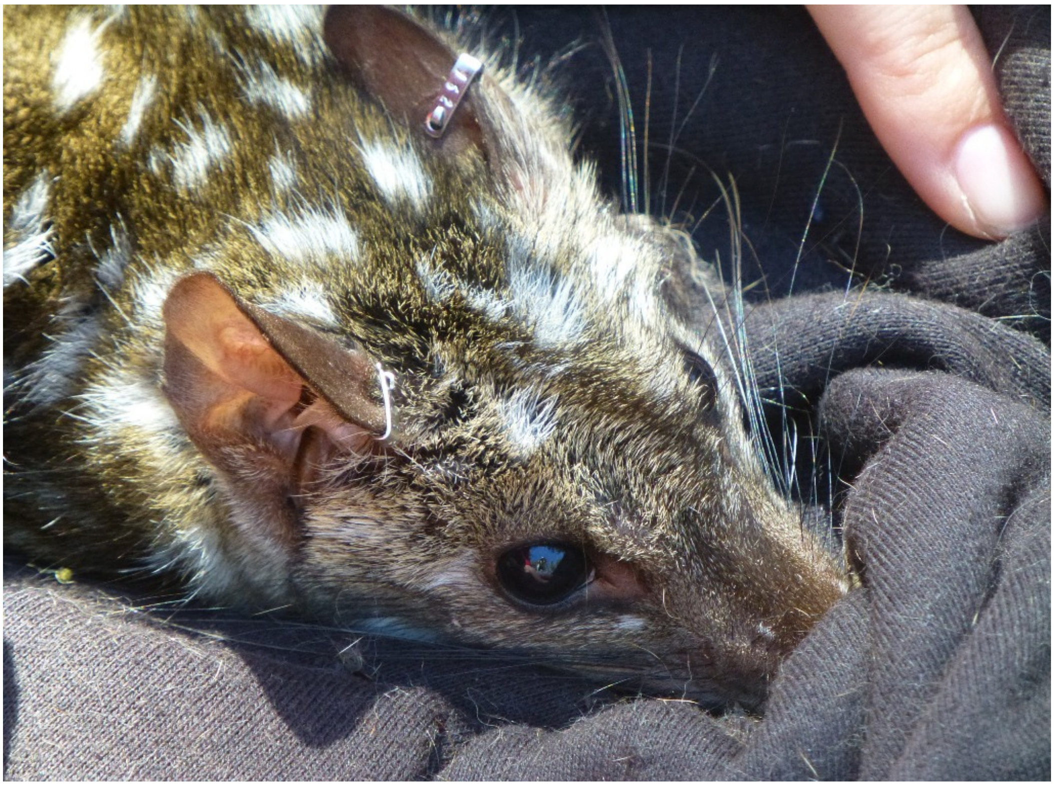
Fauna Management Course 2015





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9-13 November 2015

Department of
Parks and Wildlife

Contents

1. Fauna Conservation in DPaW and Fauna Project Planning

1.1. Legislation, licenses and approvals

Legislative requirements

The two pieces of legislation defining the Department of Parks and Wildlife's (DPaW) role in wildlife conservation are the:

- *Conservation and Land Management Act 1984* (CALM Act) [& Conservation and Land Management Regulations 2002] , and
- *Wildlife Conservation Act 1950 (*& Wildlife Conservation Regulations 1970).

There are two other pieces of legislation that influence DPaW's activities in relation to wildlife conservation:

- *Animal Welfare Act 2002*, and
- (Commonwealth) *Environment Protection and Biodiversity Conservation Act 1999* (EPBC Act).

Each of these Acts are summarized below with specific reference to how they influence and set the context for fauna management in the department.

Western Australia's Acts and Regulations can be viewed and downloaded from the State Law Publisher website: http://www.slp.wa.gov.au/Index.html

Australian Government legislation can be viewed and downloaded from the ComLaw website: http://www.comlaw.gov.au

Not all fauna require active management, nor do we have the resources to manage all fauna. DPaW has a legal responsibility to manage several groups of fauna in Western Australia and these groups can be summarised as follows:

- Gazetted fauna (as listed in Government Gazette) -
  - Schedule 1: "Fauna that is rare or is likely to become extinct",
  - Schedule 3: "Birds protected under an international agreement", and
  - Schedule 4: "Other specially protected fauna";
- Commercially harvested species covered by a management program, eg kangaroos and crocodiles;
- Dangerous fauna, as covered by Wildlife Conservation Regulation 4, eg venomous snakes and crocodiles where they come into direct contact with humans; and
- Fauna causing damage to property, as covered by Wildlife Conservation Regulation 5, eg kangaroos and wallabies in crops, possums in roofs, parrots in fruit orchards etc.

- Fauna declared in open season and restricted season notices in the Government Gazette, eg pest birds, some ducks.

The Wildlife Conservation Act 1950* & *Wildlife Conservation Regulations 1970

Licences to take or keep fauna

A licence is required for a person or persons to take or keep native fauna for scientific, educational or public purposes irrespective of who they work for. The issuing of licences is administered under the *Wildlife Conservation Regulations 1970* and the following licences are commonly applicable to fauna management activities.

- Licence to take fauna for scientific purposes (Reg 17): Activities that require this licence include trapping or catching animals for scientific research or management purposes.
- Licence to keep fauna for educational or public purposes (Reg 16): Activities that require this licence include the display of live animals for educational programs and public exhibitions.
- Licence to take fauna for educational or public purposes (Reg 15): Preference is given to using animals that are already in captivity for purposes of rehabilitation or captive breeding and approval to take from the wild is given sparingly.

Any person 'taking' fauna (trapping, catching, handling, etc) or keeping fauna as part of fauna management activities must either:

1. have the appropriate licence, or
2. be under the direct supervision (i.e. in the physical presence) of a licenced person.

Before commencing any activity involving the taking of fauna (e.g. surveys or monitoring) or keeping of fauna (e.g. educational displays) check that the people involved are covered by the appropriate licence.

Contact the Wildlife Licensing Branch of DPaW for advice and issuing of licences, further information about licences can be sourced from: http://www.dpaw.wa.gov.au/plants-and-animals/licences-and-permits/134-fauna-forms?showall=&start=2

Animal Welfare Act 2002 and Animal Welfare Regulations 2003 (General and Scientific)

Licence to use animals for scientific purposes

In December 2002 the Animal Welfare Act was gazetted. This Act replaced the *Prevention of Cruelty to Animals 1920* Act. The *Animal Welfare Regulations 2003* (General and Scientific) followed shortly after.

The Animal Welfare Act applies to all vertebrate animals (excluding fish) in Western Australia and requires all establishments that use animals for scientific purposes (including DPaW) to be licenced.

This is a licence to use animals for scientific purposes DPaW has this but the licence conditions require the licensee to adhere to the *Australian code of practice for the care and use of animals for scientific purposes* and this includes having access to a properly constituted Animal Ethics Committee. All projects or activities involving the use of animals must be assessed via written applications and approved by the Animal Ethics Committee before commencement.

Before commencing any activity involving the use of fauna for scientific purposes the project must have AEC approval. This will be dealt with in more detail in section 3.

1.2. The conservation status of Western Australia's vertebrate fauna

Refer to policy number 35

1.2.1. Conservation Status

In the last 200 years Australia has suffered the worst record of any continent for the extinction of species. Twenty seven mammal species have become extinct and eleven of those species occurred in Western Australia. Many other species of fauna have seriously declined and are currently threatened with the possibility of extinction.

It is important for the conservation of these species to determine the extent to which each species is threatened with extinction so that conservation efforts can be prioritised and directed to species in greatest need. This requires knowledge about the biology and distribution of species and a robust system of assessing the threat of extinction and categorising species accordingly.

IUCN Red List Categories

This approach has been used by the World Conservation Union (IUCN) to develop the IUCN Red List Categories. Each category has a set of quantitative criteria with which to evaluate species for listing. These categories are intended to be an easily and widely understood system for classifying species at high risk of global extinction. The general aim of the system is to provide an explicit, objective framework for the classification of the broadest range of species according to their extinction risk.

The IUCN Red List Categories and Criteria have several specific aims:

- to provide a system that can be applied consistently by different people;
- to improve objectivity by providing users with clear guidance on how to evaluate different factors which affect the risk of extinction;
- to provide a system which will facilitate comparisons across widely different taxa;
- to give people using threatened species lists a better understanding of how individual species were classified.

These categories and criteria have become widely recognised internationally and were last revised in 2008. They were adopted by the Commonwealth Government under the *Endangered Species Protection Act 1992* and are used to rank species under the EPBC Act. They have also been adopted by WA's Threatened Species Scientific Committee for the purposes of reviewing the status of WA species for listing under the Wildlife Conservation Act.

The IUCN categories are defined as follows: http://www.iucnredlist.org/technical-documents/categories-and-criteria/2001-categories-criteria

Extinct (EX)

A taxon is extinct when there is no reasonable doubt that the last individual has died.

Extinct in the Wild (EW)

A taxon is extinct in the wild when it is known only to survive in cultivation, in captivity or as a naturalised population (or populations) well outside the past range. A taxon is presumed extinct in the wild when exhaustive surveys in known and/or expected habitat, at appropriate times (diurnal, seasonal, annual), throughout its historic range have failed to record an individual. Surveys should be over a time frame appropriate to the taxon's life cycle and life form.

Critically endangered (CR)

A taxon is Critically Endangered when it is facing an extremely high risk of extinction in the wild in the immediate future.

Endangered (EN)

A taxon is Endangered when it is not Critically Endangered but is facing a very high risk of extinction in the wild in the near future.

Vulnerable (VU)

A taxon is Vulnerable when it is not Critically Endangered or Endangered but is facing a high risk of extinction in the wild in the medium-term future.

Near threatened (NT)

A taxon is Near Threatened when it has been evaluated, does not satisfy the criteria for any of the categories Critically Endangered, Endangered or Vulnerable, but is close to qualifying for or is likely to qualify for a threatened category in the near future.

Least concern (LC)

A taxon is least concern when it has been evaluated, does not satisfy the criteria for any of the categories Critically Endangered, Endangered, Vulnerable or Near Threatened. Widespread and abundant taxa are included in this category.

Formerly used but no longer retained by IUCN, this next category is still used in Australia:

Conservation dependent

A taxon is Conservation Dependent when it is the focus of a continuing taxon-specific or habitat-specific conservation program, the cessation of which would result in the taxon qualifying for the threatened categories within a period of five years.

Listings under the Wildlife Conservation Act 1950

The Wildlife Conservation Act provides for species to be declared as 'likely to become extinct or rare, or otherwise in need of special protection', by Ministerial Notice in the Government Gazette.

The Gazette Notice groups species into Schedules according to their status as follows.

Schedule 1 - Fauna that is rare or is likely to become extinct

These species are usually termed 'threatened' and can be defined as;

native fauna which are

- well defined in taxonomic literature, or if undescribed, represented by a voucher specimen in a record collection,
- in imminent danger or threatened with extinction,
- dependent on/restricted to vulnerable habitats, and
- very uncommon, even if widespread.

Species in this schedule have been ranked as Extinct in the Wild, Critically Endangered, Endangered, or Vulnerable under the criteria for the IUCN Red List Categories described above.

Schedule 2 - Fauna presumed to be extinct

Species in this schedule have been ranked as Extinct under the criteria for IUCN Red List Categories.

Schedule 3 - Birds protected under an international agreement

Species protected under this schedule include birds that are subject to an agreement between governments of Australia and Japan, China and The Republic of Korea which relates to the protection of migratory birds and birds in danger of extinction.

Schedule 4 - Other specially protected fauna

Fauna under this category are also known as Specially Protected Fauna. Specially Protected Fauna are likely to be taken because of high commercial value or are uncommon, but not currently threatened, but are either of commercial or intrinsic value or are perceived to be damaging to a commercial or hobby enterprise and taking may lead to the species becoming threatened.

DPaW Priority Fauna List

DPaW manages fauna according to the Wildlife Conservation Act schedules. In addition DPaW maintains a 'Priority Fauna List' that contains taxa that do not currently meet the criteria for the threatened categories but are of concern for various reasons. Taxa in this list would fall into the IUCN Red List Categories of Near Threatened (including the formerly used category Conservation Dependent) or Data Deficient. The list is not supported by legislation. Taxa are allocated to one of five priority categories as follows:

<u>Priority One Taxa with few, poorly known populations on threatened lands.</u>

Taxa which are known from few specimens or sight records from one or two localities on lands not managed for conservation, eg. agricultural or pastoral lands, urban areas, active mineral leases. The taxon needs urgent survey and evaluation of status before consideration can be given to declaration as threatened fauna.

<u>Priority Two Taxa with few, poorly known populations on conservation lands.</u>

Taxa which are known from few specimens or sight records from one or two localities on lands not under immediate threat of habitat destruction or degradation, eg. national parks, conservation parks, nature reserves, State forest, vacant Crown land, water reserves, etc. The taxon needs urgent survey and evaluation of status before consideration can be given to declaration as threatened fauna.

<u>Priority Three Taxa with several, poorly known populations, some on conservation lands</u>

Taxa which are known from few specimen or sight records from several localities, some of which are on lands not under immediate threat of habitat destruction or degradation. The taxon needs urgent survey and evaluation of conservation status before consideration can be given to declaration as threatened fauna.

<u>Priority Four Taxa in need of monitoring</u>

Taxa which are considered to have been adequately surveyed, or for which sufficient knowledge is available, and which are considered not currently threatened or in need of special protection, but could be if current circumstances change. These taxa are usually represented on conservation lands.

<u>Priority Five Taxa in need of monitoring (conservation dependent)</u>

Taxa which are not considered threatened but are subject to a specific conservation program, the cessation of which would result in the species becoming threatened within five years.

Listings under the (Commonwealth) *Environment Protection and Biodiversity Conservation Act 1999*

The EPBC Act provides for the listing of species as threatened. The following categories are used and are based on the IUCN Red List Categories:

- Extinct
- Extinct in the Wild
- Critically Endangered
- Endangered
- Vulnerable
- Conservation Dependant

Only those species in the categories marked * are of national environmental significance under the EPBC Act.

