just a few days by a reasonably skilled observer. Reliable field guides are available to aid identification, and calls of species can also be downloaded and carried in the field; an experienced bird-watcher will record as many species by call as by sight.

While confirming the presence of species through observation is useful, measures of abundance of birds can also be obtained. Waterbirds can be counted because they are conspicuous and occur in discrete sites (wetlands). Large flocks of waterbirds are counted by estimation. For example, an estimated 10% of a flock can be counted and therefore the flock size calculated to a fair degree of accuracy. In mixed flocks of waterbird species, a proportion of the flock can be counted and the composition by species estimated and then extrapolated to the whole flock. For example, if approximately 10% of a flock of ducks is counted and contains 500 birds of all species, and it is estimated that the whole-flock composition is 50% Pacific Black Duck, 30% Grey Teal, 10% Hardhead and about 2% each of Pink-eared

Duck, Hoary-headed Grebe, Australian Shelduck, Blue-billed Duck and Eurasian Coot, then robust estimates of numbers of each species can be calculated quite quickly.

Aerial surveys of waterbirds are also possible, either by fixed wing light aircraft or by helicopter flying as low as possible (usually about 150m) and at an airspeed that gives the observers time to count. A hand-held digital voice-recorder is essential for this exercise. Such aerial surveys can aim to get estimates of absolute numbers of waterbirds and even recognise the most distinctive species, but it is also necessary to group other species (eg. large shorebirds, small shorebirds, small terns). However, if very large wetlands are being surveyed, a different approach is needed that involves flying fixed transects and counting only waterbirds within this transect. A similar approach can be used for marine mammals. In this aerial transect approach, if a set altitude is flown between two known points, then marks placed on the window or wing-strut can define the limit of the transect giving a known width so that the precise area surveyed can be calculated.

Bushbirds cannot usually be counted in total, and therefore a range of bird census techniques has been developed in order to get measures of abundance. Davies (1984) provides an old but still useful review of such census techniques. Broadly speaking, bird census methods can be divided between point and area search techniques. An example of a point technique is to count all birds seen or heard within 25m of a census point over a period of three minutes, and to do this at a number of points within the same vegetation type in a survey area. This technique can be combined with checking a line of widely-spaced pitfall traps or similar. A typical area search technique involves walking around a pre-defined area (eg. 2 or 3 ha) for a set period of time (eg. 15 or 20 minutes) and counting all birds seen or heard within this area. The area search technique would also be replicated in the same vegetation type. The point census technique tends to be more reliant on skilled observers than the area search technique, and often underestimates birds that are cryptic. Both techniques provide counts for a unit area and with replicated recording sites within vegetation types, the bird assemblage (species and abundance) can be compared between vegetation types.

Such structured census techniques require experienced observers and time strictly allocated to the census activity. They are usually carried out in the first three hours after sunrise to coincide with peak activity (and therefore detectability) periods of birds. However, more flexible approaches can be undertaken that still provide valuable data. For example, birds observed during each visit to a site to check traps can be recorded (and counted if possible); each visit becomes an independent sampling event and the presence/abundance of species can soon be compared between sites. In a long-term study of changes in a bird assemblage in a suburban garden, we have an allocated bird day once a week on which all bird species seen within the garden are noted. Over a period of 20 years, massive changes in the number of species recorded and the seasonal and annual frequency of recording have been documented.

Watson (2003) discusses how recording species over time can allow for assemblages to be compared between sites without the need for structured census sites and periods. This has the advantage that the survey effort is not limited to points or predefined areas, and the survey can take place at any time of the day as the survey ends only when a “stopping rule” is applied; this rule comes into effect when no new species are added to the list after a pre-determined period of time (such as 10 or 15 minutes).

Western Spinebill caught for banding (photo M. Bamford)

BOX 3 *Binoculars and telescopes*

Binoculars are widely used during bird surveys and can also be used to identify reptiles and mammals at a distance, with small telescopes generally used only in waterbird surveys where the extra magnification is useful. When counting waterbirds, the telescope can be used to slowly scan a flock and can be paused and left on a group of birds while notes are written. In forest and woodland, however, the narrow field of view combined with the rapid movement of birds renders telescopes virtually useless.

There are many makes and models of both binoculars and telescopes and in general you get what you pay for both in terms of reliability and quality of image. The most expensive binoculars and telescopes have very sharp resolution, are filled with gaseous Nitrogen to ensure a stable atmosphere around the interior of the lenses, are waterproof and some come with a lifetime replacement guarantee. However, there are very good binoculars to be had for a few hundred dollars, and good telescopes for around a thousand dollars.

Binoculars come in two basic styles depending on the arrangement of the prisms in the lens barrels; roof prism binoculars are more compact. Things to look for in binoculars and telescopes for wildlife-watching are:

Magnification and field of view

The magnification and size of binoculars are indicated by two numbers, such as 10 X 40. The 10 is the magnification and the 40 is the outer lens diameter. Most binoculars used by birdwatchers range from 8 X 40 to 10 X 40; more powerful binoculars generally have a narrower field of view, and a wide field of view makes it easier to find birds through the lenses. Telescopes commonly used by birdwatchers have a zoom lens in the range 20X to 40X or 60X. Most observers find themselves using their telescope at 20X, as with higher magnification there is considerable loss of field of view and resolution (sharpness of image), while heat haze even early in the morning also distorts the image, and this effect is worse at high magnification.

Eyepiece structure

This is an important and often neglected feature. Many observers wear spectacles or will be wearing sunglasses, and therefore the eye is kept away from the eyepiece of the binoculars or telescope. Look for binoculars/telescopes with a wide, soft eyepiece that allows glasses to be pressed as close to the lens as possible.

Left-right adjustment

In binoculars, it is usually possible to adjust the left and right lenses individually to compensate for slight differences between the eyes of the observer. This is essential if you do a lot of work with binoculars but means that other people may find the view through your binoculars slightly distorted. Setting the adjustment at neutral (assumes left and right eyes are the same) is a compromise when sharing binoculars.

Focussing

Most binoculars come with a single focussing wheel; this should move freely and be in a comfortable position. There are binoculars with separate left and right focus, with no focussing adjustment (the binoculars have an infinite depth of view and focussing is done by the eyes of the observer) and even with an internal movement suppression system (so that the image of the bird appears to stay still in the field of view with apparently no binocular movement). Binoculars with an unusual focussing system can take a bit of getting used to.

Minimum focus distance

Being able to focus on close subjects (less than about three or four metres) is a very useful facility, especially when looking at reptiles. A close focus can also help in bird observation or just enhance the enjoyment of seeing a bird. Expensive binoculars tend to have a smaller minimum focus distance than less expensive units.

Camera attachments

Some binoculars and telescopes can be attached to cameras with special fittings, and the quality of photographs can be good; the quality can also be poor. This capability may be a consideration for some observers.

(photo S. Cherriman)

BIRDS; AURAL SURVEYS AND CALL PLAYBACK

The calls of birds are often diagnostic and skilled observers rely heavily on call recognition to identify species during surveys. For a few species, however, aural surveys are the primary means by which they can be detected and censused, and in some cases the aural survey approach can be supplemented by playing back the call of the species to get a response.

Species for which aural surveys are important are generally cryptic and in some cases nocturnal: Western Ground Parrot, Australasian and Little Bittern, Noisy Scrub-bird, Western Bristlebird, Western Whipbird, Red-eared Firetail and Barking and Masked Owls. Call playback is especially useful for the Firetail and the two Owls, and is used in specialised capture techniques for the Noisy Scrub-bird and Western Whipbird.

Aural survey techniques for these species will soon be available in a guide to survey techniques for significant species (Bamford, Gamblin, Browne-Cooper and Turpin in preparation). These approaches require some training of personnel and familiarity with the calls of the species (training can include listening to recordings of the calls), and are usually undertaken as coordinated surveys involving government conservation agencies. However, anyone working within the range of these species should make themselves familiar with their calls. Times of day when these significant species may call are:

Western Ground Parrot – before sunrise and after sunset.

Australasian and Little Bitterns – around sunset and occasionally throughout the night.

Noisy Scrub-bird, Western Bristlebird, Western Whipbird, Red-eared Firetail – throughout the day but often at greater intensity in the first hour or so around sunrise.

Barking and Masked Owls – around sunset and occasionally throughout the night.

The use of call playback to assist in surveys of cryptic (and other) species should be limited. It does disturb the birds since it effectively involves playing the call of a potential rival in the territory of a resident bird. Call playback sessions should be short and should cease immediately if a bird shows signs of extreme excitement or distress, which probably means that a nest and/or young are very nearby. Call playback should not be used simply to attract birds into view.

SPOTLIGHTING/HEADTORCHING

Spotlighting usually refers to operating a strong, hand-held spotlight from a slowly-moving vehicle (commonly around 20kph) while looking for nocturnal fauna. It is widely used in some parts of the world where there is a rich fauna of nocturnal and arboreal mammals and birds, but the assemblage of such species is very poor in Western Australia. However, spotlighting can still be effective not just for birds and mammals, but for nocturnal reptiles and frogs. Often, more species may be seen on the road in the vehicle headlights than are seen with the spotlight. Spotlighting can also be used as a means for censusing feral species such as Foxes and Cats.

Spotlighting is usually carried out with a driver, someone operating the spotlight and often one or more observers. Greatest success for nocturnal frogs and reptiles is often achieved on warm to hot summer nights, often after rain, and often numbers are greatest on bitumen roads, probably because these retain warmth and the reptiles are basking on the surface. Spotlighting on bitumen roads therefore requires awareness of other vehicles. Mammals and birds are usually detected by their “eyeshine” due to the light from the spotlight reflecting back to the observer. Other species are usually detected by their movement.

A wide range of hand-held spotlights is available; most can run directly from the car battery (either via a 12V plug in the cabin or clipped to the battery). While this means the person holding the spotlight can’t easily leave the vehicle, such units are preferable to those with their own battery, as these tend to be very heavy to hold out through a car window for prolonged periods.

Headtorching is carried out on foot preferably with a headtorch, although a hand-held torch will do. If using a hand-held torch, it should be carried on the shoulder close to the eyes as this gives good eyeshine from animals; the light from the torch shines into the eye of the animal and directly back at the torch, thus the eyes of the observer must be as close to the source of light as possible in order to see the eyeshine. Probably the best headtorches currently available for fauna survey work are the LED Lenser HD series as these use very powerful LED lenses, have an adjustable beam width and strength, and run on 4AA batteries; they will also work on rechargeable batteries. Headtorching allows the observer to leave vehicle tracks, to scan slowly across the ground, over rocks and up and down trees, and is excellent for finding nocturnal reptiles, frogs and invertebrates. Spiders have very bright eyeshine; geckoes and frogs have dull eyeshine but with practice it can be recognised. As with spotlighting, the effectiveness of headtorching is very dependent upon conditions, with warm to hot nights being good for reptiles and even many frogs.