1.2.2. Current threatened and priority fauna rankings

Current Threatened and Priority Fauna Rankings are available in PDF format on the DPaW website, this should be checked periodically for updates. http://www.dpaw.wa.gov.au/plants-and-animals/threatened-species-and-communities/threatened-animals

1.2.3. Forming and reviewing of conservation status

DPaW Policy 35, *Conserving Threatened Species and Ecological Communities*, establishes the Threatened Species Scientific Committee. This committee oversees and reviews the threatened and specially protected fauna species for listing under the Wildlife Conservation Act. This occurs at least every three years. If species meet certain criteria they can be added or removed from the lists.

Threatened Fauna

A species may be recommended for declaration as threatened fauna (Schedule 1) by the Threatened Species Scientific Committee if it satisfies the following criteria:

i The taxon is part of the indigenous fauna of Australia or its external territories, and is well defined in the taxonomic literature, or represented by a suitable voucher specimen.

ii It has been established that the taxon in the wild is either;

- Presumed to be extinct
- In imminent danger of or threatened with extinction
- Dependent on or restricted to habitats that are vulnerable and/or subject to factors that may cause its decline
- Very uncommon even if widespread

The Threatened Species Scientific Committee may recommend that the taxon be removed from the threatened fauna schedule where recent zoological survey has shown that the taxon no longer meets the above criteria or the taxon is no longer threatened.

Priority Fauna

The Committee also prepares a 'Priority Fauna List' which is also reviewed at least every three years. This includes animal taxa:

- that have recently been removed from the list of threatened fauna
- that have a restricted distribution, are uncommon or are declining in range and/or abundance
- for which there is insufficient information for the Committee to make an assessment

Specially Protected Fauna

The Schedule of Other specially protected fauna (Schedule 4) is dealt with in the same way as Schedule 1 for threatened fauna. The criteria used to add taxon to the list is the same as for threatened fauna except criteria (ii) which is;

It has been established that taxon in the wild is either;

a) Likely to be taken because of high commercial value and the standard penalty for taking is insufficient deterrent; or

b) Uncommon, but not threatened at present, but is either of commercial or intrinsic value or is perceived to be to be damaging a commercial or hobby enterprise.

The Threatened Species Scientific Committee may recommend that a taxon be removed from Schedule 4 where;

i Recent zoological survey has shown that the taxon no longer meets the above criteria
ii The commercial or other incentive to take has disappeared or has been removed by other means.

1.2.4. Nominating a species for listing or delisting

Anyone with appropriate knowledge can submit a nomination to the Threatened Species Scientific Committee to change the conservation status of a species, with supporting data and information.

It is recommended that nominees carefully read and follow the advice provided in the "Guidelines for Using the IUCN Red List Categories and Criteria" when preparing a nomination. These guidelines are available online at: http://www.iucnredlist.org/documents/RedListGuidelines.pdf

The Threatened Species Scientific Committee meets once a year, usually in February or March to consider nominations for listing and de-listing.

Nomination forms are available by contacting DPaW Species and Communities Branch phone: (08) 9219 9511) or from the DPaW website. http://www.dpaw.wa.gov.au/plants-and-animals/threatened-species-and-communities/118-call-for-public-nominations-for-listing-and-delisting-of-threatened-plants-and-animals

1.2.5. Recovery plans

Recovery Plans are detailed, costed plans describing how a particular species, or group of species, should be managed in order to recovery the species from its current threatened situation.

Recovery Plans have the following advantages:

1. They clearly state the known threats and/or issues associated with the recovery of the species,

2. They provide written guidance on recommended recovery actions, priorities and costs, and

4. They support the engagement of Recovery Teams, which ensure key players work together to a common aim.

DPaW is currently preparing Interim Recovery Plans for all taxa ranked as Critically Endangered under Policy 50. These are written for three year periods and propose actions aimed at preventing extinction.

The Recovery Process

The conservation of threatened species must be approached in a logical way to ensure that resources are allocated to the most threatened species and that money and staff time are put to the best use.

A sequential program is:

1. Review the conservation status of taxa and list taxa as threatened as appropriate,

2. Prepare priority lists of threatened taxa,

3. Produce costed Recovery Plans,

4. Conduct the necessary research.

And for each Recovery Plan:

1. Obtain funding,
2. Implement (with advice from a Recovery Team), and
3. Monitor and review.

How far are we, DPaW, along this route?

Conservation status

We are well advanced, with good processes in place to review the lists of threatened vascular plants and vertebrates. At present we have insufficient capability to deal adequately with listings of invertebrates and non-vascular plants, although some invertebrates have been listed and conservation actions have commenced for some species.

Ranking

All threatened taxa are now allocated a ranking, based on the IUCN Red List categories and criteria under Policy Statement No. 50.

Recovery Plans

DPaW is producing full Recovery Plans or Interim Recovery Plans for all animals and plants ranked as Critically Endangered (see the DPaW website for a full list of recovery and interim recovery plans which is located at: http://www.dpaw.wa.gov.au/plants-and-animals/threatened-species-and-communities/197-approved-recovery-plans

Refer to policy number 44:

Research

DPaW has been in the forefront of scientific research into threatened species of animals. Research has informed recovery actions and includes input into fauna conservation initiatives such as *Western Shield*.

Funding

Funding for threatened fauna research and management comes from internal DPaW cost centre allocations, the Western Shield central funding, and external sources including Caring for our Country, NRM grants and Biodiversity Fund. Competition for funding for threatened fauna is high and funding is limited. DPaW recognises the need to partner with a range of stakeholders to improve funding opportunities and spend limited resources efficiently and effectively.

Implement

Recovery Teams provide advice and assist in coordinating actions prescribed in recovery plans. They include representatives from organisations with direct interest in the recovery of the species, including those involved in funding and implementing recovery actions. Recovery teams should help to guide and direct implementation using the Recovery Plan as a guide. There are active Recovery Teams for many of our threatened fauna.

Monitor and Review

DPaW, in consultation with the Recovery Team, evaluate the performance of the recovery plans, and in particular the performance against the recovery objectives and the criteria for success and failure so as to review and change the recovery actions in response to new information. Progress is reported annually in DPaW annual reports. A Recovery Plan is reviewed within five years of its adoption and again ten years after adoption if not replaced prior to that time.

1.3. Fauna project planning

1.3.1. Developing a plan for fauna projects

Science Division has developed a Science Project Plan (SPP) and guideline (Science Division Guideline No 7) to with assist planning research projects. The template is available on the Science Division intranet site. The template can be used and/or modified for other management programs, including district fauna projects, to assist with good project planning. SPP forms and guidelines can be found on the intranet at:
http://intranet/science/Documents/Forms/Staff%20Guidelines.aspx

There is also a document on the DPaW website "Designing a monitoring project for significant native fauna species" which provides guidance on developing monitoring programs.

Science and Conservation Division staff and the Western Shield Zoologist can assist with project planning.

Summary of steps to develop a fauna project or research program are:

- Identify the objectives of the project and the expected outcomes – these should be relevant to DPaW's goals and objectives as outlined in legislation, DPaW's Corporate Plan and various Division strategic plans.
- Ensure that the objectives are measurable.
- Identify the scope of the project (think about the expected outcomes and the anticipated users of the knowledge to focus the scope)
- Identify the strategies by which the objectives are to be achieved – ensure that strategies and tasks are feasible.
- Identify the tasks required to implement these strategies and timeframes
- Identify the appropriate personnel to undertake these tasks

    – Do they have the skills required?
    – Do they have the appropriate licence(s)?
    – Do they require training?

- Identify plant and equipment requirements
- Allocate a budget for each of these tasks
- Identify monitoring requirements and method of data collection
- Identify reporting - information must feedback to interested users of the knowledge (generally via written reports) so that all monitoring results and recommendations are documented and we can all learn from the project.

- Undertake evaluation - review the achieved outcomes against those identified at the beginning of the project; review the outputs that have been produced.

Prepare relevant documentation for approval

Some of the tasks identified may require specific documentation for approval before they can be undertaken. The types of documentation required may include:

- Licence to take or keep fauna for scientific, educational or public purposes
- Animal Ethics Committee approval
- Translocation Proposals
- Entry permits for Disease (dieback) Risk Areas

1.3.2. Translocation of fauna

Translocations, particularly reintroductions, are an extremely useful wildlife management tool. Since 1971, DPaW have undertaken over 230 translocations. WA is fortunate in that effective fox control programs have been implemented over large areas, and that we have many introduced predator-free offshore islands that provide a secure location and source populations.

Conservation Translocation is the intentional movement and release of a living organism where the primary objective is conservation benefit this will usually comprise of improving the conservation status of the focal species locally or globally, and/or restoring natural ecosystem functions or processes. Three main types of conservation translocation are recognised:

- **Introduction** is the intentional movement and release of an organism outside its indigenous range.
- **Re-introduction** is the intentional movement and release of an organism to inside its indigenous range from which it has disappeared. The indigenous range of a species is the known or inferred distribution generated from historic (written or verbal) records, or physical evidence of the species' occurrence. Where direct evidence is inadequate to confirm previous occupancy, the existence of suitable habitat within ecologically appropriate proximity to proven range may be taken as adequate evidence of previous occupation.
- Supplementation is the addition of individuals to an existing population of conspecifics. A supplementation aims to enhance population viability, for instance by increasing population size, increasing genetic diversity, or increasing the representation of specific demographic groups or stages.

Inappropriate or poorly planned translocations can be detrimental to wildlife conservation objectives. In WA IUCN guidelines have been adopted to ensure translocations are properly implemented.

Why do we translocate?

The primary reasons to translocate fauna are:

- Enhancement of biological diversity - to improve single species conservation status, or reconstruction of flora and fauna.

- Wildlife salvage or rehabilitation: release of animals that have been rehabilitated or relocation of animals that are under threat, e.g. due to clearing for development
- Education – placing animals into education programs, e.g., wildlife parks.

Only the first reason constitutes a 'conservation translocation' (see definition above).

When should we translocate, and when shouldn't we?

For conservation agencies, translocations should only occur to improve the conservation status of a species or community, or to lessen the impact of habitat destruction on in situ populations. They usually involve threatened species, but this is not always so. They may involve more common species to reconstruct the flora and fauna of an area, or to preserve genetic variability.

Introductions should only be considered if there are clear benefits to the conservation of the species and reintroduction options are not available. It also needs to be clear that there will be no detrimental impact of the introduced organism on the existing biota. Examples of introductions with conservation benefit are the marooning of the Shark Bay Mouse *Pseudomys fieldi* on Doole Island in Exmouth Gulf, the Greater Stick-nest Rat *Leporillus conditor* on Salutation Island, Shark Bay, and the transfer of Mala *Lagorchestes hirsutus hirsutus* to Trimouille Island.

Re-introductions are the usual type of translocation undertaken by DPaW for conservation purposes. They should only be undertaken using the same genetic stock as originally present, if this is possible. The eradication or control of the original cause of extinction (e.g. introduced predators) is essential before any translocation occurs, and the ecological requirements of the species must be known and met at the translocation site.

Supplementation is usually undertaken to improve the genetic composition of the existing population or to boost numbers

The IUCN has released Guidelines for reintroductions and other conservation translocations (2013), available on their website. DPaW uses these guidelines in the review and endorsement process for conservation translocations.

Translocation Proposals

Corporate Guideline 36 Recovery of Threatened Species Through Translocation and Captive Breeding or Propagation guides the translocation process. The process by which translocations are assessed in WA is by means of completion of a Translocation Proposal. You must have a Translocation Proposal approved by the Director Science and Conservation, and have Animal Ethics Committee approval, before undertaking a translocation. Translocation Proposal documents are often referred externally before being considered for approval. Contact the Principal Zoologist, Species and Communities Branch for the Translocation Proposal template and other supporting documentation.

In general, translocation proposals cover:

- Name and affiliation of the proponent
- Background on the species: description, taxonomy, former range, current distribution, conservation status and biology.
- Threats and causes of decline.
- Translocation history
- The translocation:
    - Justification for the translocation
    - Source population and environment
    - Details of the host environment including causes of local extinction and demonstration that these have been mitigated
    - If an Introduction, the impact on the existing biota needs to be assessed.
    - If an Introduction to an island, it must demonstrate that it will have no effect on possible other translocations to that island.
    - If a translocation from an island to mainland, it must demonstrate that the mainland taxon no longer exists.
    - Founder number and principle of conservation genetics.
    - Logistics, capture, handling, transport and release protocols.
    - Success and failure criteria
    - Details of post release monitoring to address criteria. Monitoring of source population and threats.
- Funding – source and long term commitment.
- Animal Ethics Committee approval – Code of Practice (NHMRC 2004).
- Endorsement by proponent’s organisation, and the Department.
- References.
- Attachments

The template for translocation proposals can be found at:
http://intranet/aec/Documents/DPAW%20Translocation%20Proposal%20Template%202014.doc

2. Monitoring and survey design

2.1. Surveying and monitoring

The terms ***survey*** and ***monitoring*** are frequently used in reference to the study and management of flora and fauna.

In general terms, ***survey*** refers to the activity of collecting information relating to a particular subject. A *fauna survey* or *trapping survey*, for example, is the activity of using traps to catch animals for the purpose of examining animals and collecting information such as species identifications and various body measurements relating to those animals. Other types of fauna surveys include nest box surveys, spotlighting and hair tube surveys.

Monitoring refers to the long term appraisal of a particular subject by means of regular surveys to detect trends and changes.

For the purposes of DPaW operational fauna management, survey and monitoring can be defined as follows:

<u>Survey</u>

A single or repeated exercise of sampling to assess presence/absence of species and/or collect biological data on species.

<u>Monitoring</u>

A consistent and regular sampling effort using standard and consistent methods to assess population changes, for example native fauna response to predator control or other variables.

Knowledge of the selected species' biology, behaviour and habitat preferences will assist in selecting an appropriate survey technique (e.g. trapping or spotlighting) and, in combination with knowledge of the area being considered, will assist in selecting an appropriate survey design (e.g. grid or transect) and sites with appropriate habitat types.

2.2. Survey techniques

There are a number of survey techniques used by wildlife researchers and these techniques vary in their reliability of species identification. Techniques are listed here in order of confidence:

1. ***Physical contact***: Trapping, Nest boxes, Mist netting, and Hair tubes
2. ***Visual/auditory contact***: Sighting transects, Bird and bat surveying techniques
3. ***Secondary signs***: Animal signs – nests, tracks, diggings, scratches and scats

Techniques that do not involve physical contact or disturbance to animals may not require a licence or Animal Ethics Committee approval but you should contact Animal Ethics Committee Executive Officer before monitoring to determine if approval is required. You should also familiarise yourself with DPaW Standard Operating Procedures for various survey techniques that are available on the Animal Ethics Committee website.

Much of the information in the following sections has been taken from the Office of the Environmental Protection Authority (OEPA) and Department of Environment and Conservation (2010) Technical Guide – Terrestrial Vertebrate Fauna Surveys for Environmental Impact Assessment (eds B.M. Hyder, J. Dell and M.A. Cowan). This document is available on the OEPA's website. http://www.epa.wa.gov.au/Policies_guidelines/reports/Pages/TerrestrialVertebrateFaunaSurveysforEIA.aspx References provided in text below can be found in the Technical Guide.

A summary of species groups and the major detection methods is provided in Table 1.

Table 1. Species group and major detection methods (Primary detection methods denoted by X, supplementary methods by S)

Group	Pit traps	Funnel Traps	Medium Aluminium Box	Large Aluminium Box	Cage	Spot-lighting from vehicle	Spot-lighting on foot	Head torching	Diurnal Observation/ Active Searching	Searching for tracks & signs etc	Sound/ calls	Recording Techniques including Anabat	Mist netting	Harp traps	Trip lines	Remote camera	Hair tubes
Small Mammals < 30g (eg Sminthopsis)	X		X			S	S			S						S	S
Medium Mammals <2500g (eg Isoodon)	S		X	X	X	X	S			X						S	S
Large Mammals >2500g (eg Petrogale)				X	X	X	S	S	X	X						S	S
Bats (Megachiroptera)									X				X		X		
Bats (Microchiroptera)							S	S	X			X	X	X	X		
Birds						S	S		X	S	X	S	S				
Small snakes <45cm(eg Parasuta)	X	X				X	X	X	X								
Medium-Large Snakes> 45cm (Demansia)		X				X	X	X	X	S							
Small –medium Lizards <150mm(eg Pogona)	X	X	S			S	S	X	X								
Large lizards>150mm (Varanus)	S	X		S	S	S	S	S	X	X							
Frogs	X	S				S	S	X	X	S	X	X					

Office of the Environmental Protection Authority (OEPA) and Department of Environment and Conservation (2010) Technical Guide – Terrestrial Vertebrate Fauna Surveys For Environmental Impact Assessment (eds B.M. Hyder, J. Dell and M.A. Cowan).

2.2.1. Trapping

Trapping is one of the most invasive methods of surveying fauna, but can yield very useful and detailed information on animal biology, abundance and distribution if undertaken effectively. There is risk of injury or death to animals, as well as risk of injury to personnel; however these risks can be significantly reduced by exercising appropriate care.

The well-being of any trapped animal should be the primary concern of the animal handler.

Trapping is generally used for fauna surveys, monitoring, harvesting for translocations and captive breeding programs.

All SOP's mentioned can be sourced from http://www.dpaw.wa.gov.au/plants-and-animals/96-monitoring/standards/99-standard-operating-procedures

2.2.1.1. Cage Traps (SOP 9.2)

Cage traps made of wire mesh are available in a variety of sizes ranging from those suitable for rodents, bandicoots and possums up to sizes large enough for rock wallabies. They operate through a treadle and wire link holding open a door. Animals are enticed into the traps through use of bait. To access the bait the animal must cross a treadle, at the back of the trap, which causes the trap door to be released and locked in a closed position. Traps are available in a rigid or collapsible form, the latter being particularly suitable where large numbers require transportation or carrying any distance.

Bait is usually a ball of oats and peanut butter but sometimes with other additives such as bacon, sardines, fruit, honey, truffle oil, etc, depending on the target species. A number of important considerations should be taken into account when using bait in traps. Firstly, some additives may increase the likelihood of ants being attracted to the trap thus increasing the risk of ant attack on captured vertebrates. Secondly, the inclusion of sardines or other fish can pose a health risk to animals if they have "gone off" prior to ingestion. Therefore it is essential to replace bait daily where these ingredients are used.

Cage traps are used for larger mammal species such as Quokka (Setonix brachyurus), Water rats (Hydromys chrysogaster), Chuditch (Dasyurus geoffroii) and Brushtail possums (Trichosaurus vulpecula vulpecula).

Cage traps may be used as the sole trap type, usually for species-specific projects, or in combination with other types such as aluminium box traps for more general survey and where larger mammal species are expected to occur. The layout of such traps may be along a transect, or as part of an array or grid design. A study of Chuditch by Wayne et al. (2008) showed that the most efficient trap densities for this species was a spacing of 200 metres between traps along transects.

Cage traps require careful placement to minimise exposure to the elements. Additional protection may be provided by placing a hessian sack, calico bag or other insulative material over the traps. This also aids in keeping captured animals calm when the traps are approached for inspection, thus minimising the risk of injury to the animals. Occasionally animals suffer abrasion to snouts and limbs trying to escape through the wire, this should be *carefully monitored.*

For larger macropods (quokkas, wallabies up to 5kg), Thomas or Bromilow traps are often used. These are wire-framed traps with a shade cloth or fabric bag attached. They are more suitable than rigid cage traps for trapping animals such as quokkas and wallabies, however the fabric can be chewed by smaller mammals.

<u>Large cage (Sheffield)</u>

- Galvanised wire mesh cage trap (45cm x 45cm x 90cm) with a treadle release mechanism.
- Used primarily for Quokka and Tammar Wallaby.

<u>Small cage (Sheffield)</u>

- Galvanised wire mesh cage trap (20cm x 20cm x 56cm) with a treadle release mechanism and usually a hook to hold the bait.
- Used for most medium size mammals such as Chuditch, Quenda, Brushtail Possums and Woylies. Will also catch small dasyurids and rodents as well as varanids, large skinks and occasionally birds.

<u>Northern quoll cage (Sheffield)</u>

- Galvanised wire mesh cage trap (17cm x 17cm x 50cm) with a treadle release mechanism.
- Judy Dunlop or Julia Lees (Science and Conservation Division) can be contacted for use of these traps.

<u>Bromilow trap</u>

- Strong cotton or synthetic mesh 'cage' suspended within a collapsible aluminium frame with sliding drop door and treadle release mechanism.
- Used primarily for rock-wallabies but is suitable for Tammar Wallaby and other similar size macropods.
- Not manufactured commercially and is expensive to make.

Reference: Kinnear, J. E., Bromilow, R. N., Onus, M. L., Sokolowski, R. E. S. (1988) The Bromilow trap : a new risk-free soft trap suitable for small to medium-sized macropodids. Australian Wildlife Research. - Vol. 15, no. 3

<u>Thomas trap</u>

- Shade cloth 'bag' attached to a collapsible galvanised steel rod frame by cable ties (for easy replacement of the shade cloth) at the front and shock-cord loops hooked to the rear of the trap. The door and treadle release mechanism is similar to that of a cage trap and the door consists of shade cloth attached to a steel rod frame by cable ties (for easy replacement of the shade cloth).
- This trap was designed by Neil Thomas (Science Division, Woodvale), with assistance from Sheffield Wire Products, specifically for use with small to medium-size macropods.
- Manufactured commercially by Sheffield Wire Products. Two sizes have been manufactured:

frame 30cm high for hare-wallabies and frame 45cm high for tammars and rock-wallabies.

2.2.1.2. Aluminium box traps (SOP 9.1)

Elliott and Sherman traps are brands of collapsible aluminium box traps. Aluminium box

traps are available in a variety of sizes. They all operate by means of a trigger plate on the floor of the trap, which is set off when the animal enters, allowing a hinged door to flick up into the closed position. Similar to cage trapping, bait is used to entice animals into the trap.

Aluminium box traps are typically used in arrays or along transects and are useful in capturing most rodent species (larger rodents e.g. Rattus fuscipes may eat their way out) and larger marsupials including quolls, Mulgara, and bandicoots, providing the appropriate sized traps are used for the expected fauna. Aluminium box traps do not appear to be particularly effective for many small dasyurids or most reptile species although they have worked for targeted collection of certain species including the Kultarr (*Antechinomys laniger*), skinks (e.g. *Ctenotus inornatus*, *C. leonhardii*) and some varanids (e.g. *Varanus acanthurus*). Captures of a variety of other reptile species may occur occasionally, but not reliably.

A number of important issues require consideration when using aluminium box traps. Traps should be located to avoid large numbers of ants and daily observations are required to ensure that ants are not becoming a potential animal welfare risk. Traps should be stable when deployed and checked to ensure they are working well. Both cold and hot conditions may also present problems for animal welfare as aluminium is an excellent thermal conductor.

Where possible, traps should always be placed to take advantage of natural cover and insulation so that the trap does not get over-heated prior to being checked in the morning. Traps should be checked more regularly or closed if there is a risk of animals overheating. Where overnight conditions may be very cold, some form of insulative material should be placed in the trap to form a barrier between the animal and the aluminium surface. If wet conditions are expected, through rain or condensation, traps can be inserted in sturdy plastic bags such as those used by the mining industry for soil samples.

Distance between individual traps usually ranges from 10 to 20 metres and the overall layout may incorporate a combination of different size aluminium box traps.

Experience in a number of regions has shown that capture rates in aluminium box traps often improve after several days, perhaps because animals are initially wary of alien objects with shiny metallic surfaces or unfamiliar scents. Whatever the reason, it is essential to not only consider adequate effort in terms of trap numbers but also to ensure that survey work is undertaken over a sufficient number of days to give the best chance of detecting species (James 1994; Moseby and Read 2001).

<u>Large Elliott</u>

- Aluminium folding box trap (15cm x 15.5cm x 46cm) with a treadle release mechanism.

- These traps are not as robust as cage traps, easily damaged by larger animals and are more expensive hence not widely used.
- May be used for small to medium size mammals such as Phascogales, Quenda and Chuditch.

Medium Elliott

- Aluminium folding box trap (9cm x 10cm x 33cm) with a treadle release mechanism.
- Used for small mammals (up to 25g) such as Phascogales, Mardos, other small dasyurids and rodents, as well as some reptiles.

Small Elliott

- Aluminium folding box trap (8cm x 9cm x 23cm) with a treadle release mechanism.
- May be used for small mammals (up to 30g) but is not often used.

2.2.1.3. Pit traps (SOP 9.3)

Pit trapping in various forms and configurations has been used for many years. It is particularly productive for sampling small to medium sized reptiles and mammals.

Pit traps are usually a form of plastic bucket or PVC pipe buried with the open top flush to the ground, thus providing a means of passively capturing either unsuspecting animals that fall in, or overly inquisitive animals that deliberately enter. They vary in depth and diameter but typical dimensions that are effective for general survey are 20L plastic buckets (450mm deep x 300mm wide) and PVC pipe (600mm deep x 150mm wide). A variety of modifications can be made, for example the incorporation of funnels, to enhance the effectiveness of shallow pits. This restricts the aperture through which animals can escape.

In general, the wider a pit the more effective it is at initial captures and the deeper it is the better overall retention of those captures. Therefore, buckets tend to have significantly higher captures than narrow 150 mm PVC Pipe (Cowan 2004), but the increased depth of PVC pipe (600 mm) is advantageous in retaining some species that are effective jumpers, for example, hopping-mice (*Notomys* spp.) and Mulgara (*Dasycercus cristicauda*) (M. Cowan unpublished). Providing elevated pit-trap covers to shield the bucket from rain or to prevent overheating of captured animals can also increase capture rates of some reptile species (Hobbs et al. 1999).

It is common practice, to use “drift fences” of fly wire, or some similar barrier, running over the centre of a pit and often linking together a number of equidistantly spaced pits (Webb 1999). These fences are buried at their base and are usually 20 to 30 cm in height. Drift fences direct animal movement towards the pits thus increasing the likelihood of capture. Moseby and Read (2001), in their study of chenopod shrub lands in South Australia, found that capture rates where trap arrays incorporated fences were up to five times greater than those that did not.

The design of pit trap arrays has been assessed by a number of authors (Friend 1984; Friend et al. 1989; Hobbs et al. 1994; Morton et al. 1988; Rolfe and McKenzie 2000) and while it is recognised that traps are most effective when used in conjunction with a drift fence the configuration of pits along such a fence is still the subject of frequent debate. For example, should traps be positioned singly, in pairs or in larger numbers along a continuous fence line, and what spacing should exist between traps? There are some logical aspects to note in these design considerations that should help in devising a suitable trapping design.

If traps on a long drift fence line are positioned too close together they make each other partially redundant as an animal caught in one trap may otherwise have continued on and been captured in the next trap, had the first not been present. It may also be the case that if spacing between traps is too great, then some animals will turn away from a fence and not be captured at all. However, providing there are enough traps in the overall sampling design, this is not a significant issue as most of the effort in establishing a trapping grid, particularly in hard substrate, is usually invested in installing the pits rather than the fence. It is therefore important to ensure captures per trap are maximised and this is achieved by having an appropriate distance between traps.

If a single trap is located in the centre of a drift fence it can be expected that at least 50% of animals coming into contact with the fence will miss the trap. If animals contact the fence to the left of the trap and continue to move left they will miss the trap and similarly if contact is to the right of the trap and they move right they will also avoid the trap.

It is advisable to check the area for ant foraging trails prior to installation of pit traps and you must be prepared to close individual traps or entire lines should ants become unmanageable.

Overheating is also a potential problem in bucket-type pits, especially in northern latitudes in late spring-early summer, when the sun is at its highest; under these conditions, temperatures in uncovered buckets can reach 66oC (Hobbs and James 1999). The pits themselves need to be shaded, or adequate amounts of shelter need to be placed within the pits, and it may be necessary to check pits more frequently than once a day, or to close them under really hot conditions. This is less of a problem with deep, narrow pits.