It is extremely easy to become disoriented when headtorching and therefore personnel should have a plan that involves staying in contact with each other, knowing roughly where they intend to walk, and having a return time to the vehicle. Leaving the parking lights of the vehicle on or placing a lamp on the roof of the vehicle can be helpful. If available, personnel can carry GPS units with the track setting on so they can retrace their steps if necessary. Carrying a backup torch and spare batteries is recommended.

Woylie *Bettongia penicillata* in the spotlight (photo Simon Cherriman).

REGURGITATED PELLETS AND OTHER REMAINS

Some birds of prey routinely regurgitate pellets of undigested material and these pellets can be found beneath roosts. In caves where successive generations of Barn Owls have roosted, there can be massive accumulations of such pellets. These pellets contain often recognisable bone fragments and can be used to confirm the presence (and past presence) of a range of mammal species. Identifying mammals from such fragmentary skeletal remains is a specialised task, but where such material is found it is well worth collecting a sample and taking to the DPaW or WA Museum. Note that permission from traditional owners may be needed before caves are entered and material removed. When collecting regurgitated pellets, note the location, describe the site and if possible keep recent and older material separate. Note that regurgitated pellets are usually dry and virtually odourless when more than a few days old.

The scats of some carnivorous reptiles and mammals can also contain bone fragments, although such scats tend to be scattered rather than accumulated beneath a roost as with bird pellets, so provide only a small volume of material. It can be worthwhile checking such scats when found. Becoming familiar with the bones of common small mammals, such as Rabbits, can make it easier to recognise bones of less common species when these are encountered.

Dead animals are occasionally found in the field and stopping to check on such corpses can be worthwhile. The only two specimens of the Night Parrot collected in the last 75 years were found dead and dehydrated: one on the roadside and one under a fence in outback Queensland. Fresh roadkill predators (Fox, Cat, large reptiles) are well worth dissecting. The Pygmy Bluetongue Lizard was rediscovered in the stomach contents of a roadkill snake (living animals were found nearby), while dissection of roadkill cats provides information on what they eat and on what native wildlife may be in an area (Figure 38).

A regurgitated pellet from an Eastern Barn Owl containing the bones of at least two rodents and a small bird (photo T. Gamblin).

Figure 38. A roadkill feral cat and its last meal of seven *Ctenotus fallens,* two *Delma greyii,* two *Strophurus spinigerus* and two honeyeater chicks. The *D. greyii* were very rarely recorded in a pitfall trapping program being carried out in adjacent bushland (photo M. Bamford).

SAMPLING METHODS FOR INVERTEBRATES

Unlike studies on vertebrates where specimens are almost always released after examination, or may be observed only as in bird censusing, sampling for invertebrates often involves the collection and preservation of specimens for later identification and sorting. Some sampling techniques are destructive (that is, specimens are killed by the sampling method), and by-catch of vertebrates is a major concern and needs to be considered when conducting invertebrate sampling.

Identification of invertebrates is a complex and specialised area, but non-specialists can at least assist experts in the field collection of specimens and then take these to invertebrate specialists for close examination. The following sections describe the main techniques for the collection of invertebrate specimens that are regularly used in Western Australia, and provides information on the minimisation of impacts upon vertebrates where appropriate. Boxes 4 and 5 provide details of specific and non-destructive sampling protocols for butterflies and day-flying moths, including two rare species, and for a species of trapdoor spider.

Pitfall Traps

Pitfall traps set for invertebrates operate in the same way as those set for vertebrates; that is, they are a passive interception trap that relies on invertebrates walking along and falling into them. However, pitfalls are usually used without drift-fences when sampling invertebrates. Key points to understand when using pitfalls for invertebrates are:

Pitfall diameter

Invertebrates will fall into pitfall traps of almost any size, and the large pitfalls used in vertebrate surveys are an excellent source of invertebrate specimens. However, small containers are usually used when sampling invertebrates to reduce the amount of digging required. Plastic specimen containers with a diameter of about 40mm (and either 50 or 100mm deep) are commonly used and have the advantage they can be removed from the ground and the lid can be screwed on immediately. An important reason for using pitfalls of small diameter is that it reduces the by-catch of small vertebrates, although even test-tubes used as pitfalls will catch small lizards and frogs. Pitfall traps set for invertebrates are often used with a preservative (see below) and if this is present, vertebrates can be inadvertently killed.

Wet or Dry

Invertebrate pitfalls are often used with a preservative so they can be left out for an extended period of time and any animals caught will be killed and preserved for later examination. As noted above, this can cause mortality of vertebrate by-catch. Preservatives used include 70% ethanol (usually with glycerol added to reduce evaporation) and Galt's solution. In addition to killing anything that gets caught, preservative may bias the sample by preferentially attracting some fauna groups. Dry pitfalls that have to be checked regularly are an alternative to pitfalls containing preservative (wet pitfalls) and have the advantage that many vertebrates can escape. However, so too can many invertebrates. Invertebrate pitfalls are often used in very large numbers (100s) and thus daily checking of this number of traps can be very demanding.

Soil disturbance and other biases

Some invertebrates, particularly ants, are attracted to freshly-disturbed soil around a newly-installed pitfall, and this can lead to their over-representation in samples (which may not be a problem if ants are being targeted!). A solution to this is to insert PVC sleeves into the soil that have an internal diameter just slightly more than the external diameter of the pitfall. The sleeve and capped pitfall can then be left *in situ* for a few days while disturbance effects abate, and only then can the lid to the pitfall be removed for sampling to commence. The use of the pitfall/sleeve setup is very good for long-term monitoring when sampling might be carried out for just a few periods each year. Other biases can be due to the type of preservative used, which will either attract or repel some species. Even the colour of plastic used may affect captures.

Season

Pitfall captures of invertebrates are reliant upon activity, and invertebrates are highly seasonal in their activity levels. Overall, activity is greatest when conditions are warm and at least some soil moisture is present; for example in the South-West, activity levels peak in spring to early summer. However, some groups have different annual cycles. The males of trapdoor spiders often have a particular season when they move in search of females and this can be in autumn or winter; or may be in spring. Other invertebrates (slaters, millipedes) may only be active in the cool, wet winter months in the South-West.

Length of sampling

This may depend upon logistical and other planning factors, but sampling periods for invertebrate pitfalls are usually in the order of 5-10 days.

Examination of samples

Invertebrates collected in pitfall traps are commonly mixed up with sand, twigs, leaves and each other. Sorting is usually carried out in a laboratory using a dissecting microscope, with the invertebrates initially being roughly sorted into major taxonomic groups (eg. ants or beetles) before being very closely examined so that they can be sorted more precisely. Even if they cannot be identified to species, they can still be sorted into groups of specimens that appear to belong to one species. Species identification may not be possible because the specimens belong to an undescribed species, or because no experts in a particular group are available to carry out identification.

A scorpion *Urodacus* sp. and a pie-dish beetle in a pitfall set for vertebrates (photo A. Bamford).

Leaf-litter Samples

Leaf-litter samples are an excellent source of invertebrates that might be too small or slow-moving to be caught in pitfalls. Groups such as small land snails and pseudoscorpions are often well-represented and can be of taxonomic interest. Leaf-litter samples are most useful when collected at the right time of the year and from the right location, and require an extended sorting period to extract specimens. Considerations are discussed below.

Type and amount of material to collect

A single leaf-litter sample typically consists of a bolus of material about the size of a football, with the material slightly compressed into a well-sealed plastic bag. The material should consist of litter, friable humus and even some surface mineral soil. Several litter samples should be collected from each location, but note that litter samples require a long period for sorting. Half a dozen bags may be sufficient to occupy sorting facilities for several days.

Source of material

Litter samples should be collected in mesic refugia such as between logs or rocks, in a hollowed-out stump or from the base of a hollow tree. Litter can also be collected from inside small caves. A mixture of soil, kangaroo scats and plant fragments from the back of a cave may also harbour unusual invertebrates, particularly if moist but even if dry.

Season

Litter samples are usually collected in the moist time of the year (winter in the south, usually summer elsewhere), as this is when litter invertebrates are most likely to be active.

Handling of samples

Samples should be kept away from extreme temperatures. Invertebrates will remain alive in a sealed bag for several days if not exposed to heat and if the material doesn't dry out.

Examination of samples

Some sorting can be carried out by hand, such as by spreading the material across a white tray and looking for invertebrates moving about, but the primary phase of sorting is to extract specimens from the litter in a Tulgren Funnel. Tulgren Funnels direct heat at the top of a sample that is placed in a funnel, and the invertebrates move down through the material to escape the heat and dry conditions, eventually falling from the spout of the funnel into preservative. They can then be separated from sand, twigs and leaves, and examined more closely for identification.

Light Traps and Black Light

Many winged insects (moths, beetles, crickets) and even some non-flying invertebrates such as centipedes are attracted to lights, and this is widely used in sampling. Light traps are most effective at warm times of the year when insects are most active, and typically consist of a source of light (such as a UV tube powered by a battery), with baffles over a collecting chamber filled with material such as scrunched newspapers; insects fly towards the light, strike the baffles and drop into the chamber, where they shelter in the newspaper. There are both commercially available and home-made light traps of this sort. Another home-made light trap is as simple as a light directed onto a white sheet; this is very effective for moths than can simply be plucked from the sheet.

Black lights are fluorescent tubes that produce ultra-violet light. These can be used to actively search at night for scorpions which fluoresce ('glow') under black light.

Sampling Vegetation for Invertebrates

Many invertebrates occur on vegetation: amongst flowers, on foliage, under or on bark and even inside wood. Flowers and foliage are an especially rich source of invertebrates and collection techniques are variations on the theme of somehow dislodging specimens so that they fall onto a collecting surface. This can be as simple as laying a white sheet on the ground and shaking or beating a bunch of foliage so that invertebrates fall onto the sheet. Foliage can also be fumigated with insecticide to dislodge invertebrates. Working in the canopy and using a cherry-picker for access, the white sheet can be replaced with a collection surface such as an upside-down, white umbrella that can be suspended below the foliage with a collection container fitted into the apex so that specimens fall into it.

Bark-dwelling invertebrates can be especially cryptic and while they can be found by searching, this can be destructive of the micro-environment. An alternative that is also quantitative, in that it samples a roughly defined area, involves the installation of what are effectively small drift fences into the side of the tree, which guide invertebrates into containers containing preservative.