<u>Pitfall</u>

- Plastic 20L buckets with snap on lids 30cm diameter, 40-45cm deep.
- PVC pipe 600mm deep x 150mm wide.
- Flywire fence 20-30cm in height
    - Use 5-7m fences on each pit if recording movements of individual animals over a grid.
    - Use a continuous fence through a line of pits, or web, to maximise the total number of captures.
- Ensure adequate refuges/cover are provided in the bottom of buckets.

2.2.1.4. Funnel traps (SOP 9.17 draft)

Funnel traps have been utilised in various forms overseas for many years (Fitch 1951; Clark 1966; Hall 1967), although their common use in Australia has only occurred relatively recently. Funnel traps are generally made from some form of mesh (e.g. dense shade cloth) covering a wire framed rectangular prism with small opening funnels at either end. Laid parallel to a drift fence, animals may enter the internal space through either funnel but have difficulty in finding a way out.

Most survey work using funnels has involved the placement of pairs of funnels, one either side of a drift fence, alternating with pit traps along the fence. Funnels are effective in capturing reptiles such as snakes and some larger varanids that readily escape from pit traps and there may be other reptile groups that they are at least as effective in capturing as are pits, although this is unclear at this stage. Funnels do not appear to be effective in capturing mammals. A significant advantage of funnels is their ease of deployment in sampling areas where the substrate precludes establishment of pit lines, such as on granites or other similarly hard surfaces.

As funnel traps are positioned on the surface they have even greater exposure to temperature variability and extremes than the bottom of pit traps, so their use in hot conditions should be considered very carefully or avoided. Where possible, and if the sampling design permits, they should be positioned under natural shade but failing this, adequate shade and insulation may be provided through covering the top with grass, Spinifex or leaves etc. If it is necessary to operate in extreme conditions (i.e. summer months through the semi-arid to arid parts of the state) and if adequate protection or insulation from radiant heat cannot be provided, then leaving the trap so it cannot catch animals following the morning check, and returning in the late afternoon to re-set the funnel, should be considered as death from exposure to daytime captures is almost certain.

2.2.1.5. Key points to remember when setting traps

- Traps must be set so that they are **not** readily visible to the public to avoid curiosity and possible theft or interference.
- Cage and Elliott traps should be set in level positions using natural cover wherever possible. Traps must be kept well shaded during the warmer months as animals can easily die of heat stress.
- Cage traps must be covered with hessian or an appropriate cover for the weather conditions (insulating in the cold and thermo-regulating during the heat). This provides animals with security and shelter from the elements, reduces stress and protects the bait from the elements
- Standard bait for Elliott traps and small cage traps – peanut butter, rolled oats and sardines combined into a moist but firm doughy mix
- Variations on the standard bait recipe or other baits may be more appropriate for particular species e.g. oats and apples for Tammar Wallaby and Quokka or "Chuditch bait" (also known as "smelly bait") where Woylies are very abundant
- Ensure no bait or other material has rolled underneath treadle plates in cage and Elliott traps
- Check and test trap door and mechanism before leaving each cage and Elliott trap
- Keep a count of all traps placed out

<u>Checking and picking up traps</u>

- Traps must be checked for captures early in the morning and completed within Animal Ethics Committee required times (usually 3hours after sunrise).
- Traps should be closed after the morning check and reopened in the afternoon to avoid non-target captures.

- Do not set more traps than you can handle
- Bait must be checked daily in every trap and replaced where necessary
- No bait reduces chance of capture and therefore reduces statistical validity of trapping results
- Do not forget to reset trap after processing a capture
- Keep a tally of traps checked or picked up. If your final tally is less than the number of traps put out, then find the missing traps!

Points on pit traps

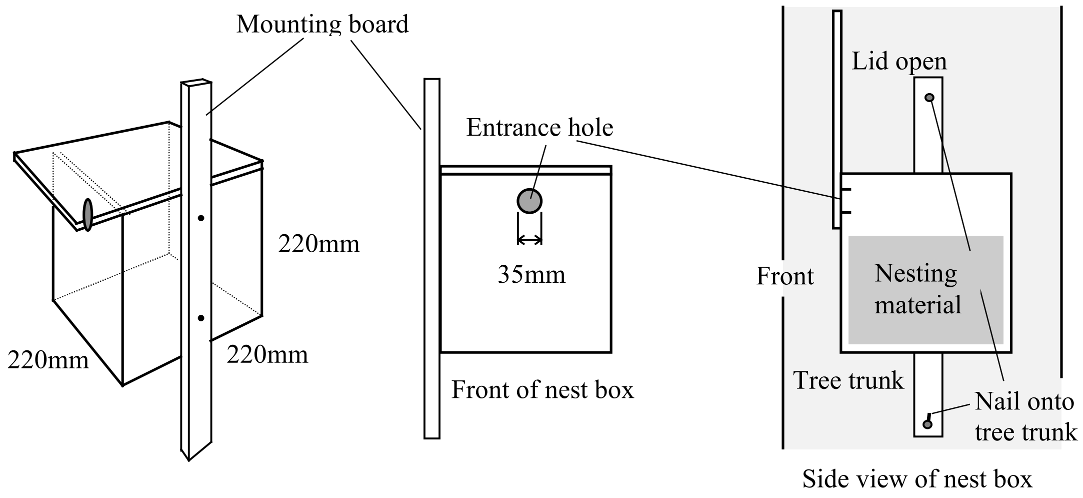
- Pit traps must be provided with some form of shelter at the bottom for trapped animals – egg cartons or polystyrene food trays with corner cut out
- Pit traps should be closed if invaded by ants because they harm and often kill trapped animals. Spraying with *Coopex* is an effective way to control ants in and around susceptible pits and can negate the need to close a pit. Avoid spraying inside the pit trap.
- Pit traps should be closed if there is heavy rain and/or very cold temperatures
- When closing pit traps, ensure that lids are secure and cover lightly with soil to protect from sunlight (UV).

2.2.2. Nest boxes (SOP 9.4)

Nest boxes can be used to survey or monitor small arboreal mammals that are difficult to trap, particularly the Brush-tailed phascogale. Pygmy possums, mardos and sometimes dunnarts also use nest boxes of the same dimensions.

Nest boxes can be erected along transects or in a grid – for general survey and monitoring purposes, ten or more nest boxes should be erected along road transects at 200m intervals. Nest boxes should be positioned to provide shelter from the sun and rain and numbered.

Nest boxes can be left for long periods of time without checking, however, they should be monitored twice a year in January/February and in June. Spare next boxes should be kept on hand to replace any damaged ones.



Nest box construction

2.2.3. Mist net, harp net and trip lines (SOP 9.10)

Consult DPaW Species and Communities Branch if considering the use of these survey methods.

Mist nets are used for catching small birds and bats in flight, while trip-lines and harp traps are only used for catching bats. As with other forms of trapping, mist netting poses risks of injury or death to animals caught and must be conducted with care and expertise. The use of nets and trip wires is supervised and regulated by the Australian Government under the auspices of the Australian Bird and Bat Banding Scheme (ABBBS) and persons using nets and trip-wires must be licensed. Data is recorded and forwarded to ABBBS for inclusion in the national database.

<u>Bats</u>

Working with bats can pose potential health risks (see Section on 'Hygiene and fauna handling techniques') and handling bats needs to be undertaken carefully.

Mist nets, harp traps and trip lines are effective in the capture of the smaller low flying bat species although they have very different modes of operation. In addition, use of these devices can occasionally be the preferred way to sample the presence of particular species such as the non-echolocating nectarivorous blossom bats.

Mist nets are constructed from a mesh of thin fibres which are difficult for bats to detect. When unfurled and tensioned properly mist nets have four or five horizontal pockets spaced evenly across the height of the net. When a bat flies into the net it generally drops into the pocket below, where it becomes entangled in the mesh.

Harp traps have a series of vertical tensioned nylon wires spaced evenly along a rectangular frame with a canvas bag positioned directly below. The nylon wires are difficult for bats to detect and they fly into the wires getting caught between them and subsequently drop down into the canvas bag. The bag has clear plastic flaps on the inside that restrict the bat from climbing out.

Harp traps have a distinct advantage over mist nets in that they do not require an operator to be continuously present and can just be checked for captures early the following morning. While mist nets are a far more effective method for capturing bats (Jones et al. 1996), they need to be constantly monitored so captures can be immediately disentangled. Mist nets are also relatively cheap, easy to set up and can be used under a wide variety of conditions.

Trip lines consist of a grid of fine fishing lines set over small water bodies. When bats fly in to drink they hit the lines and fall into the water then swim to the edge where they are collected. Water bodies with trip lines must be monitored constantly from dusk to dawn. Larger waterbodies may require more than one person to collect the bats, which should be allowed to dry before being released. The technique is best used on bodies of water with surface dimensions less than 50m across (University of Ballarat n.d.).

Positioning of mist nets, harp traps and trip lines is critical to success and consideration should be given to locations that intersect foraging, drinking and commuting pathways or are adjacent to roosting sites. These areas may include caves, creek lines, and the periphery of dense vegetation or over pools of water. Often an early evening observation will assist in assessing the potential of a specific location.

Trip lines can be useful for the capture of bat species not normally captured in harp traps (Helman and Churchill 1986) or where a dam is not suitable for mist netting. When installed over dams and small water bodies in warm weather, they can assist in capturing high flying species such as freetail bats (Mormopterus spp.). Like mist nets, trip lines are labour intensive, require constant supervision and are suitable at relatively limited sites and in suitable weather periods. They may be utilised, however, when conditions are suitable as they assist in obtaining individuals of species not readily captured by other methods (Murray et al. 2002).

Performing bat surveys using these types of nets and traps have a number of disadvantages compared with ultrasonic detector based surveys. The five most important of these to take into account when designing a survey are:

- mist nets and harp traps can only sample an extremely small area relative to that used by free-flying bats and so site selection is extremely important;
- mist nets and trip lines must be monitored continuously;
- bats left in harp traps overnight can be attacked by terrestrial predators or succumb to exposure;
- trapping will under-represent high-flying species and very low/slow flying gleaning species; and
- traps can be detected by bats, so equipment is more effective if it is positioned in situations where the bats are surprised, deceived or cornered. Bats also have a good capacity for learning, so traps are less effective on subsequent nights, unless shifted and for some considerable time after sampling at a location.

2.2.4. Hair tubes

Hair tubes are generally used as a supplementary technique in combination with other trapping or observation techniques. Hair tubes do not trap animals, but their method of evidence collection (by hair removal) will have an impact on the animal. This technique requires an ability to identify hair either through physical identification (microscope and reference collection) or DNA techniques. Mammal hair can be identified to species or genera in most cases; identification is based on the cross section and scales on the hair. The advantage of hair tubes is that the technique can be used to survey large areas cheaply for the presence of species and the tubes can be left in place for extended time periods without checking as there is no risk to animals.

Two types;

1) A simple type consists of plastic sheet lined with double-sided sticky tape and bent into an arch and pegged into position. Choose a position where animals are likely to be corralled through the arch.
2) A plastic tube or funnel with bait in it. If designed well it is suitable for mammals of all sizes. Placement with this type is not as important as the bait will attract animals.

2.2.5. Remote cameras

For a comprehensive summary of remote camera trapping use and monitoring guidelines refer to Meek PD, Ballard G and Fleming P (2012). *An Introduction to Camera Trapping for Wildlife Surveys in Australia.* PestSmart Toolkit publication, Invasive Animals Cooperative Research Centre, Canberra, Australia. www.feral.org.au/camera-trapping-for-wildlife-surveys/

Remote digital cameras triggered by infrared movement sensors are readily available. Cameras are particularly useful for many of the larger and distinctive mammals although the definition and detail of images is not always ideal, particularly where an animal is distant or relatively small, or where cameras have infrared filters.

Motion sensors are set so that they only activate the camera when an animal is in the field of view and close to the camera. Cameras can be left to operate for many days through to months, depending on batteries, and therefore provide information beyond what is attainable while in the field. Locations that are suitable for camera use include along tracks and runways where there are signs of activity, focused on burrow entrances or on some form of lure or bait.

2.2.6. Observation (SOP 7.2)

There are a number of observational activities that are usually required for effective fauna survey, as trapping and other techniques alone are unlikely to maximise detection of species present in an area. These techniques may include:

- Spotlighting/head torching;
- Active searching;
- Searching for tracks and other signs;
- Bird observation; or
- Bird or frog calls (including recording).
- Bat detector surveys
- Sand plots

Each of these techniques is relevant to specific faunal groups and requires some degree of expertise to maximise the information gained for the effort undertaken. Less experienced individuals will invariably detect fewer species than those who are more experienced; as a result consideration should be given to how equal and adequate effort is applied to each site. A good summary of the use of these techniques for herpetofauna survey can be found in Bush et al. (2007).

Spotlighting/head torching

Spotlighting and head torching at night from vehicles and on foot are important survey techniques as much of our fauna is nocturnal or crepuscular, particularly many threatened taxa, and many of these are more often observed than trapped.

Spotlighting may be useful for a variety of species including mammals, nocturnal birds, geckoes, snakes and frogs. Spotlighting can be done from a vehicle which covers large distances along roads and tracks. Portable spotlights can be used while walking to investigate at a finer scale or in areas where vehicle access is not possible.

Some species will remain motionless when caught in the high intensity beam of a spotlight and may be difficult to see, e.g. quails, while others will immediately take flight and be difficult to identify. It is therefore essential that some idea of what may be encountered is known beforehand and that experienced individuals are involved in the exercise.

Spotlighting on foot may be effective for cryptic species such as button-quails which may otherwise be unseen. Some nocturnal bird species are sensitive to disturbance, for example, Grass Owls and Masked Owls are particularly sensitive during the breeding season and Bush Stone-curlews will abandon nests if repeatedly disturbed (DPaW NSW 2004).

Spotlighting from a vehicle may involve a number of people searching with a spotlight, recording and catching fauna as required. Fauna observations are made as the vehicle is driven at low speed, along a predetermined transect. Where fauna cannot be immediately identified it may be necessary to stop the vehicle so the surveyor can collect/observe fauna. When spotlighting from a vehicle, consideration should be given to preparing operating procedures to ensure the safety of the personnel participating in the spotlighting.

The dimmer light (compared with spotlights) makes head torching a more useful method for detecting the eye shine of vertebrates like geckoes and frogs. In general, the light colour from incandescent lights is better than that from the newer LED torches, particularly for seeing reflected eye shine, but this is somewhat of a personal preference.

Both spotlighting and head torching are most productive for reptiles on warm evenings (Read and Moseby 2001) when activity for many species becomes elevated. However, cooler conditions should not necessarily negate undertaking night work. Head torching for frogs is most successful after rains.

Active searching

Active foraging for reptiles and amphibians will involve searching particular microhabitats and may include digging up burrows, turning over rocks and logs, splitting fallen timber, raking soil and leaf litter, peeling off bark and searching soil cracks around water bodies and holes in fence posts.

Effective active searching requires some knowledge of which species could be present in an area and their specific habitat preferences. Active searching can be physically demanding, particularly in warm conditions. Timing is important as in hot and dry conditions reptiles are hard to detect and with high body temperatures they may also be very quick and elusive. Therefore, in hot conditions searching early in the day may yield the best results. This method frequently provides considerable supplementary information to trapping programs as many species that may have low capture rates in traps, may be readily caught by hand, for example when trapping reptiles at cooler times of the year. It is important to minimise impact on habitat and all rocks, logs and debris should be returned to their original location and orientation where possible.

Searching for tracks and other signs

Searching for tracks, diggings, nests, scats, claw marks on tree trunks and other signs requires persistence, well developed observational skills and knowledge of the natural history of the local fauna. Identifying tracks also relies on knowledge of the size and shapes of animal foot pads.

These activities are well suited to detecting species that are not readily trapped because they are either too large (e.g. some varanids), avoid traps (e.g. arboreal species) or are at low densities (e.g. some snakes). Species with clumped distributions, for example Mulgaras (*Dasycercus cristicauda*) or Bilbies (*Macrotis lagotis*), are often difficult to detect with standardised trapping regimes but may be readily detectable through observation as much larger areas can be assessed than just the specific trapping locations.

Sand is an ideal substrate for looking for tracks although wind and rain will often mask these

quite quickly, particularly for small animals that only make shallow impressions. Look in places without vegetation, where the ground is soft enough to take the imprint. Creek and dam banks, roadside dust, dried out puddles and clay pans and snow-covered ground are all suitable sites for tracks. Best tracks are found in the early morning as the imprint holds better and the tracks are easiest to see in slanting light, ie early morning or dusk.

Diggings often occur in a variety of substrates and can last for quite some time, often many months or even years after an animal has been present, so detection of these may only indicate general or historical usage of an area but not necessarily presence at the time of observation. Therefore it is often desirable to undertake targeted trapping or some other form of confirmation to be certain of correct identification and determine the continued presence of a species of interest.

Scats are fairly characteristic sign of a species and can be easy to find. Scats can indicate relative time of habitation (recent, long past) and what an animal has been eating. They are also useful indicating the owner of a shelter. A couple of disadvantages of using scats for identification are; some animal's scats are difficult to find, and they can look like those of a similar species, especially if they have the same diet. This is made easier with the help of distribution maps and habitat information.

Other signs include:

- The width of a burrow or hollow entrance as well as its depth can give an indication of which animal constructed the burrow.
- Feeding signs - mammals often leave obvious traces where they have been feeding. For example the Short-beaked Echidna broaches ant and termite nests with a conical whole up to 20cm deep, with the mark of its snout at the end. Feral cats may leave the tail and hind quarters of a small possum, or the wings of a bird.
- Markings on trees - many animals leave marks on the trunks of trees. Some possums gouge wood-boring grubs out of trunks of wattles and other trees. The urine marking of foxes, dogs and dingoes, if frequently on the same tree, can leave a mossy-looking stain on the trunk directly above it. Possum scratching's up trees can be highly visible if the tree-hollow is well used.

Bird observation

See OEPA 2010, DEWHA 2009 and Birdlife Australia website for comprehensive information on monitoring birds through observation.

Birds are one of the more readily observable faunal groups and there have been a wide variety of methods proposed for standard site assessment (Bibby et al. 2000; Craig 2004; Craig and Roberts 2001; Gregory et al. 2004; Loyn 1986). These may incorporate fixed time and position counts; transect searches, area searches or variations and modifications to any of these techniques.

Bird surveys should be conducted in the period of optimal activity. Typically this may be post-dawn and before dusk, but in hot climates in very open habitats, such as low sparse samphire, survey may need to begin before dawn, as the dawn chorus might be the only time that some species can be readily detected. Bird activity is lower in wet, windy or extremely hot conditions.

Birds can be recorded in terms of presence/absence or a measure of abundance. It takes much more effort to get abundance data compared to presence/absence data. Abundance data can be important in providing information on the comparative importance of different habitats, provided allowance is made for bias caused by visibility differences between habitats. In addition, it is often important to understand temporal variation in abundances before spatial variation in abundance can be interpreted (e.g. Ives and Klopfer 1997).

Birds often respond to different components of the environment compared with terrestrial species, and ideal locations for trapping grids may not represent the ideal location for conducting bird surveys. Therefore the location of trapping grids will not always be the optimum location for bird survey. For example a stand of flowering grevillea species are likely to yield additional bird species.

All of these methods are highly observer dependent and different individuals will have varying degrees of success. To reduce these biases it is important to record sites a number of times and, where multiple observers are involved, ensure that each site is not consistently assessed by the one observer who may be more or less skilled than others on the project, i.e. rotate the skill levels across all sites to ensure some degree of consistency. Alternatively two or more observers may work at the same site concurrently. Imitating the calls of a bird will frequently entice a variety of species in close so as to allow a visual identification.

Two common methods for bird surveys are area searches and point counts. Area searches involve walking around a designated area for a pre-determined period of time. The Birdlife Australia Atlas project uses 20 minute surveys where an experienced ornithologist records numbers of each species seen while actively searching a 2 ha area. Area searches can be used for either density estimates (especially in open habitats) or for species richness studies. Larger areas (e.g. 16 ha) will be more appropriate in arid regions. Point count methods involve making observations from a pre-determined series of points or habitats for a predetermined period of time. For example, York et al. (1991) conducted a series of 10 minute observations at five points, 100 m apart along a 500 m transect. Such methods are most appropriate for population density estimates in dense habitats.

Both area searches and point counts have advantages and disadvantages, and the choice of technique will depend on the objectives of the survey. If the aim of the survey is to record a species inventory and obtain the most species in the shortest amount of time, an area search has a slightly higher chance of recording small cryptic species. Walking through the habitat also increases the chance of flushing more cryptic species. Some species may require specific search techniques. For example, raptors tend to use thermals on warm days and can be spotted from high ground overlooking the canopy.

The timing of surveys should take into account seasonal migrants e.g. waders (especially as many of these are listed in International Agreements), cuckoos and some nectarivores. The amount of time spent surveying each site will depend on the nature of the habitat. Complex habitats are likely to have higher species richness. Dense vegetation may require more survey effort than open vegetation where species are easier to detect.

The identification of water birds especially migratory shorebirds can be particularly difficult compared to other birds (observed from great distances e.g. mud flats, middle of lake; different stages of molt affecting plumage colour and presence of vagrants) and it is important that surveyors have experience in their identification and survey techniques.

Count surveys are the preferred technique for migratory shorebirds. However, when it is not possible to survey the site during the appropriate time, and good regional information is available, a thorough habitat assessment will identify potential habitat. The characteristics of the site (landform, hydrology, flood levels, etc.) should be assessed and used to predict the extent of migratory shorebird habitat. Where possible, survey methodologies should be consistent to allow comparison between data sets. Methods for counting migratory shorebirds are outlined by DEWHA (2009).

Survey effort will depend on many variables. It is recommended that at least two people undertake the counts and surveys are replicated during the period when survey is undertaken. For detail on recommended survey effort see DEWHA (2009). Further information is available on the Birdlife Australia website http://birdlife.org.au/.

Bird and frog calls

Birds and frogs produce audible calls and recording of these will often produce information in addition to that gathered through other survey techniques.

The optimum time for listening for bird calls is at dawn and over the following few hours, particularly on still mornings. However, birds may call at all times of the day and even the night so it is important to always be listening while in the field. Listening for calls at night is a useful way to detect presence of many nocturnal species, at least in the breeding season. Bird calls may vary throughout the day as well as across a species geographic range. There are a number of resources for assisting in song identification such as the CD set produced by the Bird Observers Club of Australia (BOCA 2001).

As with visual observation for birds, it is important to accurately record the location of individuals of interest to ensure they are assigned the actual habitat in which they occurred. For example records from a wooded drainage tract adjacent to a site with quite different habitat characteristics should not be assigned to that site without reference to the actual location and habitat attributes.

Different frog species call in different seasons, therefore survey timing needs to reflect this. It is only the male that calls and this is primarily during the breeding season. Calling in the south west is mainly during rains in the late autumn and winter months, for the Kimberley during the wet season, and over most of the arid zone following heavy rainfall (see Table 2). Dusk and the early part of the night are the best times to listen for calls. Where surveyors are not familiar with calls they should locate and identify the calling animal or record the sound for later identification. A range of frog calls from across Western Australia can be heard on the Western Australian Museum Frog watch website: http://museum.wa.gov.au/explore/frogwatch

Calling frogs can be difficult to locate as the resonance from the call may give the impression that the animal is somewhere else other than its true location. Experience in knowing preferred calling locations will help. Individuals which are hard to locate can be found by triangulation. This is often easier with two people, enabling the direction of the call to be determined from two positions simultaneously (Bush et al. 2007).

Playing pre-recorded calls of target species through amplification (call playback) will improve the chances of locating bird and frog species. In a study on a number of nocturnal bird species, Kavanagh and Peake (1993) found that call playback more than doubled detection rates of all species. Call playback for nocturnal birds provides better results in the early evening or before dawn. Further information on call playback techniques is available in DEC NSW (2004).

Bat detector surveys

Bats constitute a significant proportion of mammal richness across Australia. There are two basic groupings, primarily insectivorous species that use high frequency echolocation to find and catch their prey, and primarily frugivorous species that do not. Bat survey is often more problematic than for other vertebrate groups. Bat detection devices, such as the Anabat system (Titley Electronics, Ballina, New South Wales) or the range of systems available from Pettersson Electronik (Sweden) or a number of other manufacturers, have become important tools in detection and identification of these species. These systems work through converting ultrasonic frequencies into audible signals that can be recorded on a tape, minidisk, and compact flash or directly to a computer hard disk. Analysis of the call structure can then be undertaken on a computer with appropriate software. This system can be used in conjunction with a visual/aural based survey for the non-echolocating large fruit-bats (e.g. Pteropus sp.) and a net/trap based survey for the small non-echolocating nectivorous species (e.g. Macroglossus minimus).

Identifying echolocation calls is a difficult task as bat call structure is complex and not all species can be easily distinguished from one another this way (O'Farrell et al. 1999). A number of good quality call recordings are often needed to confirm the presence of particular species. There are four basic methods (heterodyning; frequency division; continuous recording of ultrasonic signals; time expansion) for analysing these call recordings.

All require access to a library of reference calls from the bats of the study region to enable comparison of the features of the recorded calls.

Sand plots

Sand pads may be a useful way of detecting the presence of certain species although the reliability is very dependent on the observer's skills and knowledge of animal tracks.

The technique entails clearing a series of square or rectangular plots of sand and identifying tracks in the sand. It is particularly useful for surveying small, nocturnal or otherwise secretive and inconspicuous mammals, particularly foxes and cats.

Tracks can usually be identified to family or genus but often it is difficult to distinguish tracks of closely related species unless there is a marked difference in size and shape. There may also be differences in size of tracks between sexes and different age groups within a species.

The use of other information such as scats and known distributions and habitat preferences can help to narrow the identification to species level.

Tracks are most identifiable in the early morning before the sand has dried and the wind has blurred them, and are more easily seen in slanting light. Tracks are also most identifiable in clean, firm and slightly damp sand. Where this type of sand occurs naturally at a survey site, plots can be made simply by clearing and raking smooth a square plot at the desired locations. Where such sand forms the main substrate along extensive vehicle tracks and firebreaks the technique can be extended to using the tracks as a 'plot' by smoothing a 1.5-2m wide strip or batter to batter across the road. In most cases sand will have to be brought in specially to create the sand plots. The best sand to use is the yellow 'brick-layers sand' often used on building sites and it is very important that this sand is clean and declared free of *Phytophthora sp.* and other known plant pathogens.

The distance between neighbouring sand plots will depend on the species targeted and the size of the area being surveyed. For small dasyurids and rodents sand plots can be as close as 50m. For medium-sized mammals and as a general purpose survey, use a 200m – 500m distance. The sand plots can be split up into separate transects of 20 plots each to cover a wider area and range of habitats.

2.2.7. Radio and satellite telemetry (SOP 13.4)

This technique enables activity and movements of individual animals to be monitored. Transmitters are fitted to animals, and emit radio signal pulse at a unique frequency that is picked up with a radio receiver and aerial. The technique is obtrusive to animals (fitting radio/GPS collars) and can cause problems. The equipment is very expensive and generally only used for translocations and research projects. Science Division or Species and Communities Branch should be contacted before radio-telemetry is used.

2.2.8. Scat and stomach analysis

Checking scat and pellet contents

Bones and hair samples collected from owl pellets or the scats of carnivorous mammals can provide valuable additional information on the presence of other vertebrate species. The best locations for finding material are below the nests or perches of raptors, along breakaways, under rock overhangs or in cave entrances although scats of mammal predators such as Quolls and Dingoes may be found out in the open. The positive identification of bone and tooth fragments collected this way can be problematic and may require the expertise of a paleontologist. Hair samples may be able to be identified through the use of an electronic key (Brunner and Triggs 2002) through reference collections or comparative material in museum collections. The collection of material from a specific location does not mean that it is the origin of the remains as birds and large predators can forage over large distances.