Hand Collection

Invertebrates such as land snails, trapdoor spiders, millipedes, slaters and scorpions can be found simply by searching, and are taxa that are often of great taxonomic interest. Snails, millipedes, slaters and sometimes scorpions can be found under rocks, logs and even loose bark, although under warm, moist conditions snails, millipedes and slaters can be found active even during daylight hours. If searching under rocks and similar, the material should always be replaced to restore the micro-climate. Many scorpions excavate deep burrows that can be excavated to secure the animal. Scorpion burrows typically spiral downwards; some desert species in red sandy loams can reach depths of over a metre.

Aquatic Invertebrates

This is a broad area of sampling. Using an assortment of nets and other devices, aquatic invertebrates can be sampled from the water column (ie. swimming or drifting effectively mid-water), from submerged aquatic vegetation and from the benthos (the floor of the lake or stream). Mud-samples taken with an auger can be sorted for benthic invertebrates. Aquatic invertebrate assemblages and the means used to sample them can differ greatly depending upon the type of wetland environment. For example, more or less static water bodies (lakes and swamps) can have abundant invertebrates in the water column and a deep, soft benthos containing burrowing species. In contrast, fast-flowing streams may have only benthic invertebrates that cling on or under rocks and gravel, requiring a quite different approach to sampling.

Subterranean Invertebrates

Invertebrates that live underground, such as in soil cavities, crevices within rock or even cave systems, can be divided into stygofauna (effectively aquatic species present in flooded subterranean environments) and troglofauna (present in subterranean environments that are not flooded). Subterranean fauna are often associated with particular geological formations (such as limestone 'karst' topographies), but can occur anywhere that suitable subterranean environments occur. Subterranean fauna can often be of great taxonomic interest, with relictual species reflecting past fauna assemblages on the surface that have survived in a constant environment while terrestrial ecosystems change over periods of tens of millions of years. Subterranean fauna are also very hard to sample, often requiring a drilling program with heavy equipment to depths of tens of metres. Even once this is done, the animals may occur at such low densities (because subterranean ecosystems are very unproductive as they rely on a trickle of organic material from above) that long periods of time are required to collect even a few specimens. Specialists in the area often deploy traps (consisting of containers filled with leaves) that are left deep in a drill-hole and drawn back to the surface to check for subterranean fauna that may have colonised the bunch of decaying foliage. Drill holes created to sample for subterranean fauna, or as part of geological exploration, should always be well-covered to prevent terrestrial fauna falling into the hole.

Netted Dragon sampling a large moth (photo A. Bamford)

BOX 4

Survey methodology for butterflies and day-flying moths

TRAINING

Currently, surveys are commonly conducted for either the Graceful Sun-Moth (GSM, *Synemon gratiosa*, formerly listed as Endangered, now Priority 4) or the Arid Bronze-Azure Butterfly (ABAB, *Ogyris subterrestris petrina*, listed as Critically Endangered under WA legislation, and currently being assessed Federally), but the sampling protocols for these species may be applicable to other butterflies and day-active moths. Importantly, the protocols are intended to provide confidence regarding the presence and abundance of these species. Only people who have completed a DPaW training course for the GSM or ABAB can lead field surveys. A trained person must have planned the survey and be present in the field for the duration of the survey. The survey protocol for each species is based on the standard butterfly walk transect method, discussed below, which is the internationally recognized method of surveying for the presence and abundance of butterflies and day-flying moths. This method is used at more than a thousand butterfly monitoring sites throughout Europe and North America (see Pollard and Yates 1993, or visit the UK butterfly monitoring website at http://www.ukbms.org). In addition to transect surveys, a program survey for the ABAB must include surveys for the host ant, *Camponotus terebrans* (for further information see Gamblin *et al.* 2009; Williams and Williams 2005).

SURVEY METHODOLOGY

The methodology and development of transect monitoring for butterflies is reviewed in detail elsewhere (Pollard and Yates, 1993, Williams 2008; Williams 2009). In brief, a fixed-route walk (strip transect) is established at a site, and butterflies are recorded along the route on several occasions at regular (usually weekly or two weekly) intervals under reasonable weather conditions (Williams 2008). In south-west WA, 'reasonable weather' is considered to be days with fine weather and forecast maximum temperatures above 21°C, with a wind speed of 5m/s or less. Sampling should be conducted between 1000 and 1500 hr during the main flight season of the targeted species. For the GSM, surveys should be conducted from mid-February through to late March. For the ABAB and virtually all butterfly species, surveys in late September through to mid-October are ideal, but can be conducted at other times. Specific advice on survey guidelines for these species should be sought.

At each site, an aerial photograph and/or vegetation map is used to determine the location of tracks and firebreaks, major vegetation types, landforms and fire history. Using this information, a transect route is determined in advance that includes as many of these features as possible. Both recently and long-unburnt areas should be examined (Bishop *et al.* 2010). Transect routes are chosen to sample all of the habitat types and management activities on each site. Care must be taken in establishing a transect route as it must then remain fixed to enable butterfly sightings to be compared from year to year (UKBMS). Transects are typically about 1-5km long, take between 45 minutes and two hours to walk, and are divided into sections corresponding to different habitat or management units (UKBMS). Each transect can be divided into sectors approximately a hundred metres long, with boundaries that coincide with prominent features such as crossroads or changes in vegetation type. Transect route(s) and length(s) should also be recorded for the site (Bishop *et al.* 2010). Butterflies/moths are identified and tallied within a fixed-width band (5m either side of the transect mid-line, i.e.10m wide) (Williams 2009). A regimen of 10-m-wide walk transects sampled on six occasions at 2-week intervals during the austral spring (mid-September to mid-December) will give an almost complete inventory of resident butterfly species for each site (Williams 2008).

Data recorded for each transect should include the following information: weather conditions (wind speed, temperature (C), and average cloud cover), timing (date, start and finish times), and the number and identity of each observer.

With patience and stealth, sun-moths can be captured after settling on the ground (photo A. Williams)

(photo M. Bamford)

BOX 5 *Non-destructive sampling of a spider*

BACKGROUND

Approval of the Karara Mining Limited (KML) Iron Ore Project (located approximately 320km north-northeast of Perth, in the southern Murchison region) included a commitment to document the regional distribution of and establish a monitoring program for the Shield-backed Trapdoor Spider *Idiosoma nigrum*. The Shield-backed Trapdoor Spider was found to be abundant on ironstone rocky hills and mid to lower slopes on Karara and adjacent stations (Bamford 2006; Bamford and Metcalf 2008 ; Bamford and Turpin 2009). To meet KML's commitment, regional surveys, density estimation and intensive monitoring of marked spider burrows is carried out annually.

The commitment was required because the species was likely to be impacted by the project and is listed under Schedule 1 of the *WA Wildlife Conservation Act* (1950), and, as of early 2013, as Vulnerable under the *Environment Protection and Biodiversity Conservation Act 1999* (EPBC Act). These listings are due to its limited geographic distribution and the nature of ongoing threats (TSSC, 2011). Threats include altered fire regimes, habitat degradation due to grazing, salinity/altered hydrology, habitat fragmentation and impacts from competing land use, particularly iron ore mining.

The Shield-backed Trapdoor Spider is a long-lived species; females can live for over 20 years. Like many mygalomorph spiders they have limited dispersal capabilities (Harvey 2002), remaining in their burrows and only venturing short distances to find food, except in the breeding season when adult males disperse up to 1km (and subsequently apparently die). Emergent spiderlings generally establish their burrows within a metre of the matriarch, forming a family or 'matriarchal cluster' (Figure 39), but Bamford and Metcalf (2008) suggested that the presence of isolated animals may be due to male spiderlings dispersing further than females. Hence matriarchal clusters consist of a large (matriarchal) burrow surrounded by several (sometimes over 20) juvenile burrows of varying ages.

Burrows of the Shield-backed Trapdoor Spider are distinctive, with a fan of twigs, leaves or phyllodes radiating around 180° or slightly more from the burrow entrance, usually with a slightly clumped (moustached) effect on opposite sides of the entrance, and the lid is decorated with mostly horizontally-aligned but sometimes vertically-aligned twigs (Figure 39). The burrow has a distinct constriction at a depth of 3-5 cm; this is the point that the spider blocks with its armoured abdomen. Therefore, burrows rather than the animal itself are monitored, though where uncertainty occurs a milliscope can be used to check for spider presence. Identifying and recording burrow data (diameter relates to age class) enables detailed information on population structure, recruitment and survival to be gathered (Bamford *et al.*, 2012; Main, 2003). In repeat monitoring surveys, previously recorded burrows are scored as 'located' or 'not located' and where located, data recorded. It is assumed that burrows not located are where the spider is no longer present (ie. has become uninhabited and degraded or eroded; or has been destroyed by natural or other means).

SURVEY METHODS

The spider monitoring program was established to evaluate:

(1) presence/absence of populations (distribution)

(2) population density and

(3) impacts from mining operations (noise, dust and vibration) such as changes in recruitment, mortality and growth.

An array of sampling methods and statistical analyses were reviewed in the literature and with the addition of expert advice on methodology (Williams, M.R, pers comm) a subset of these were trialled in field investigations, resulting in a suite of methodologies considered robust and efficient for this species. Methods used in the program are outlined below.

DETERMINING DISTRIBUTION

Early in the study of the species, it was found that they were most abundant in shrublands on the slopes of ridges where the soils were a gravelly to rocky loam. The species thus occurred in sub-populations associated with ridges, and was absent from intervening plains of loam to clay soils supporting York Gum Woodland. To determine the distribution of a population on a ridge, transects were laid out that started from the adjacent plains, extended across the ridge and then terminated on the plains on the opposite side. Several such transects were selected across each ridge, with the spacing between transects ranging from 200 to 500m. Quadrats (initially 10 x 10m, later 2 x 50m) were then placed at regular intervals (50 or 100m) along each transect and were searched for spiders by a team of usually four people. If the aim was simply to determine presence, then 10 x 10m quadrats were used and the boundaries of the quadrat were often only approximate. Searching was limited to 20 minutes; after 20 minutes the team was confident that if no spiders were found, the species was not present. This was a presence/absence search. Where density information was also required, 2 x 50m transects were searched and a very rigorous approach was used to find all spiders. This approach is discussed below.