Examination of feral predator gut contents

Feral predators such as cats and foxes are known to feed on a variety of native vertebrate fauna (Martin et al. 1996; Risbey et al. 1999). Therefore the examination of the gut content of feral predators can provide valuable records on the presence of other vertebrate species not obtained using primary survey techniques. For example, during fauna surveys of the Diamantina Shire and the Mount Moffatt Section of Carnarvon National Park, Queensland, the gut content of feral cats was the only source of records of particular species e.g. Limnodynastes fletcheri (Long-thumbed Frog) and Acrobates pygmaeus (Feathertailed Glider) respectively (G. Porter pers. com.). Examining the gut content of feral predators obtained for example, as road kill or by shooting or trapping, can be useful in establishing additional species occurring in the vicinity.

2.3. Survey design

The target species and the purpose of the survey will determine the most appropriate trap types and which design is most appropriate for the survey. The above descriptions will help in selecting the most appropriate trap types. What is the most appropriate survey design?

Some designs may be more effective than others and even suboptimal ones may provide sufficient data for a particular purpose given adequate effort in terms of number of traps and/or time spent sampling. The designs outlined below highlight the benefits and shortcomings of different design to assist staff when designing surveys or monitoring projects.

Survey design will vary with the nature of the environment being investigated, its spatial extent, and the species targeted. Detection of a threatened species may only require a limited number of techniques such as ground searching for signs of presence, followed by targeted trapping. Cryptic species may require a more systematic approach which may include targeting known habitats with appropriate trap types or recording devices. Documentation of assemblages or species richness will generally require a more comprehensive approach incorporating a number of techniques applied with consistent and adequate effort. Factors such as survey timing and duration, and the number, type and layout of traps, are key elements.

Parameters that also need to be considered will include numbers and types of traps, their layout and the number of days over which they are operated. The following sections provide guidance on survey design.

Site selection and sampling effort

When selecting survey and monitoring sites, consideration needs to be given to:

- Accessibility – seasonal road conditions/road maintenance
  - vegetation disease status (dieback)
  - security - likelihood of theft or interference
- Management – fox control implemented
  - land use/status
  - recreation impacts
  - fire management regime – alterations to protect or enhance habitat

The number of sites monitored will vary with each survey and is depending up factors such as the type and variety of substrates, vegetation and topography. Sites should be chosen with consideration of the question being asked, e.g. sites may be positioned well within the habitat type to remove edge effects in sampling, or if the question relates to ecotones sites should be positioned on the periphery of habitat types.

The sampling effort (in terms of design, duration, timing and sampling technique) needs to be considered carefully to ensure that the aims of the project will be met.

Timing

Western Australia can be divided into three broad climatic regions based on Beard's (1980) Northern, Eremaean and South West Botanical Provinces. A survey must consider both seasonality and the timing of peak activity, particularly for herpetofauna or other species with temporal variability in activity.

The best time for survey within each province should be assessed in relation to the geography of the survey area, expected climatic conditions and type of weather over the preceding months. For example, pit trapping for reptiles along the south coast would usually be most effective in late spring or early summer, rather than early spring due to cooler temperatures when compared with the northern extent of the South West Province (e.g. How 1998). Refer to OEPA *Technical Guide – Terrestrial Vertebrate Fauna Surveys for Environmental Impact Assessment* for recommended timing for vertebrate fauna surveys across each province:

Some species, such as the dasyurids (e.g. Chuditch, Phascogale) are seasonal breeders and are most mobile and easily trapped during their mating season. Trapping later in the year when they have dependent young can be detrimental and should be avoided.

When planning the timing of fauna survey, animal ethics must be considered to ensure that animals that are breeding, lactating or have dependent young are not unduly stressed.

Seasonal or repeat surveys

The activity patterns of fauna are often closely linked with seasons and sampling across these will produce a more comprehensive understanding than can be obtained from just one season. For example, in the South West Province most reptile species breed in mid to late spring when many species are particularly active and readily caught. However late summer to autumn is when the offspring emerge and are readily trapped.

Repeated surveys will generally yield higher numbers of species than single surveys, and will account for temporal differences in activity patterns (Cowan and How 2004; How and Cooper 2002; Moseby and Read 2001).

For mammals in particular, repeated or frequent trapping may have an impact and consideration must be given to ensuring frequent trapping does not modify an animal's behaviour. Some animals can become "trap happy", i.e. the reward of a free morsel of food can entice animals to keep getting caught in traps, or animals can become "trap shy", i.e. they may associate traps with bad experiences and avoid them. As well as having detrimental impacts on the animals, these behaviour changes will affect the results of the trapping surveys.

Trapping design for terrestrial mammals and herpetofauna

Choose a point or quadrat design if:
- Observing species presence
- Assessing the distribution of a species
- Require flexibility of random sampling

Choose a grid design if:
- Measuring population density and dynamics
- Measuring individual animal movements
- Targeting small, less mobile species
- Sampling a small area (<5ha)

Choose a transect design if:
- Measuring relative abundances
- Targeting wide ranging species
- Sampling a wide range of habitats over a large area
- Require convenience of utilising pre-existing tracks

Choose a web design if:
- Measuring density and relative abundance
- Have good access by foot
- Sampling a small or medium size area
- Have good detectability of species of interest
- Have resources to cope with demanding logistics

Trap locations or spacing will depend on the trap types and target species. For example, Elliott traps are generally placed closer together than cage traps because they target smaller animals that move over smaller areas. Generally, pitfall traps and small and medium Elliott traps are placed 10-50m apart, and small cage traps are placed 20-200m apart depending on what species are targeted. For example, small cage traps can be placed in lines of five to ten traps, 20-50m apart to target Quenda, or along road transects at 200m intervals to target wider ranging species such as Chuditch and Woylies. More than one trap type can be incorporated into a survey design.

Having chosen appropriate trap types and survey design, trap locations need to be pre-determined using maps showing vegetation types, topography and access routes. For general surveys, traps are best located so that as many habitats/vegetation types as practical are sampled.

Trap locations must be marked clearly in the field with the correct trap number written on the marker. Trapping grids or transects and trap types, locations and numbers must be clearly recorded on data sheets and marked on a detailed map attached to the data sheet. The personnel setting up a trapping transect or grid may not always be the ones checking the traps or picking them up. It is imperative that all traps taken out into the field can be accounted for before and after a trapping survey. **ALL TRAPPING – KNOW WHERE YOUR TRAPS ARE!!**

Some frequently used survey designs

- Surveying for medium size mammals and monitoring medium size mammals in general, use a minimum of 50 cage traps set at 200m intervals along road transects. This is the design used for *Western Shield* monitoring. One or two Elliott traps at each trap point can also be added to sample small mammals and some reptiles.
- Surveying for presence of reptiles, frogs and small mammals and monitoring relative changes in abundances of these species use pitfall traps. Based on previous studies it is expected that generally 10 to 12 pit traps should be used at a site during inventory surveys. However, trap numbers are dependent on site characteristics, for example in hard substrates pit trap placement may be difficult and other trap types may need to be increased. A combination of trap types can be used for example deep PVC) pipes and 20 L buckets; or 20 L buckets on their own. Narrow diameter (150mm) PVC pipes are not recommended for use alone as they are not as efficient for small vertebrate captures as 20 L buckets. Deep narrow PVC pipes may be efficient in capturing some rodent species e.g. hopping mice which are known to jump out of 20L buckets.
- Monitoring changes in population densities and movements of small to medium size vertebrates use integrated grid design (see Fig 1 illustration) incorporating different trap types.

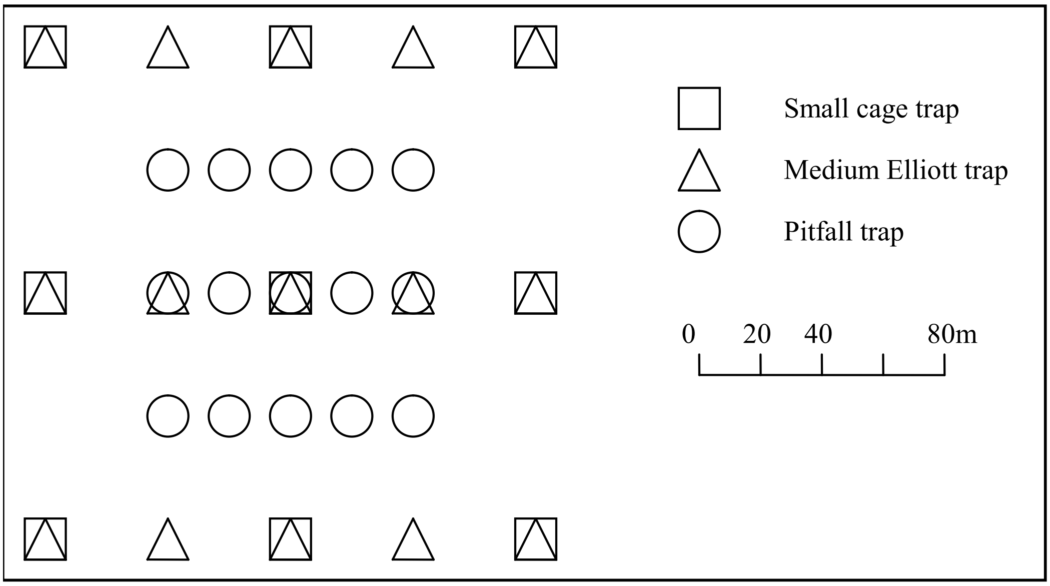


Figure 1. Integrated trapping grid design.

3. Animal welfare considerations

3.1. Animal Welfare Act 2002 and Animal Welfare Regulations 2003 (General and Scientific)

3.1.1. Licence to use animals for scientific purposes

In December 2002 the Animal Welfare Act was gazetted. This Act replaced the *Prevention of Cruelty to Animals 1920* Act. The *Animal Welfare Regulations 2003* (General and Scientific) followed shortly after. Any additional information about the act and regulations and how the department follows these can be sourced from the intranet at http://intranet/aec/Pages/Legislation.aspx

The Animal Welfare Act applies to all vertebrate animals (excluding fish) in Western Australia and requires all establishments that use animals for scientific purposes (including DPaW) to be licenced.

This is a licence to use animals for scientific purposes DPaW has this but the licence conditions require the licensee to adhere to the *Australian code of practice for the care and use of animals for scientific purposes* and this includes having access to a properly constituted Animal Ethics Committee. All projects or activities involving the use of animals must be assessed via written applications and approved by the Animal Ethics Committee before commencement.

Before commencing any activity involving the use of fauna for scientific purposes the project must have AEC approval. This will be dealt with in more detail in section 3.

Since 1990, DPaW has operated an Animal Ethics Committee (AEC) following the guidelines provided in the Code of Practice, which was supported by an Administrative Instruction endorsed by Corporate Executive in 1991.

The Code of Practice requires that an Animal Ethics Committee must have a membership that comprises at least four persons with a separate person appointed to each of the following categories:

- Category A: a person with qualifications in veterinary science, with experience relevant to the activities of the institution.
- Category B: a person with substantial recent experience in the use of animals in scientific activities.
- Category C: a person with demonstrable commitment to the welfare of animals, not employed by, or associated with the institution.
- Category D: an independent person who does not and has not previously conducted scientific activities using animals, and is not employed by the institution.

The DPaW Animal Ethics Committee is comprised of up to 12 members, which includes at least two representatives from each of the Categories above, the Executive Officer and Committee Chair.

The need for the committee has arisen from:

a) The higher profile of animal welfare issues in the community.

b) Legislation has now been passed in WA in the form of the Animal Welfare Act and Animal Welfare Regulations 2003.

c) The requirements of some external funding agencies for animal ethics approval before granting funds.

d) The requirement of some Australian scientific journals for animal ethics approval before accepting articles for publication.

3.1.2. Animals ethics approval

Before undertaking fauna research, survey, monitoring or animal handling which involve living, non-human vertebrate animals (excluding fish) DPaW staff must gain written Animal Ethics Committee approval.

Any project that does not target vertebrate species (i.e. invertebrate surveys, etc.) but has a risk of vertebrate by-catch must also seek Animal Ethics Committee approval before commencing.

The role of the DPaW Animal Ethics Committee is to ensure that all staff involved in the use of animals for scientific purposes act in a humane fashion and in accordance with the Code of Practice. Approval of projects is through the completion of an application form by the Chief Investigator in charge of the project.

The Animal Ethics Committee meets 5 times a year to review new applications and amendments to existing projects, and holds a special meeting in December to review annual reports. When planning research or experimental projects involving vertebrate animals, sufficient time needs to be allowed for the Animal Ethics Committee to consider the project at scheduled meetings. Applications are assessed by the Animal Ethics Committee using the principles of the three R's – *reduction, refinement* and *replacement*. The value of the work being proposed is also assessed by the committee, particularly if pain or suffering of animals is involved. Animal Ethics Committee approval is by consensus and any queries or concerns raised by members are returned to the applicant. It is important that sufficient supporting documentation is included in the application so that the Animal Ethics Committee can make an informed decision.

3.1.3. DPaW corporate licence to use animals for scientific purposes

DPaW staff undertaking approved activities must carry with them a copy of the corporate license to use animals for scientific purposes. Members of the public are entitled to see a copy of this license on site, on demand. Failure to show a license is a breach of license conditions and is subject to penalties under the Animal Welfare Act. You must also carry your Regulation 17 license at all times when carrying out hands-on activities involving animals, as part of license conditions under the Wildlife Conservation Act.

3.1.4. Animal ethics reporting

If you have Animal Ethics Committee approval, you are required to complete an annual report for the duration of the project (usually 3-year approval period), whether or not you actually used animals within the calendar year. These reports must provide a summary of progress and achievements, and details of any problems, animal injuries and deaths as well as actions that were, or could be, taken to avoid the same problems in future. Each approved project must submit an annual report. All annual reports are due in November each year for review at the December meeting (unless otherwise arranged). Failure to submit an annual report may result in the suspension of the project.

Chief Investigators must also immediately notify the committee if there are any unexpected deaths or other major incidents regarding animal welfare, advice of any amendments to the project before they are undertaken, and advise the Animal Ethics Committee when the project is completed. All reporting is a requirement under the Code of Practice and Chief Investigators who do not submit these reports face having their Animal Ethics Committee project suspended and their Regulation 17 license revoked.

3.1.5. Standard operating procedures

DPaW Animal Ethics Committee has developed a series of standard field operating procedures for routine research activities. This set of standard operating procedures has been developed to assist proponents prepare their applications and to ensure projects follow prescribed guidelines. This also helps the Animal Ethics Committee in efficiently assessing proposed projects. The Standard operating procedures, as well as Animal Ethics Committee application form, annual report form, legislation and general information about the Animal Ethics Committee are available to DPaW staff on the Department's intranet, Animal Ethics website: http://intranet/aec/default.aspx Standard operating procedures are available on the DPaW website at http://www.dpaw.wa.gov.au/plants-and-animals/monitoring/96-standards/99-standard-operating-procedures.

3.1.6. Animal ethics forms

Animal Ethics Committee Application Form

Information provided in the application must be sufficient to satisfy the Animal Ethics Committee that the proposed use of animals is justified. All background documentation relating to this project, e.g. translocation proposal, management plan, etc. should be provided with the application when submitted. Allow adequate time for all project approvals required in your project planning timeframe.

Animal Ethics Committee Renewal Form

Animal Ethics Committee projects are approved for a maximum of 3 years. As the date for expiry of the project approval approaches, a project renewal application must be lodged and approved before the project can continue. If the project renewal is not approved by the expiry date of the current project then no work may be undertaken until renewal approval is given. The renewal application must contain a summary of the animals used over the previous 3 year period and justification as to why the project needs to continue.

Animal Ethics Committee Amendment Form

All changes to the protocol, including animal numbers or type of use, must be submitted to the Animal Ethics Committee for approval. If you only require the addition or removal of personnel and/or locations, please contact the Executive Officer directly (amendment form not necessary). Failure to seek Animal Ethics Committee approval prior to making changes may result in the suspension of the project.

DAFWA Fieldwork Notification Form

Notification of timing and location of all fieldwork activities for Animal Ethics Committee approved projects must be submitted to the Department of Agriculture and Food WA as part of DPaW's licence conditions. From June 2012 DAFWA has instituted a proforma "Notification Form - Licence to Use Animals for Scientific Purposes" to be used. This should be submitted by the Chief Investigator to the Executive Officer for inclusion in the project file. The EO will forward the notification on to the DAFWA contact

Translocation Proposal Template

A translocation proposal template has been developed to assist Chief Investigators who are submitting a translocation project for Animal Ethics Committee assessment. Please note that approval of this TP is required before final Animal Ethics Committee approval will be granted.

Unexpected Animal Death or Emergency Euthanasia

The Animal Ethics Committee must be notified as soon as possible of the event if any animal becomes terminally ill or dies unexpectedly. It is a requirement that you arrange a Post Mortem of the animal, unless you receive special exemption from the Animal Ethics Committee.

Competency Checklist

This form must be completed for all personnel involved in the project. Competencies must also be updated regularly as the skills and proposed animal use of participants change. The purpose of this is to demonstrate that you have the skills and experience to undertake the proposed procedures. It is advisable to keep a log or a diary of you fauna related experience to support this.

All forms must be signed and a copy sent via email to the Executive Officer animalethics@dpaw.wa.gov.au.

For further information contact Department’s Animal Ethics Committee Executive Officer on (08) 9334 0438 or via email, animalethics@dpaw.wa.gov.au or visit the intranet site http://intranet/aec/default.aspx

3.2. Hygiene and fauna handling techniques

3.2.1. Animal handling ethics

All procedures involving animals must be done in ways that will avoid or reduce the risk of injury, suffering or unintentional death to animals. The Chief Investigator for approved projects must report to the Animal Ethics Committee on any incidents where animals experience injury or suffering or where animals die unintentionally. The report should detail the circumstances of the incidents and any action that was taken or could be taken to avoid or reduce the problem in future. The Animal Ethics Committee may approve the remedial action or make further recommendations, but most importantly it can incorporate appropriate changes to standard procedures and make other users of the procedures aware of changes arising from real or potential problems, thus promoting best practice. Personal liability may only be an issue where individuals have used non-approved procedures or non-approved variations on procedures.

Ethical codes of practice apply to feral animals as well as native animals.

3.2.2. Hygiene and safety (SOP 16.2)

Anyone involved in animal trapping and handling should refer to the DPaW Animal Ethics Committee SOP 16.2 *Managing Disease Risk in Wildlife Management*

- Diseases can be transferred between animal populations and humans via contaminated traps, holding bags, waders, boots, nets and temporary housing/caging.
- Most animals bite and scratch in self-defence and there is risk of serious injury to both animal and handler. Animals must be handled with care by experienced persons or under the supervision of an experienced person.
- Always have ready access to a first aid kit.
- Treat scratches and bites immediately as there is risk of infection or disease.
- Handle animals in appropriate bags, to reduce stress on animal and reduce risk of injury to the handler.
- Maintain effective hygiene to reduce the risk of disease transmission
- wash hands thoroughly after handling animals
- use clean equipment and clean it after use
- Clean hands and equipment between handling animals if there is a risk of transmission.
- Bats have the potential to carry Lyssavirus, a genus of viruses closely related to the Rabies virus.

It is essential that anyone handling bats is vaccinated against Rabies.

Points on trap maintenance and hygiene

- Traps must be kept clean and maintained in good working order
- Elliott traps should be cleaned after use by removing the pin and unfolding the trap to expose the interior, scrub clean and then reassemble
- Cage traps may need washing with high pressure hose to remove animal waste
- Trap mechanism may need adjustment on some traps
- Movement of dirty hessian bags and traps from working sites to other isolated sites may pose a disease risk for animal populations in those isolated sites – better to use one batch of hessian

for each site or connected group of sites, or wash/soak hessian bags in bleach solution and dry in the sun before using at another site.

3.2.3. Handling techniques of vertebrate fauna (SOP 10.1, 10.2)

Equipment

Personnel undertaking trapping should be equipped with a trapping field kit, comprising a plastic fishing tackle box or plastic tool box (plastic makes less noise from rattling contents) containing a minimum of the following items:

- Spring balances (5kg, 2kg, 500g, 100g and/or 50g);
- Dial callipers;
- Surgical scissors;
- Ear punch (x 2);
- Ear tags;
- Ear tag applicator;

- Folding eyeglass 10x magnification
- Fixomull Stretch Tape (for areas where there are woylies and bandicoots which eject their pouch young);
- Plastic rule (30cm);
- Betadine®;
- Ethanol (70%);
- Assortment of calico bags.

For advice on where these items can be sourced please contact Western Shield Zoologist.

Removing animals from traps

- Always check inside and around traps for ejected pouch young or young at foot.
- Always carry the appropriate handling bag to the trap.
- Only the handler carrying the handling bag should approach cage traps.
- Approach trap quickly and quietly.

Cage traps

Small cage traps**:** *medium size mammals*

- Open the handling bag and place over the door end of the trap, lift the trap door and hold in the open position while securing the bag around the sides of the trap.
- Gently encourage the animal into the bag. Usually this can be done by blowing puffs of air at the animal.
- Remove animal from the cage as quickly and efficiently as possible and secure it in the handling bag.

Large cage traps, Bromilow and Thomas traps: *wallabies, hare-wallabies, Quokka*

- Position the trap so the door is facing skywards, open the door.

- Place a bag over the animal, grip the animal by the base of the tail and lift it from the trap. It may be helpful to take a firm grip of the hind feet as you lift the animal from the trap to restrain the animal and avoid injuries.
- Immediately place animal in hessian bag and secure the opening.
- With Thomas and Bromilow traps there can be difficulties with animals gripping the mesh and making it difficult to lift them clear of the trap. Care must be taken not to injure the animal by pulling against the animal's grip.
- If entanglements occur, cut the mesh carefully around the entanglement to free the animal.

Elliott Traps

- Check what kind of animal is in the trap
- If safe put hand in the trap and remove the animal from the trap
- Or place a calico bag over the end of the trap and gently slide the animal into the bag
- If the animal is difficult to remove, place the trap at the base of the handling bag, pull the trap release pin and open the Elliott trap into the bag.

Pit Traps

- Check the type of the animals in the trap
- Use a stick to scratch through material in the bottom to check for small reptiles and frogs as well as centipedes, scorpions or spiders.
- If safe place hand in trap and remove animal/s

When weighing animals in wet conditions, reweigh handling bags often, particularly hessian bags, as the weight can change significantly depending on how wet the bag is.

General handling

All animals are to be handled in a way that minimises stress and the risk of injury or stress-induced disease. Stress and risk of injury can be minimised by:

- using appropriate handling bags
  - heavy jute bags for medium sized animals, calico bags for smaller animals
  - material with dense weave so that the animals can't see through bag
- keeping animals in the bag when handling as much as possible so that eyes are covered
- processing animals as quickly and efficiently as possible
  - be gentle but firm
- avoiding loud or sudden noises – soft talking is OK
- avoid shuffling feet or stepping on twigs
- avoid using wet handling bags
- not smoking or wearing strong smelling chemicals such as insect repellents when handling animals

Release of animals

Animals should always be released at the site of capture unless an alternative site has been justified and approved. Animals should be released immediately after the appropriate measurements and data have been recorded, or at a time when they would normally be active, with all reasonable care taken to avoid injury and predation, particularly from raptors if released during daylight hours.

3.2.4. Marking and measurements (SOP 12.1, 12.2, 12.3, 12.5, 12.9)

Marking

Animals can be marked for different purposes. Unique markings can be used to mark animals to enable the identification of individuals. Simpler markings can be used to identify animals as having been captured. Permanent makings such as ear notching and toe clipping are not warranted for short-lived species if the populations are only monitored once each year. For longer-lived species or for populations that are monitored more intensively such marking techniques may be justified. Often ink or die marks will last long enough to meet study aims.

Marking to indicate captures

Ear notches can be used for the first capture of an animal and non-toxic ink or die to mark animals recaptured in the same trapping period. This form of marking is usually sufficient for estimating population size or density.

Marking for individual identification

Ear tags

All ear tags should be ordered through the WS Zoologist to reduce the chance of duplication of number series.

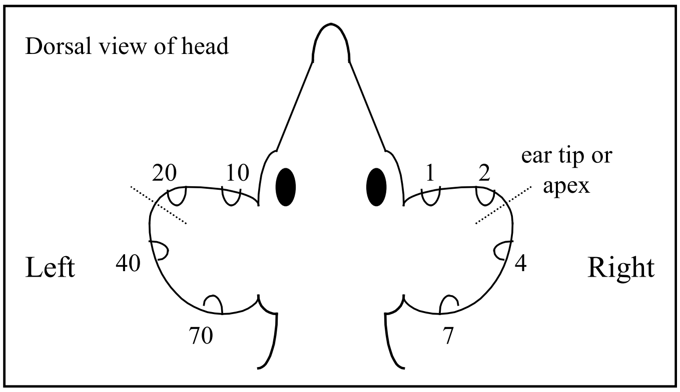
- Ear tags are ordered from the manufacturer in number series with a 1-2 letter identifier so that there is no duplication of ear tag numbers with the same identifier.
- Usually, two tags are applied to an animal so that if one is lost then the other may still be attached to enable identification of the individual.
- Ear tags must be applied so that they are positioned close to the head, without crushing any of the intricate structures near the ear opening and they sit flush with the margin of the ear.
- Ear tags must **not** be used on phascogales and smaller dasyurids or on rodents.

Microchip implants (or Passive Implant transponders PITs)

- Microchips are the size of a large grain of rice and are inserted just beneath the skin.
- Contain a passive transponder with a unique code. Common brands include Allflex and Trovan.
- The code is read using a hand held scanner. Note: some scanners will only read one brand of PIT – if multiple brands of PIT are being used in an area/district/region, a scanner that reads multiple brands should be used.

Ear notching and toe clipping

- Small mammals (dasyurids and rodents) can be ear notched using pre-designated positions on the ears. The numbering system below is often used.
- Numbers used must be crossed off on a number recording sheet to avoid duplication.
- Ears and toes can be naturally damaged and notched which can confuse identification of recaptured individuals.
- Both ear notching and toe-clipping involve removal of body tissue and should only be used where identification of individuals is vital.
- Removed tissue can be retained for genetic studies.



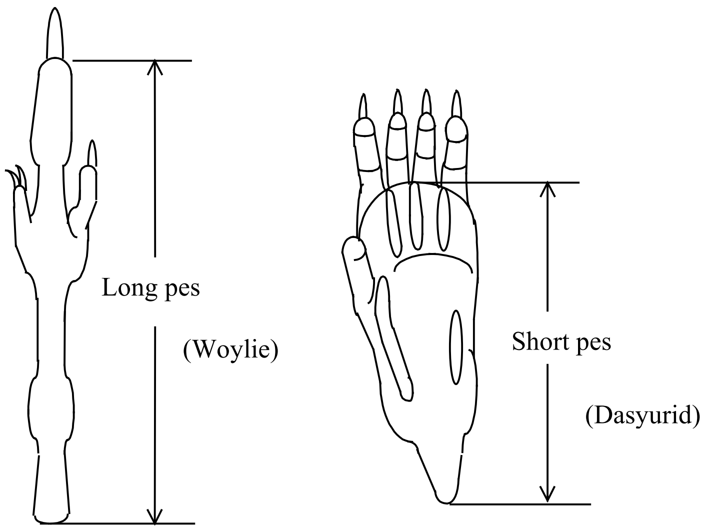
Ear notch numbering system

Ear notching and tagging equipment must be kept clean. After positioning in applicator ear tags should be dipped in alcohol before application. Ear punches should be cleaned regularly with alcohol.

For Western Shield fauna monitoring, all medium size mammals should be marked or ear tagged for individual identification. Phascogales, Mardos, dunnarts, bush rats and other rodents can be ear notched for identification as recaptures (R) and marked with dye/ink to indicate recapture during current trapping period (RT).