DETERMINING DENSITY

Quadrats of 2 x 50m were used to determine density because it was found that the narrow quadrat could be searched more accurately and rigorously than the 10 x 10m quadrat. Because the distribution of spiders is strongly clumped in matriarchal clusters, the long, narrow quadrats are less likely to be biased in the determination of density than the square quadrats, which have a greater probability of completely missing or completely including whole clusters. Furthermore, in dense vegetation, establishing the boundaries of a 10 x 10m quadrat is difficult, whereas the 2 x 50m approach meant that a tape measure could be used to ensure that personnel adhered strictly to the survey area. The 2 x 50m transects were surveyed by four people, working in pairs from each end, so that each person is searching a strip only 1m wide. The distance of spiders along the quadrat is recorded and measurements and notes made on each burrow (see below).

MONITORING IMPACT

While direct impacts from clearing are unavoidable, indirect impacts from disturbance (such as dust, noise and vibration) are being monitored by repeat visits to marked matriarchal clusters. Impact (close to sources of disturbance) and control (at least several hundred metres from disturbance) clusters are permanently marked with a GPS coordinate and two labelled metal pegs, and the location of each burrow in relation to these pegs is determined with a distance and a bearing (Figure 40). This makes it possible to re-find each burrow or, upon searching, to be confident that a burrow has disappeared. Such matriarchal clusters are re-visited annually with every burrow being re-located (or noted as not present), every burrow found measured (internal diameter to the nearest 0.1mm), and every new burrow being recorded and measured. New burrows are recognised as such because the area of the matriarchal cluster is searched very thoroughly on each visit (personnel often lie down to view the ground closely; Figure 41), and the search area of each cluster is defined either by additional pegs, or by the distance to the burrow furthest from the origin peg being used as the radius of the cluster search area.

Figure 39. (A) Matriarchal cluster of Shield-backed Trapdoor Spiders; there are at least eight burrows present (photo M. Bamford). (B) Burrow of Shield-backed Trapdoor Spider, with trapdoor open (photo M. Bamford).

Figure 40. A marked matriarchal cluster set-up for re-locating burrows. The 360° protractor is placed over the origin peg, with the second peg placed at 0° (approximately north). The tape measure is then used to give the distance and bearing to each burrow (photo M. Bamford).

Figure 41. Recording all the burrows in a matriarchal cluster. Each piece of pink tape is placed upon a burrow (photo M. Williams).

MAKING OBSERVATIONS COUNT

Once a specimen has been caught, it is prudent to collect as much information from that individual as possible before release. Specimens need to be accurately identified, while a lot of other information can be collected that helps to develop an understanding of the biology of a species and the status of a population.

Identification

Accurate identification of specimens, whether a bird flying past or a lizard in a pitfall trap, is an essential part of fauna studies. While the abundance of a lizard species between sites or over time can be examined without knowing the lizard's Latin name, it is not possible to source other information on the lizard, compare results with other studies or even determine if it is a species of conservation significance. Identifications that are uncertain should always be recorded as such; there is little value in an identification that appears to be certain but which, in reality, is little more than a guess!

Identification is a matter of experience, including experience in using resources that aid in identification. Invertebrate identification is best left to specialists, although there are some books that help non-specialists at least recognise major invertebrate groups (eg. Harvey and Yen 1997), and some illustrated guides that provide information on a few of the most conspicuous invertebrates in particular regions (eg. Williams *et al.* 2009, Daniels 2011). For vertebrates, there are books (and other resources such as recordings of calls of frogs and birds) that aid in the identification of all groups. Some notes on how to approach identification in each major vertebrate group are presented below.

FRESHWATER FISH

Morgan *et al.* (1998) provide details on the freshwater fish, including introduced species, of the South-West region. Allen *et al.* (2002) cover the entire freshwater fish fauna of Australia.

FROGS AND REPTILES

The Western Australian Museum has produced a series of identification guide books to the frogs and reptiles of Western Australia (Storr *et al.* 1983, 1990, 1999 & 2002; Tyler *et al.* 2000), although unfortunately some of these are out of print. There have also been recent taxonomic revisions that are currently only available from the scientific literature. The books rely heavily on the use of dichotomous keys and details of external morphology for identification, but also provide photographs. The keys in the reptile books require familiarity with the names of head shields (enlarged scales on the head, see Figures 42 and 43) and details of foot structure, but at least some explanatory illustrations are provided. It can take some practice to be able to use these keys and this can only be gained through experience; especially through working with someone who is already familiar with the books and the reptile fauna of Western Australia. Wilson and Swann (2003) is a single volume that covers all Australian frogs and reptiles, and while it lacks dichotomous keys, the photographs and concise text make this very helpful in the field. There are also some local guides that provide a lot of information on species within a region (eg. Bush *et al.* 2010 cover the frogs and reptiles of the Perth region).

The calls of frogs are species specific and surveys for frogs can be based on calls alone (page 35). There are some commercially available collections of recordings of frog calls, and recordings are also available at: http://museum.wa.gov.au/research/collections/terrestrial-zoology/herpetology-reptiles-and-frogs-collection/frog-calls.

A selection of these books for the field identification of frogs and reptiles is recommended, and it is useful to have copies of scientific papers with recent taxonomic updates as a supplement. In some situations, specimens or tissue samples may need to be collected to confirm identification and this is discussed below (page 54). It is also very helpful to take photographs of specimens before release, particularly if there is some uncertainty about an identification, or if the species to which a specimen is believed to belong is not considered to occur in the study area based on distribution maps in books. To be useful for identification, clear photographs should be obtained of the dorsal and lateral views of the head, dorsal and lateral views of the body, and views of any other features that were used when carrying out the identification. Note should also be made of the characters used when keying out the specimen (such as the number of fingers and toes).

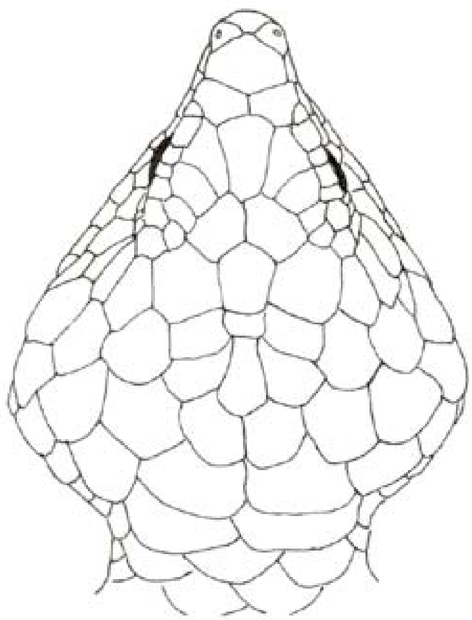

Figure 42. The arrangement of the head shields of reptiles are often used in identification keys (Bobtail *Tiliqua rugosa*). The shields are named and keys are provided in most identification guides. (illustration M. Bamford)

Figure 43. Examining the head-shields of a small skink with aid of a hand lens. Binoculars used back-to-front also work as a magnifying glass. (photo A. Bamford)

BIRDS

There is a wealth of bird identification guides available and to some extent "the best" comes down to personal familiarity and preference. Guides that are most useful are those with clear (even slightly stylized) illustrations, precise, informative text and sufficiently compact to be useful in the field. Paintings rather than photographs still provide the most useful visual reference for identification and are used in almost all guides, although there are some photo-based guides available. There are also 'coffee-table' bird books that are lavishly illustrated but impractical for the field. To supplement standard hard copy field guides, there are now apps available for smart phones that provide text, illustrations and even calls, while recordings of calls are available separately. The eight volume 'HANZAB' (Handbook of Australian, New Zealand and Antarctic Birds, published from 1990 to 2006) provides tremendous detail on distribution, plumage, diet, reproduction and behaviour, and photocopies of relevant sections can be useful in the field if targeting particular species. Johnstone and Storr (1998, 2004) is a pair of volumes with information on Western Australian birds, including colour plates of the eggs of almost all species. The most commonly-used field guides are: Slater *et al.* (1989); Pizzey and Knight (2012), Simpson and Day (2004) and Morcombe (2000). At least the latter is also available as a phone app. Serventy and Whittell (1976) is an old book on Western Australian birds that is good for natural history, and is one of the few books to document traditional (Aboriginal and colloquial European) names for birds.

MAMMALS

Mammals present a challenge for field identification. There are field guides with paintings (Menkhorst and Knight 2001) and photographs (Jones and Parish undated), and Van Dyck and Strahan (2008) is almost a handbook that can be carried in the field. Churchill (2008) is a field guide that deals only with Australia's bat fauna.

Many of the smaller species of rodents and dasyurids can be difficult to distinguish without being familiar with the species, and the available guides do not always provide the useful identification tips that come from such familiarity. Some experience, such as recognising the smell and the head to body proportion of the introduced House Mouse compared with the smell and "large headedness" of native rodents of about the same size, can only be gained from time in the field handling animals.

For rodents and small dasyurids where identification may be uncertain, detailed notes, measurements (see page 48) and photographs should be taken. Photographs should include whole of animal (dorsal and lateral, including tail in at least one image), belly (especially if this is distinctly marked in any way, or has a sharp transition in colour from the dorsum to the venter) and the underside of the hind feet. The latter often have patterns of small tubercles that are species specific, especially in some rodents. Dentition is occasionally important and this may be mentioned in field guides (such as the number of pre-molars in Mulgara *Dasycercus* spp.). The presence of a notch in the upper incisors on the House Mouse is mentioned in guides and is diagnostic, but can be difficult to see and some individuals have been reported in which the notch is virtually absent. Tissue samples can be taken from mammals and used for DNA identification as discussed on page 54.

Experienced field-workers can accumulate a reference collection of their own (and shared) photographs as an aid to identification. Figure 44 illustrates two species of planigale; similar in size but one is slightly smaller-eyed and they have distinctive pelage. However, if either one of these species is caught without the other, they can be difficult to recognise, while such subtle differences are rarely clear in guide illustrations.

Figure 44. Two recently described species from the Pilbara: *Planigale tealei* (left) and *Planigale kendricki*. The comparison provided by this photograph is a useful reference if either species is caught, although a scale would have been useful (photo J. Vos and J. Raines).

Measurements, age, reproductive condition and other observations

Measurements and a range of observations of each specimen can contribute a great deal to the scientific value of handling or even observing a specimen. For example, skilled observers can age migratory shorebirds from a distance (through a telescope) by examining the relative length of the tertial feathers, making it possible to calculate the proportion of juveniles in a flock and therefore the breeding success of the species in the high Arctic several months before. Similarly, simple body length measurements in frogs and reptiles allow for the recognition of age cohorts and therefore whether or not a population is recruiting or consists only of old animals. This may be very significant if monitoring populations in disturbed or rehabilitated environments, or if studying a threatened species.

MEASUREMENTS AND OTHER OBSERVATIONS: FROGS AND REPTILES

There is a suite of basic measurements that can be taken on frogs and reptiles as outlined below.