Measurements

- Measurements usually taken on mammals include body weight, head length and pes.
- Pes measurements are differentiated into short and long pes (see diagram below). Short pes measurements are taken on possums and dasyurids, and long pes measurements are taken on bandicoots and macropods.
- Measurements usually taken on reptiles and frogs include total length (reptiles only), snout-vent length, leg (tibia) length and head width.
- Tail length is sometimes diagnostic in small dasyurids and rodents and should be measured if this is so.
- The sex of individual animals is generally obvious in marsupials but may be difficult to determine in sexually immature rodents. Generally, male rodents can be distinguished from females by having a larger and often pigmented gap between the anus and the genital papilla. The determination of sex in reptiles and frogs is not practical or required for general survey and monitoring purposes.



Pes measurements

3.2.5. Euthanased animals (SOP 15.1)

The Animal Ethics Committee SOPs for euthanasing wildlife should be referred to and a euthanasia plan must be developed before any monitoring or survey takes place.

Ethical codes of practice apply to feral animals as well as native animals.

All fauna specimens have scientific value. Euthanased animals are no exception and the WA Musuem should be consulted to determine if a specimen should be kept as a reference specimen or taxidermied display (refer section 7 for WA museum collection procedures http://museum.wa.gov.au/research/development-service/collection-management).

To assist with the decision of whether the humane killing of a native animal is necessary, generalise guidance criteria are provided below (Figure 1).

Note: non-native pest species must not be released back into the wild. For further information go to contact the nearest office of the Department of Agriculture or telephone, free call 1800 084 881 for advice regarding the appropriate fate of vertebrate animal pests.

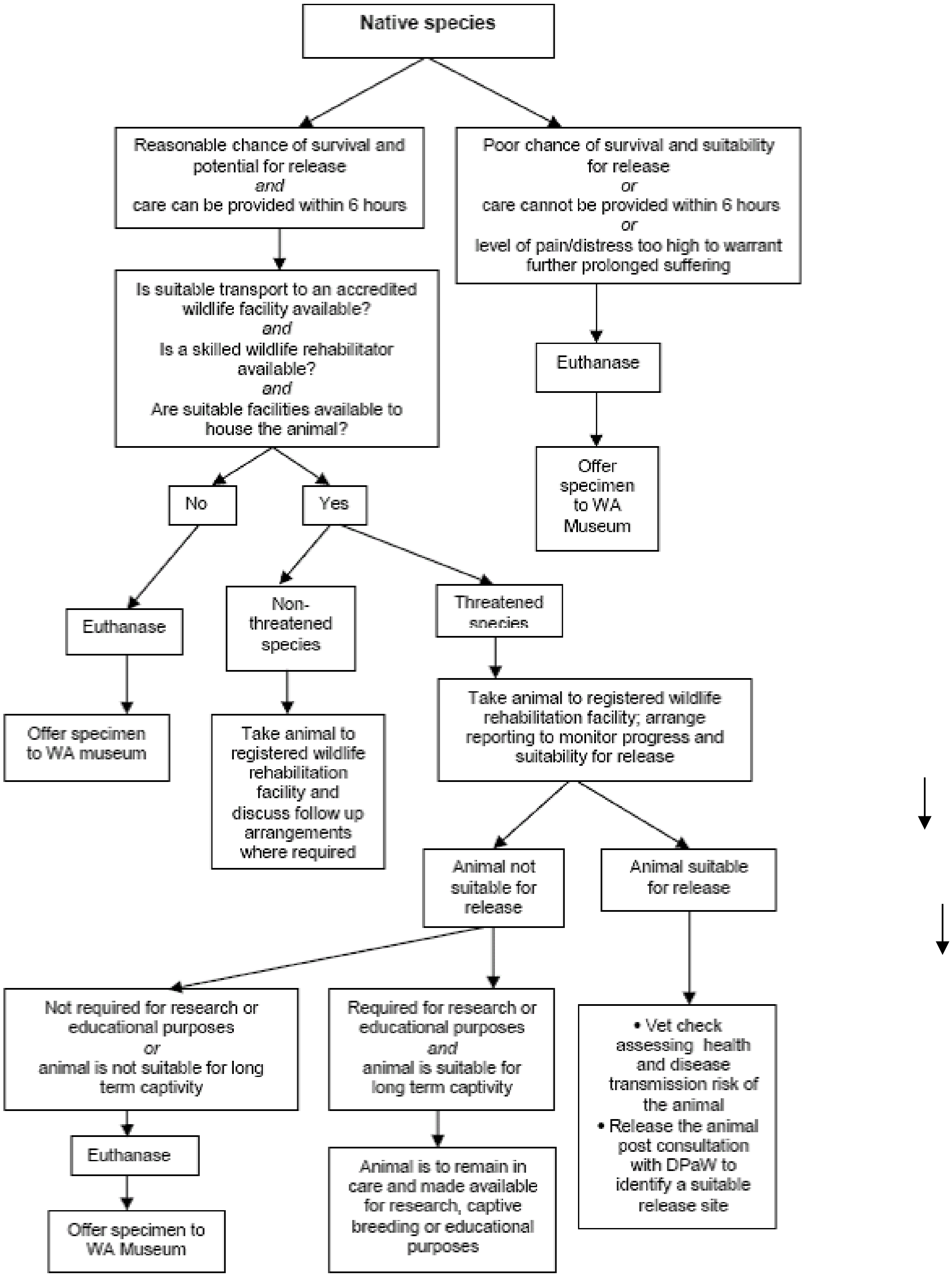


Figure 1. Decision making chart for sick, injured or orphaned fauna (from SOP 15.1)

4. Resources

For questions about…	…	at…	on….
Animal ethics	e	DPaW Kensington	9219 9515
	cs fficer	DPaW Kensington	9334 0438
Butterflies	Matt Williams	DPaW Kensington	9219 9040
Cat baiting	Dave Algar	DPaW Woodvale	9405 5145
	Paul Blechynden	DPaW Narrogin	9881 9213
Chuditch	Keith Morris	DPaW Woodvale	9405 5159
Dibbler	Tony Friend	DPaW Albany	9842 4523
Earthworms	Allan Wills	DPaW Kensington	9334 0305
Fauna management - General	Manda Page	DPaW Kensington	9219 9515
	Juanita Renwick	DPaW Kensington	9219 9515
FaunaFile database	Juanita Renwick	DPaW Kensington	9219 9515
Fox baiting on DPaW-managed lands and 1080	Dennis Rafferty	DPaW Bunbury	9725 5987
	Ashley Millar	DPaW Kensington	9334 0261
Gilbert's Potoroo	Tony Friend	DPaW Albany	9842 4523
Heath Mouse	Keith Morris	DPaW Woodvale	9405 5159
Lancelin Island Skink	Dave Pearson	DPaW Woodvale	9405 5112
Land for Wildlife	Penny Hussey	DPaW Kensington	9334 0530
Licensing - Aviculture	Graeme Zekulich	DPaW Kensington	9219 9835
Licensing - General	Pauline Goodreid	DPaW Kensington	9219 9830
	Danny Stefoni	DPaW Kensington	9219 9833
Licensing - Kangaroo shooters	Tony Bennell	DPaW Kensington	9219 9164
Licensing - Reptile (Pet Herpetofauna)	Adrian Coleman	DPaW Kensington	9219 9834
Lodging specimens - Frogs and Reptiles	Paul Doughty	WA Museum	9212 3700
Lodging specimens - Mammals		WA Museum	9212 3700
Lodging specimens - Birds	Ron Johnstone	WA Museum	9212 3700
Turtles - Marine	Scott Whiting	DPaW Kensington	9219 9752
Nominating fauna for listing as threatened	Species & Com	DPaW Kensington	92199511
Numbat	Tony Friend	DPaW Albany	9842 4523
Project Eden	DPaW Denham	DPaW Denham	9948 1208

For questions about…	…	at…	on….
Pythons	n	DPaW Woodvale	9405 5112
Quokkas - General		DPaW Manjimup	9771 7933
Quokkas - Survey	delow	DPaW Manjimup	9771 7981
Radiotelemetry	Neil Thomas	DPaW Woodvale	9405 5119
Red-tailed Phascogale	Tony Friend	DPaW Albany	9842 4523
Return to Dryandra	DPaW Great Southern District	DPaW Narrogin	9881 9222
Barna Mia	DPaW Great Southern District	DPaW Narrogin	9881 9222
Rock-wallabies	Dave Pearson	DPaW Woodvale	9405 5112
Rodent conservation	Keith Morris	DPaW Woodvale	9405 5159
Translocation proposal, fauna - Preparation	Manda Page	DPaW Kensington	9219 9515
	Juanita Renwick	DPaW Kensington	9334 0268
Western Barred Bandicoot	Tony Friend	DPaW Albany	9842 4523
Western Ringtail Possum	Kim Williams	DPaW Bunbury	9725 5910
	Adrian Wayne	DPaW Manjimup	9771 7992
Western Shield – Equipment (Eartag) Suppliers	Michelle Drew	DPaW Kensington	9334 0261
Western Shield - Monitoring	Michelle Drew	DPaW Kensington	9334 0261
Western Shield - General	Ashley Millar	DPaW Kensington	9334 0261

If you are unsure on who to contact for help, please contact Juanita Renwick (9219 8709 juanita.renwick@dpaw.wa.gov.au) or Manda Page (9219 9515 manda.page@dpaw.wa.gov.au) and they'll direct you to the best person.

Current as at October, 2014

Recovery Team Contacts

Recovery Team	Chair	Phone
Carnaby's Black-Cockatoo	Dave Mitchell	9474 7036
Forest Black-cockatoos	Brad Barton	9771 7933
Western Swamp Tortoise	Craig Olejnik	9303 7705
Lancelin Island Skink	Dave Pearson	9405 5112
Geocrinias	Kim Williams	9725 5910
Western Ringtail Possum		
Numbat	Tony Friend	9842 4523
Chuditch	Keith Morris	9405 5159
Dibbler	Tony Friend	9842 4500
Gilbert's Potoroo	Sarah Comer	9842 4513
Shark Bay Marsupials (BB, WBB, BHW)	Manda Page	9219 9515
Hairy Marron	Rodney Daffy (Dept of Fisheries)	9842 7333
South Coast Threatened Invertebrates	Deon Utber	9842 4514
South Coast Threatened Birds (Western Ground Parrot, Noisy Scrub Bird, Western Bristlebird)	Sarah Comer	9842 4513
Quokka	Brad Barton	9771 7933
Woylie	Manda Page	9219 9515
Rock wallabies	Dave Pearson	9405 5112

Fauna identification references

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Strahan, R (ed.) 1983. The Australian Museum Complete book of Australian Mammals. Angus & Robertson, London.

Tyler, M.J., Smith, L.A. & Johnstone, RE. 1984. Frogs of Western Australia. W.A. Museum. Perth.

Valente, T. & Wooley, P.A. 1982. Hair structure of some Western Australian mammals. J. Proc.Roy.Soc. W.A. 64:101-32.

Useful websites

Animal ethics forms and info

DPaW internal: http://intranet/aec/default.aspx

External SOPs: http://www.dpaw.wa.gov.au/plants-and-animals/96-monitoring/standards/99-standard-operating-procedures

Animal welfare legislation: http://intranet/aec/Pages/Legislation.aspx

Translocation proposal template: http://intranet/aec/Documents/DPAW%20Translocation%20Proposal%20Template%202014.doc

Animal sound recordings
http://www.naturesound.com.au/

Atlas of Living Australia
https://www.ala.org.au/

Bird migration and survey
http://www.abc.net.au/science/birds/

Birdlife Australia (local ornithologists)
http://www.birdlife.org.au/

Commonwealth Department of the Environment

Listed threatened species: http://www.environment.gov.au/biodiversity/threatened/index.html

DPaW

DPaW Library database search: http://intranet/icl/default.aspx

DPaW scientists' specialties: http://www.dpaw.wa.gov.au/about-us/science-and-research

DPaW Wildlife Licensing Branch: http://www.dpaw.wa.gov.au/plants-and-animals/licences-and-permits/134-fauna-forms?showall=&start=2

Designing a monitoring project for significant native fauna species: http://www.dpaw.wa.gov.au/plants-and-animals/monitoring/97-monitoring-programs

Living With Wildlife and Fauna Notes: http://www.dpaw.wa.gov.au/plants-and-animals/animals/living-with-wildlife

NatureMap: http://naturemap.dpaw.wa.gov.au/default.aspx

Policy Statements: http://www.dpaw.wa.gov.au/about-us/36-policies-and-legislation

Recovery Plans and Interim Recovery Plans: http://www.dpaw.wa.gov.au/plants-and-animals/threatened-species-and-communities/197-approved-recovery-plans

Science Project Plan and guidelines: http://intranet/science/Documents/Forms/Staff%20Guidelines.aspx

Threatened and Priority fauna rankings: http://www.dpaw.wa.gov.au/plants-and-animals/threatened-species-and-communities/threatened-animals

Western Shield: http://www.dpaw.wa.gov.au/management/pests-diseases/westernshield

Ferals, Pests and Diseases

DPaW: http://www.dpaw.wa.gov.au/management/pests-diseases

Commonwealth feral vertebrate species: http://www.environment.gov.au/biodiversity/invasive/ferals

International Union for Conservation of Nature

IUCN Red List Guidelines: http://www.iucnredlist.org/documents/RedListGuidelines.pdf

IUCN category definitions: http://www.iucnredlist.org/technical-documents/categories-and-criteria/2001-categories-criteria

Legislation

Australian government: http://www.comlaw.gov.au

WA state Law Publisher: http://www.slp.wa.gov.au/Index.html

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WA Museum

WAM: http://museum.wa.gov.au/

WAM collection procedures http://museum.wa.gov.au/research/development-service/collection-management

WAM Frog watch: http://museum.wa.gov.au/explore/frogwatch

WAM vertebrate checklist: http://museum.wa.gov.au/research/departments/terrestrial-zoology/checklist-terrestrial-vertebrate-fauna-western-australia

Order field guides from: http://www.museum.wa.gov.au/store/museum-books/#museum-books/all