Weight

Specimens can be weighed using a fine spring balance (Pesola brand are reliable, robust and come in a range of sizes from 10g to 1500g) or a small digital balance. Small frogs and lizards can safely be restrained for brief periods in a small plastic bag (zip-lock sandwich bags are excellent for this; see Figure 45), but remember to delete the weight of the bag. Record weight to one decimal place for very small reptiles and frogs, but to the nearest gram or even 10 grams for larger specimens. Weight provides a measure of condition for animals of the same species and size when expressed relative to a standard length measurement, but beware with frogs as they can be slightly dehydrated which will affect their weight.

Snout-to-Vent Length (SVL)

Effectively head and body length, taken from the tip of the snout to the vent (cloaca). On frogs this can be taken with calipers, while small reptiles are best pressed against a clear plastic ruler (Figure 46). Try to ensure that animals are not hunched or twisted as this will reduce the measurement. SVL (and other linear measurements) are usually recorded in millimetres as a standard; this avoids possible later confusion that can occur if some people use centimetres. Be realistic with the significant figure: SVL is usually recorded to the nearest millimetre or even ten millimetres for a large and wriggly reptile, but some other measurements may be taken to one decimal place (see below). SVL is a very powerful measurement for understanding population structure of frogs and reptiles. Except in aseasonal climates (some parts of the tropics), frogs and reptiles breed seasonally and experience annual variations in growth rate. Therefore, at any one time of the year, it is possible to distinguish first year animals (Year 1), and often second year animals (Year 2), from older animals (Year 3+). Figure 47 illustrates the difference between a population of the Moaning Frog *Heleioporus eyrei* in which recruitment has occurred (good representation of Year 1 cohort), compared with a population in which recruitment has not occurred (no Year 1 cohort).

Figure 45. Weighing a small lizard with a Pesola spring balance (photo A. Bamford).

Figure 46. Measuring SVL on a small lizard. The specimen is restrained firmly but gently against the ruler (photo A. Bamford).

Frequency distribution of snout-vent length in two samples of the Moaning Frog

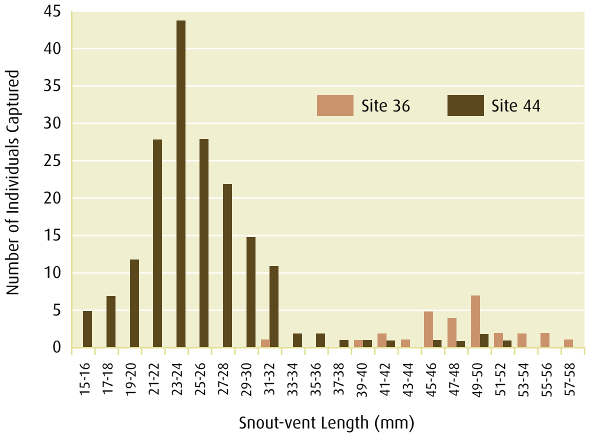

Figure 47. Site 44 was a wetland with annual recruitment of young frogs. Site 36 was a wetland where almost no recruitment occurred (from Huang and Bamford 2012).

Total Length (TotL)

This is the length of a reptile from the tip of the snout to the tip of the tail. It is therefore SVL plus Tail Length. TotL is not a particularly useful measurement as in species that drop their tail as a defence, the measurement can be greatly biased, and even in taxa that do not drop their tail (eg. agamids, varanids and snakes), the tail can be damaged. If TotL is recorded, the tail condition must also be noted (see below).

Other measurements

Three additional measurements sometimes taken on reptiles and frogs are: rear leg length, head width and head length. Other measurements can also be taken to suit requirements of particular studies (such as length of longest toe, distance between the eyes, height and width of the ear aperture, diameter of eye). Measurements other than the standard weight, SVL and TotL can be used to understand and compare the ecology of species, and can also be useful in identification. For example, rear leg length as a proportion of SVL will differ between similar species that forage in different environments, and may even differ between populations of the same species. The Sand Frog *Heleioporus psammophilus* has a shorter, rounder head, but more protuberant eyes, that the similarly-coloured Moaning Frog, and measurements may be useful in distinguishing these two. Likewise, the skinks *Ctenotus fallens* and *Ctenotus australis* are very similar in appearance, but *C. australis* has noticeably larger eyes (M. Bamford pers. obs). This feature is not recorded in guides but is very helpful, especially when dealing with mature specimens in which the colour patterns are slightly faded and can be indistinguishable. The larger eyes of *C. australis* also suggest that its feeding ecology differs from *C. fallens*. Observations such as this are accumulated by experienced observers.

Sexing

In some frogs and reptiles, there are clear differences in morphology and/or colour between the sexes. These are often described in guides. Many agamids and some skinks have seasonal breeding colours in males, while in all geckoes, males have a post-cloacal bulge. In some frogs (eg. the Western Spotted Frog *Heleioporus albopuntatus* but only some populations of the Sand Frog *H. psammophilus*), males have nuptial spurs on the inside of their forefeet during the breeding season. Male varanids often evert their hemi-penes when handled, and it is possible to make male skinks and dragons extrude their hemi-penes by applying firm pressure to the base of the tail. This requires some skill and there is potential to cause injury, so needs to be demonstrated by an experienced field worker. In some groups, males can be distinguished simply through careful observation of the shape of the underside of the base of the tail. This area is squarish and slightly enlarged in males, compared with rounded and not noticeably enlarged in females. Female reptiles can be distinguished when gravid (it can even be possible to count the number of eggs), and in female frogs the eggs are often visible through the translucent skin of the belly.

Other observations

Make note of injuries; missing toes and scars can be used for later recognition of the same animal in long-term studies. If the tail is regrown this should be recorded as not only does this affect the TotL and weight, but the proportion of animals in a population with regrown tails may give a measure of the predation pressure being experienced. Record anything that might be useful later. For example, deformities are occasionally encountered, such as frogs with missing or additional limbs, and these may be linked to pollution.

MEASUREMENTS AND OTHER OBSERVATIONS: BIRDS

Measuring, aging and sexing birds is a large field that is well-documented in the literature related to bird banding and bird research (Lowe 1989, Rogers *et al.*1986, de Rebeira 2006). Anyone handling and aging birds regularly is likely to be doing so under an ABBBS project and will have access to this literature, and will have been trained under the ABBBS program.

MEASUREMENTS AND OTHER OBSERVATIONS: MAMMALS

There is a suite of basic measurements that can be taken on mammals as outlined below.

Weight

Small species can be weighed using a fine spring balance (Pesola brand are reliable, robust and come in a range of sizes from 10g to 1500g) or a small digital balance; there are also large scales (similar to those used in butchers' shops) that can be used for mammals in the range of tens of kilograms. Mammals can be restrained in cloth bags for weighing; the Pesola balances may be more convenient for weighing small animals in bags than digital balances, which tend to have only a small pan onto which to place the bag; but a small, torpid marsupial can simply be placed on the pan (Figure 48). Remember to remove the weight of the bag from the measurement if necessary. Record weight to one decimal place for very small mammals, but to the nearest gram or even 10 grams for larger specimens. Weight provides a measure of condition for animals of the same species and size when expressed relative to a standard length measurement.

Body Length

This is a commonly cited measurement in guides, but especially small mammals are so flexible and gymnastic that obtaining an accurate Body Length is very difficult.
It is taken from the tip of the snout to the base of the tail, with Tail Length taken separately.

Crown

This is taken from the tip of the snout to the back of the skull, using calipers (Figures 49 and 50). It is a fairly robust measurement in that it can be taken to a high degree of accuracy as long as the calipers are placed against the back of the skull and are not pressed against a shoulder blade or fold of skin. It is useful as it allows juveniles to be readily separated from adults on the basis of a measurement, providing a measure of recruitment in the population. In most rodents, dasyurids and bandicoots, juvenile animals are more or less independent and can be caught separately from adults long before they reach adult size. Crown can also be used to separate species, at least in particular locations where the species and their measurements are known. For example, the House Mouse has a crown that rarely exceeds 22.5mm as an adult, and is sympatric with a variety of native rodents of more or less similar size, however, such native rodents (*Pseudomys spp.*) have proportionately larger heads than the House Mouse. Thus, in the South-West where the House Mouse and the Noodji or Ashy-grey Mouse *P. albocinereus* may both be present, the latter will have a crown of 28-31mm as an adult, and newly independent juveniles have a crown almost always greater than 23mm. In the arid zone, where the Mingkiri or Sandy Inland Mouse (*Pseudomys hermannsbergensis*) coexists with the House Mouse, adult Mingkiri will have crown measurements >24mm. Only juvenile Mingkiri can be confused with the House Mouse.

Figure 48. Weighing a torpid Honey Possum on a digital balance (photo T. Gamblin).

Figure 49. Measuring the crown of a Honey Possum. The animal is restrained by forming a cage around its body without putting pressure on it; Honey Possums do not bite and are thus one of the easier small mammals to measure (photo T. Gamblin).

Figure 50. Measuring the crown of a Western Pygmy-possum. This specimen is being held firmly by the base of the tail and has hold of a fold of skin in its mouth. It is best not to get bitten even by small animals that are unlikely to break the skin, but in this case the opportunity to get a measurement presented itself (photo T. Gamblin).

Pes

This is the length of the foot (Figure 51), from the back of the heel to the end of the toes (Short Pes) or ends of the nails (Long Pes). Pes width can also be taken, and can either be the minimum or the maximum width. Short Pes has the advantage that if the nails are damaged, the measurement is still accurate and comparable. Like Crown, Pes is a fairly robust measurement, although it can be difficult to take accurately on small mammals. Pes is useful in identification. Some small mammals, such as members of the dasyurid genus *Sminthopsis*, are similar in external appearance but have dissimilar pes morphology. On the sandplains north of Perth, *S. griseoventer* has a short, broad pes, whereas *S. aff. dolichura* is an animal of much the same size and appearance, but with a narrow pes (M. Bamford unpubl. data). This is a good example of where taking measurements led to the realisation that two species, one currently not fitting the description of any known species, were present.

Figure 51. Measuring the Pes of a small mammal (House Mouse *Mus musculus*). Note that the animal's head is shielded by the cloth bag to keep it calm (photo A. Bamford).

Other measurements

A range of other measurements can be taken on mammals, some taxon-specific. For example, measurements taken on bats, largely as an aid to identification, include forearm length, tragus length (the tragus is part of the outer ear in mammals that is very well-developed in many bats) and even the distance between the canines. Gonad measurements (scrotal length and width) can be taken on male marsupials but are more difficult to obtain on male rodents as the testes can be withdrawn from the scrotum when under stress. The width of the base of the tail may provide a condition index in some marsupials that are known to store fat in the tail.

Sexing and reproductive condition

Apart from external genitalia (often absent from rodents) and the presence of a pouch in female marsupials, sexual dimorphism is poorly-developed in most Australian mammals. Despite this, marsupials are easily sexed as the testes are generally very conspicuous (Figure 52), while a pouch may be present in females. Rodents are more challenging as males can withdraw their testes into the body cavity at will, and outside the breeding season the testes are permanently within the body so there is no evidence of a scrotum. Adult female rodents only have a conspicuous vagina during the breeding season, making it clear when they are sexually active but removing external characteristics at other times. Juveniles have no external genitalia. Outside the breeding season, rodents can only be sexed by noting the distance between the anus and the opening of the urethra: it is greater in males than females as it is in that space that the scrotum forms to accommodate the testes during the breeding season. It requires great familiarity with a species to sex juvenile and sexually inactive rodents based upon the anus-urethra distance.

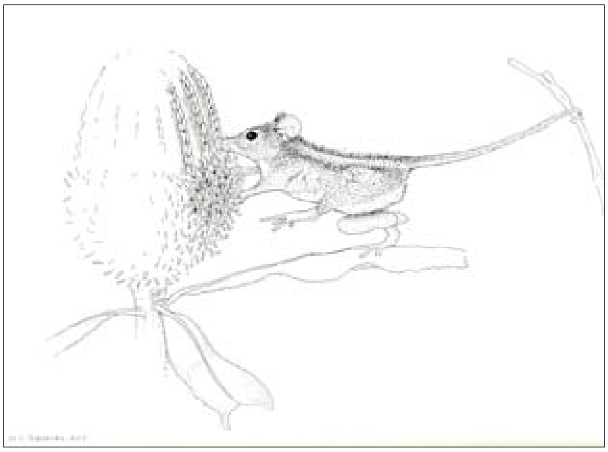

Figure 52. A male Honey Possum, illustrating the very large external genitalia typical of male marsupials (illustration M. Bamford).

Pouch condition

External development of the young in marsupials provides a wonderful opportunity to collect data on reproduction that cannot be done with Eutherian mammals. Young can be examined, counted and measured - when very small, the distance from the top of the skull to the rump (termed crown to rump) can be estimated, reflecting the hunched shape of small pouch young, while in larger pouch young measurements such as Crown and Pes can be recorded. The young can even be sexed from a very early age. However, care needs to be taken with pouch young so that they are not dislodged (page 28). This is typically a problem when the young are becoming large in groups such as bandicoots, bettongs and macropods. Young dasyurids are usually very firmly attached in the pouch, but if caught with the female when well-furred and not permanently attached to a teat, it is important that handling be minimal and that the female and her young be quietly released together. Pouch condition can be used to age females, at least in small dasyurids. Females of adult size with a clean pouch have almost certainly never raised young, so are likely to be under one year old. When they have raised one litter (or more), the fur around the pouch becomes permanently discoloured yellowish-brown, and this remains until the next breeding season. Therefore, if two adult females of a spring-breeding species are caught in May, one with a clean pouch and one with a discoloured pouch, the former will be about 8 months old and the latter will be at least a year older.

Other observations

Make note of injuries; missing toes, damaged ears/tail and scars can be used for later recognition of the same animal in long-term studies. Record anything that might be useful later. External parasites are often conspicuous on small mammals (including small orange mites around the genitalia), and changes in parasite loads may give some indication of population health.

Marking fauna

Marking fauna allows either individuals or groups of individuals to be recognised if they are recaptured or seen later. This is a complex field that is rapidly changing with the development of new technologies, and there are welfare issues with some techniques. Almost all techniques require some instruction, training and specific approval before they can be used, and therefore the following notes provide only a brief overview of the sorts of methods that are available.

BIRD-BANDING

Bird-banding (termed ringing in Britain) began in Australia between the two World Wars, and in Europe and North America slightly earlier. It is coordinated internationally and nationally, with the result that if a banded bird is recovered anywhere, it should be possible to trace the history of that bird through one of a few centralised banding databases. As part of this coordination, bands (and capture techniques) are usually only used by approved and qualified banders and only as part of approved projects. The bands come in different sizes and shapes (termed series), and are individually numbered within each series. They also come in different metals to suit the birds and the environments to which they are exposed. Thus, waterbirds often receive stainless steel bands, whereas small bushbirds may receive easier-to-fit aluminium bands. The bands are closed with specially designed pliers and once closed are very difficult to remove without injuring the bird. While the individually numbered metal bands are the mainstay of bird-banding, coloured bands and flags can be used to mark groups of birds or even individual birds in intense population studies (Figure 53).

Bird-banding is remarkable because it is so widespread, often carried out by amateurs and in many countries the bands, equipment and database management are provided and carried out at no cost to the bander. It is effectively an international citizen science project, but banders require extensive training, usually gained by joining an existing banding project as a volunteer. Information on banding projects in any region can be obtained from the ABBBS; they can even provide information on banding projects in other countries for people proposing to travel.

Figure 53. A young Hooded Plover with two coloured flags on the right leg, and a metal band on the left. The two coloured flags is a unique combination and allow this individual to be recognised from a distance (photo A. Bamford).

MARKING OTHER VERTEBRATE FAUNA

Most birds are ideally suited for banding because they have long, straight and slender legs onto which bands can readily be placed. Most other fauna groups are not so easily marked, but a variety of techniques is available as outlined below. Some of these potentially cause distress to the animal being marked, and therefore the value of the information that can be gained from marking needs to be weighed against any suffering or injury that might result; the least invasive marking techniques should always be used. Some of the techniques are also expensive and "high-tech", and thus have limited but highly valuable application.

Tissue marking

This involves permanently or temporarily marking the animal in some way by removing or colouring tissue, and includes toe-clipping, scale-clipping, ear-punching, tattooing and freeze branding. Some of these techniques have largely been superseded by transponder tags (see below), but are still useful in some circumstances.

i. **Toe-clipping** involves snipping off individual toes in combinations that give each animal (or group of animals if cohort-marking is used) a distinct number (Figure 54). It has been widely used for frogs and reptiles, and was formerly used for small mammals; Petit and Waudby (2013) note that toe-clipping of small mammals is only likely to be approved if needed for the collection of tissue samples. There are also some concerns with infection rates after toe-clipping of frogs, but at least for small reptiles it is still a quick, accurate and apparently un-traumatic marking technique. Most reptiles appear to have little nerve tissue in their toes (geckoes may be an exception). Note that reptiles have a high rate of natural toe loss that can be incorporated into a marking system and thus reduce the number of toes that need to be clipped. In general, the long toes on the hind feet are often not clipped and combinations that require the removal of more than one toe on one foot are not used. It is also possible to remove just the last joint of a toe, simply so that the nail doesn't regrow. Indeed, a modified approach for temporary marking is simply to clip the nails of the lizard using the same numbering system.

ii. **Scale-clipping.** This can be used for larger snakes (by clipping the ventral scales), small chelonids (tortoises/turtles) and very large reptiles such as crocodiles that have crest-like scales that can be clipped. As with toe-clipping, the position of clipped scales can be assigned a number (either a decimal or a digit) so that each specimen is individually numbered.

iii. **Ear-punching.** This is based on a system commonly used to mark livestock and is widely used for small mammals. There are four positions in each ear and these increase by a factor of two from one position to the next (Figure 55). Thus, animal #3 would be marked at positions 1 and 2, animal number 4 would be marked only at position 4, and animal #20 would be marked at positions 16 and 4. Usually only two positions are used in any one ear. Very small ear punches are commercially available for ear punching small mammals. When using this system, it is important to be very precise in locating marks to avoid confusion.

iv. **Tattooing and freeze-branding.** There are commercially available kits of very small metal numbers that can be used to tattoo mammals, with the tattoo usually being placed inside the ear or on the tail. Freeze-branding uses similar metal numbers that are chilled in liquid nitrogen and are very effective to permanently mark large reptiles. Both these techniques are cumbersome to use but may have application in special circumstances. A temporary variation on tattooing is to temporarily mark small mammals inside the ear with a permanent marker pen. This can persist for days and even weeks, and some variations in the position (and ear) of the mark means that several combinations are available.

Ear tags

Metal ear tags are widely used to mark livestock and very small ear tags are commercially available for marking small mammals. These have the disadvantage that even small tags can be big in the ear of a 10g mammal, and small mammals often move quickly through dense vegetation, with the result that the tag is soon torn out. In large mammals, however, a metal ear tag is distinctive and can even be read from a distance through a telescope, or can be given colour patterns that can be recognised. Similar metal tags can be used to mark marine turtles, being pushed through the trailing edge of the flipper.

Implantable Transponder Tags/Microchips/ Passive Identification Transponders (PITs)

These are widely used to mark pets but are also invaluable in wildlife research. Each PIT is about the size of a grain of rice, and can be inserted under the skin using a specially designed inserter that is like a large hypodermic needle. They are usually placed between the shoulder blades of mammals down to the size of a Mulgara (about the size of a smallish Guinea Pig), and can also be used on large reptiles (the groin is the favoured insertion point as that is where the scales are smallest). In mammals, the PIT usually stays approximately where it is inserted (between skin and muscle), but in reptiles it is often within the body cavity and thus can move around. They are referred to as passive as they are like a bar code on an item of shopping, and therefore must be read by a scanner. The scanners will read a PIT through tissue, cloth bags and even a short distance of wood or soil. Note that there are two brands of PITs (Allflex and Trovan), and these require different scanners (although some scanners claim to be able to read either PIT). There are also different models of PITs, with some of the newer versions being very small. PITs usually require the animal to be recaptured to be read, but set-ups are available where a powerful scanner can be placed beside a baiting station or a runway so that animals can be detected without being caught. Inserting PITs is simple but does require training. When using PITs in a trapping program, be careful to check an animal for an existing PIT as it is wasteful and confusing to have an animal with two numbers.

Data Transmitters

Data transmitters are now available that can be fitted to animals (usually medium-sized to large mammals) and that provide data remotely on location; they can even be designed to send information such as body temperature. Such data transmitters are valuable for detailed ecological studies.

A steady and gentle hand is required to individually mark a butterfly before release (photo M. Williams)

Radio-tracking

Radio-tracking has been used for many decades to follow the movements of individual animals, ranging in size from large mammals to large insects. The principle is unchanged: the animal is fitted with a transmitter that emits a pulse, and this is received by an antenna (usually handheld and thus mobile, but an array of fixed antennae can be used). The pulse received by the antenna is directional (ie. it is strongest from one direction), and if readings from several known locations are taken, the bearings from those locations can be triangulated to determine the position of the animal. The life and range of the transmitter varies mostly with size; transmitters half the size of a matchbox and fitted to 8-9kg wallabies had a life of over a year and a range of over 1km (Bamford and Bamford 2002). Radio-tracking transmitters can be combined with data transmitters to provide even more information. The weight of the transmitting device relative to the weight of the animal is important and recommendations range from under 5% to under 1%.

Satellite-tracking

These were initially used on marine mammals and large sharks and allowed their movements around the globe to be followed; the animal is effectively fitted with a Global Position System (GPS) unit that transmits location information to a receiver. In recent years, satellite-trackers have been fitted to animals as small as migratory birds (<1000g). In such cases, the device has been surgically implanted but the information has revealed hitherto unsuspected long-distance and continuous flights.

Geolocators

These are small units that can be attached to the legs of birds as small as 100g. They do not transmit data but simply record the movements of the bird over a period of many months, and the bird has to be recaptured to download the information. Geolocators work through having an internal clock (date and time) and recording the time of sunrise and sunset each day, which allows latitude and longitude to be calculated. They are accurate only to tens or even hundreds of kilometres, but for migratory birds that travel 10,000km in a year, this is more than adequate.

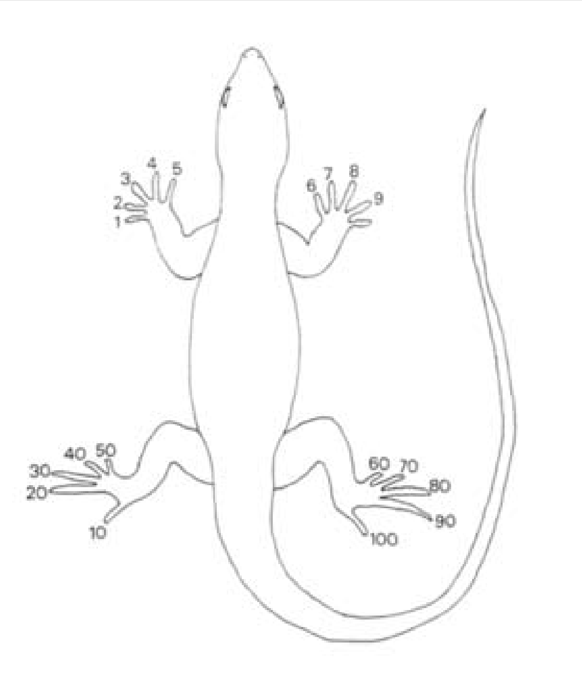

Figure 54. An example of a simple numbering system for toe-clipping (or nail-clipping) lizards (illustration M. Bamford).

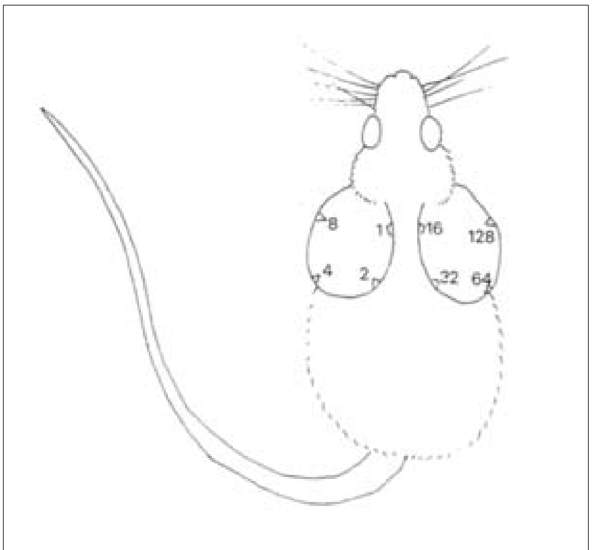

Figure 55. A binomial numbering system for ear-punching small mammals (illustration M. Bamford).

NON-DESTRUCTIVE TISSUE SAMPLING

This guide deals with the non-destructive capture of fauna for research and monitoring purposes. It does not cover projects requiring the collection of fauna or fauna series for taxonomic investigations, or for the long-term or permanent removal of animals into captivity. However, under some circumstances the information recovered from each capture can be increased using non-destructive tissue sampling.

SAMPLING OF GENETIC MATERIAL (DNA)

The vouchering of whole animal specimens from biological surveys for identification purposes is no longer standard practice (at least of protected species) and these days will normally require justification in advance through the permit application process. Whilst not vouchering whole animals may satisfy some ethical concerns, it does present some challenges in relation to the rigour and traceability of biological survey data. This is a particular problem as fauna taxonomy is in a constant state of flux, and increasingly relies on molecular methods for identifying and discriminating species.

Digital photography vouchering (digi-vouchers) is a partial solution, but for many closely-related species complexes (or future complexes), the only solution may be to collect and bank DNA samples from non-destructive tissue samples. The Western Australian Museum may be able to provide advice on the taxonomic groups where DNA sample collection may be particularly important. They may also provide advice on their capacity and willingness to house and / or analyse such samples.

For vertebrates, blood samples (100 microlitres) or vascular tissues provide the most options for DNA analysis. Tissues excised during marking such as toe-clips, scales, ear-punches or tail tips (geckoes, skinks, legless lizards), sloughed skin, fur or feather follicles can also provide material for genetic vouchering. Buccal swabs may also be adequate for some applications. Samples should preferably be stored in 100% ethanol.

For invertebrates, whole specimens are routinely collected, but to be suitable for DNA analysis they need to be preserved in 100% rather than 75% ethanol. The higher concentration of ethanol makes specimens brittle and thus unsuitable for long-term storage, so a common approach is to store the whole animal in a 75% solution, with a leg placed in 100% ethanol inside a smaller container. The small container should be placed inside the vial housing the whole specimen so the two do not become separated.

TISSUE SAMPLING FOR STABLE ISOTOPE ANALYSIS

Analysis of the stable isotope ratios (fractionations) in various consumer tissues can provide ecological information on foraging habitats and trophic- levels, and the structure of faunal communities. The most commonly-used stable isotope ratios for ecological studies are for carbon $\delta 13C$ and nitrogen $\delta 15N$. Non-destructive sampling for stable isotope analysis normally involves sampling inert, non –vascularised, keratin-based tissues including skin, toe-nails, fur and completed feathers. These materials can be stored (or banked) indefinitely in dry, sealed containers.

Feathers are particularly useful repositories of time-interval foraging and environmental information, because they are replaced in sequence over discrete seasonal or developmental periods during the moult, and are isolated from the bloodstream once completed. They can provide information on habitat use, movements, mean trophic level and community structure.

Feather selection depends on the question(s) being asked, and knowledge of the timing and sequence of the moult. The number and size of feathers being extracted should be well within the extent of feather gaps during the natural moult (particularly if any flight feathers are used). Drawn feathers are generally regrown within a few weeks of extraction even outside the normal moulting period. The rate of feather regrowth in re-captured birds may be a useful indicator of nutritional state.

Cutting out tips of feathers is not recommended as the damaged plumage will not be replaced until the next basic moult, potentially compromising survival.

TISSUE SAMPLING FOR CONTAMINANTS

Birds sequester a range of metal contaminants in their feathers. Mammal fur may also indicate levels of metal contamination. As with stable isotopes, the metals in feathers may indicate exposure from external or dietary sources in particular areas over discrete periods. Sample feathers should be drawn from the feather tracts rather than cut or trimmed. Mammal fur generally grows continuously and can be trimmed or shaved to provide samples. The sample weights needed for metals analysis (limits of detection) are considerably larger that for stable isotope analysis, and this may require the aggregation of samples from a number of individuals.

Lipid soluble contaminants such as organochlorine pesticides, PCBs or phthalates are not sequestered in keratin-based tissues such as skin, hair or feathers. Some, however, such as phthalates, may accumulate in oily secretions such as those from the preen gland in birds. Samples may be obtained for analysis by stimulating and swabbing the preen gland.

PRESERVATION OF WHOLE SPECIMENS

While vouchering of whole animal specimens is today rarely necessary or acceptable, there are occasions when dead animals in good condition are encountered, such as from trap mortality, and sometimes there may be a case for keeping such specimens rather than leaving the body to be recycled by scavengers. Invertebrates can be stored in 70% ethanol. For optimal preservation, vertebrates should first be fixed in 5% formalin and then transferred to 70% ethanol. The time spent in the formalin depends upon the size of the specimen: one day for frogs, small lizards and small mammals, and two or three days for larger animals. The body cavity should be carefully slit open for the preservation of internal organs, and formalin should also be injected through the diaphragm of mammals using a hypodermic needle. Note that formalin and ethanol are both restricted and potentially hazardous substances and conditions for their transport, storage and use may apply.

Specimens should be clearly labelled; pencil written on heavy paper is recommended (ink may dissolve in preservative). The label should include information about the specimen such as date, location, description of the environment and any measurements taken, or the label can have a reference number and these data can be recorded separately and cross-referenced.

Keeping Records

It should go without saying that records should be kept so that they are not lost, are available for the purpose intended and can be used to fulfil permitting obligations. It is important to be systematic in record-keeping in the field and not to rely on memory. A robust notebook (ie. one in which the pages will not fall out) is traditionally considered essential; digital devices are becoming useful, especially where data can be entered direct into a well-structured spreadsheet. A back-up notebook is probably still a very good idea, especially for making non-standard notes. Written records should be in pencil as pen can run and may not work under all weather conditions (many detailed and valuable notes have been scratched into sodden paper with a 2B pencil). Transferring data from notebooks/digital recorders should take place at the end of every field day and digital data should be backed-up. Keep old notebooks (preferably labelled with dates and projects).

A well-designed spreadsheet or database, especially for trapping and/or census records, will save a lot of time in keeping track of data and in their later analysis. It is better to record more information than less, on the principle that if details are not recorded, they are lost, but if they are recorded and are not subsequently used, there is no harm done.

Preserved specimens of ants and millipedes; note label written in pencil and inside the container with the specimens (photo T. Gamblin).

COMPETENCIES CHECKLIST

Background

Undertaking any sampling of fauna that disturbs or potentially impacts upon the animals being studied requires considerable expertise, and thus necessary permits are generally only issued to people with suitable experience. Most field researchers gain their experience over a lifetime but in an increasingly regulated environment, it is increasingly difficult to "get started". This is affecting students, university courses and the growing numbers of community groups engaged in on-the-ground conservation. The contents of this manual are intended to provide a guide of how to get started. For authorities charged with managing the issuing of permits for field studies into fauna, a record of experience is required. The following three forms provide a structure for keeping such records.

Competency Form A. Competency overview for trap setting and management

Form A summarises the setting and management requirements of the main types of traps used in fauna investigations. Mist-nets and cannon nets are not included as use of these nets is subject to separate assessment by the Australian Bird and Bat Banding Scheme.

Competency Form B. Fauna handling record sheet

Form B provides a means of recording every fauna handling event so that students or members of community groups who are assisting with projects can develop a record of their fauna handling experience. The form records the species of animal, how it was caught (usually the type of trap) and the steps in the handling of the animal, from removal from trap to general handling, identification, measuring and release. The form allows for the person supervising the activity to note if the person was competent or if intervention was required.

Competency Form C. Competency overview for animal handling

Form C effectively provides a summary from Form B. It recognises that fauna can be placed into groups of species for which trapping and handling requirements are similar (e.g. small lizards, small mammals). It provides a checklist and quick reference to record the student's experience with each species group in terms of types of traps and handling. For example, someone may be very competent with small lizards in their removal from pitfall but not funnel traps. They may be competent in general handling and measuring this group of species, but have no similar mammal experience. Birds and bats are not included in this form as they are subject to separate assessment by the Australian Bird and Bat Banding Scheme.

Searching a breakaway for evidence of Northern Quoll (photo M. Bamford).

Competency form A. Competency overview for trap setting and management

Participant		Supervisor	

Trap Type	**Position**	**Assembly**	**Door mechanism**	**Bait position**	**Location** (flagging tape-trap#, line#, coordinates, shelter)	**Checking protocol**	**Supervisor confirmation** (initial and date)
Cage	Shade, hessian cover, level	Back shut, mechanism works	Free from obstruction	Behind, not under plate	Check	Confirm	
Elliott	Shade, level, access	Mechanism works	Check	Behind plate	Check	Confirm	
Pitfall	Mid-day shade (vegetation and/or lid). Lip flush with (or slightly lower than) ground	Adequate drainage holes, shelter inside pit	n/a	n/a	Check	Confirm	
Drift-fence	Meets pitfall and/or funnel correctly	Check for gaps, vegetation that might obstruct movement	n/a	n/a	Check	n/a	
Funnel trap	In shade, with cover, fence inserted, soil ramped into entrance	Check for damage, zip closed	n/a	n/a	Check	Confirm	
Motion-sensitive camera	Directed towards where animals may move and so as to minimize false triggers from moving vegetation, sun/shade, etc.	Card inserted and cleared, battery life checked, camera set correctly (stable, light masked if necessary). Scale in field of view	n/a	If baited, placed in field of view	Check	Confirm	

Competency form B. Fauna handling record sheet

Trainee		Supervisor		DPaW permit no.		Location	

Date	Method	Species (and species group code)	ASSESSMENT							
			Preliminary identification		Removal from trap and handling		Measurements		Release		Supervisor initials

Each assessment event should be marked **S**-*satisfactory* or **I**-*requiring supervisor intervention*.

METHOD: Refers to trap type or other method of capture. SPECIES: Indicate species group in parenthesis (eg. small lizard, small rodent, large reptile, snake).
PRELIMINARY IDENTIFICATION: Recognition of species group and species if possible for appropriate handling. HANDLING: Specimen handled efficiently without either harm to animal or handler.
MEASUREMENTS: Standard measurements taken successfully. RELEASE: Specimen released in good condition and so as to minimise further risk to animal.

Example of a completed fauna handling record sheet (Competency form B)

Trainee	A. Train	Supervisor	A. Super	DPaW permit no.	001	Location	Somewhere

Date	Method	Species (and species group code)	ASSESSMENT								
			Preliminary identification		Removal from trap and handling		Measurements		Release		Supervisor initials
8/02/13	Funnel	*Pogona minor (Ll)*	Large lizard; identified. Further to a dragon, and keyed out to correct species.	S	Retrieved and processed	S	Weight, SVL, and total length.	S	Released in suitable habitat and hidden as body temperature low.	I	
8/02/13	Pitfall	*Ramphotyphlops australis (Ss)*	Small snake; identified. Further to non-venomous snake; and keyed out to correct species.	S	Retrieved and processed	S	Weight, SVL, and total length.	S	Released in suitable habitat and hidden as body temperature low.	I	
8/02/13	Active search by day	*Crinia pseudinsignifera (F)*	Small frog; identified to genus only; a difficult genus when not calling	I	Retrieved but processed by other	S	Weight, SVL and tibia length.	S	Released in suitable moist habitat.	I	
9/02/13	Pitfall	*Tarsipes rostratus (Sm)*	Small marsupial mammal; identified further to species level.	S	Retrieved and processed. Sexed by external genitalia	S	Weight, head length, sex, testes width.	S	Released in suitable habitat after honey feed.	S	

Each assessment event should be marked **S**-*satisfactory* or **I**-*requiring supervisor intervention*.

METHOD: Refers to trap type or other method of capture. SPECIES: Indicate species group in parenthesis (eg. small lizard, small rodent, large reptile, snake). PRELIMINARY IDENTIFICATION: Recognition of species group and species if possible for appropriate handling. HANDLING: Specimen handled efficiently without either harm to animal or handler. MEASUREMENTS: Standard measurements taken successfully. RELEASE: Specimen released in good condition and so as to minimise further risk to animal.

Competency form C. Competency overview for animal handling

Participant		Supervisor	

Animal type	Code	Pitfall	Funnel	Cage	Elliott	Preliminary identification	Handling/ retention	Measurements	Marking	Release
Frog	F									
Small lizard	Sl									
Large skink/dragon, small varanid	Ll									
Large varanid	LV									
Small snake (non-dangerous)	Ss									
Large non-venomous snake	LsND									
Dangerously venomous snake	LsD									
Small rodent	Sr									
Large rodent	Lr									
Small marsupial	Sm									
Large dasyurid	Ld									
Possum/bandicoot/potorooids	PBP									
Group specific explanations										
Group specific descriptions/ illustrations										
Note on marking techniques										
Notes on safe release										

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Gould's Sand Goanna *Varanus gouldii* up a tree (photo A. Bamford)

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Sunrise while checking traps (photo M. Bamford).

GLOSSARY

Age cohort	An identified group or class of ages in a population; may be all animals from one breeding season and thus about the same age, or all animals that are sexually mature.
Cloaca	In amphibians, reptiles (and monotremes), a common opening for all excretion and reproduction; also referred to as the vent.
Crown	A standard measurement from the tip of the snout to the back of the skull, usually in mammals. Similar measurement in birds often referred to as head-bill.
Demographic structure	The characteristics of a population such as proportion of age classes and ratio of males to females.
Dentition	The number, type and orientation of teeth.
Digit	Finger or toe.
Eutherian	A major Sub-Class of mammals often referred to as placental mammals as the young are nurtured internally by the female via a placenta for a long period of time compared with marsupials (that bear very small young which are subsequently nurtured in a pouch).
Feral	A species that has been introduced to a given area.
Fossorial	Adapted to life below ground. Includes many lizards and snakes that rarely come to the surface, and mammals such as the Marsupial Mole.
Head-body length	A standard (but not very accurate) mammal measurement from the nose tip to tail base.
Incisors	The front teeth in most mammals, usually present in the lower and upper jaw. In rodents grow throughout life.
Nectivorous	Describes a diet that includes a high percentage of nectar, though other food sources such as fruit, insects and sap may also be consumed in small quantities. The Honey Possum is described as the only strictly nectivorous non-flying mammal, but relies heavily on pollen for nutrition.
Pes	A measurement of the length of the foot from the back of the heel to the end of the toes, either including (long pes) or excluding (short pes) nails. It is an ancient Latin term originally referring to a length of approximately a foot.
Scat	Faeces in a pellet or ball.
Site-tenacity	The extent to which individuals of a species are sedentary.
Snout-vent length	A standard measurement in reptiles and amphibians from the tip of the snout to the vent (cloaca).
Tragus	A fleshy structure of the ear, particularly useful in identifying bats.
Trap-effort	The total number of trap-nights in a survey period; the number of traps multiplied by the number of trap-nights.
Trap-night	Often used as the standard unit for trapping effort, as traps are usually set one day, operated for several days and nights, and then taken up/closed on the final day. Thus, they are open for several complete nights but for part of two separate daylight periods, so counting the nights is more precise and unambiguous.
Trapping period	The number of trap-nights over which traps are operated.
Torpor	A voluntary condition in some small mammals (rarely in some birds) in which the metabolic rate (thus body temperature) is reduced to conserve energy reserves. Often seen in small marsupials when trapped.

ABOUT THE CONSERVATION COUNCIL OF WA

The Conservation Council of WA (CCWA) is the State's foremost non-profit, non-government conservation organisation. We are an umbrella group for over 90 affiliate conservation groups and have been an outspoken advocate for environmental protection and a sustainable WA for over 40 years.

With your support, we can be a powerful catalyst for transforming Western Australia's economy and protecting our natural environment.

Individuals – support us

If you would like to contribute to building a sustainable WA, you can:

- Become a voice for the Environment by making a regular donation
- Make a single donation
- Become a volunteer
- Leave us a gift in your will

Groups – join us

CCWA works in partnership with other conservation groups, large and small. Become a CCWA member group and:

- Increase your group's impact
- Improve your networks and contact base
- Gain access to support, expertise and representation

For more information on how to support CCWA see our website

www.conservationwa.asn.au

Conservation Council of Western Australia
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Fire-tail Skink *Morethia ruficauda* (photo R. Browne-Cooper)

Sampling animals, whether by structured observation or live-capture methods, is a necessary element in fauna research and monitoring programs. These programs provide vital information for wildlife conservation management and key insights into the condition of the ecosystems upon which we all ultimately depend. It is however important that the fauna sampling practices adopted are technically sound and ethically defensible.

The Conservation Council's Citizen Science Program uses the least intrusive methods available to collect data on protected fauna. However, no sampling method is completely free of disruption or risk to our animal subjects. The Fauna Sampling Manual has been developed as a guide for participants in the various fauna related projects being undertaken in the Citizen Science Program and as a resource for a fauna-training product, available to students and novice practitioners outside the community conservation sector. The manual distils several decades of practical experience gained by its principal authors and a range of other somewhat weathered Western Australian field biologists.

Having the legal requirements necessary to study our native animals is a privilege that is increasingly restricted to those who can demonstrate that they have the knowledge and experience to work responsibly. Tertiary teaching organizations have insufficient resources and supervisory capacity these days to provide their graduates with sufficient field sampling experience. Hopefully this CCWA / Bamford Consulting contribution to fauna-training will assist in raising the bar and helping the next generation of wildlife conservation practitioners jump over it.

Conservation Council of Western Australia
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Look forward

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