General Guidelines
for
Wildlife Capture & Handling

Prepared by

The **Centre for Environmental Management** (CEM)

School of Applied & Biomedical Science

Federation University Australia

DOCUMENT STATUS AND AIMS

The primary purpose of the document is to provide guidance to staff and students intending to undertake animal trapping and handling on appropriate methods and relevant permits and ethics requirements. When compiling AEC applications, this document should provide the detail of currently accepted practices and techniques. The intention is for the document to be periodically updated to reflect the decisions of the AEC, and to minimise the amount of 'going over old ground' when the AEC processes applications.

The document focuses on field-based wildlife studies, and does not cover experimental laboratory studies of wildlife or other animals. It covers techniques commonly used in wildlife inventory studies, such as Elliot trapping, cage trapping, pitfall trapping and spotlighting, as well as more recently developed research techniques including radio tracking, PIT tagging and the use of global positioning system (GPS) collars.

Some of the techniques described have not been previously used by the Federation University Australia researchers, and may never be used, or only used in special cases. These techniques have been included to provide coverage of a relatively complete range of wildlife trapping and handling techniques.

This document and the documents listed on the AEC application form must be given to all students and staff prior to their undertaking fieldwork. Students and staff are required to read these carefully before conducting fieldwork, and must obtain clarification from supervisory staff if they are uncertain of the meanings or implications of any point. Failure to complete forms correctly and to adhere to the conditions set out in these documents may prevent you from commencing your work in time or attract repercussions from authorities / agencies.

This document is adapted from the draft set of guidelines developed for internal use by Arthur Rylah institute for Environmental Research, Department of Environment, Land, Water and Planning (DELW&P - formally DEPI) and a draft 'protocol for standard fauna surveys' compiled by School of Science and Engineering staff, Ballarat University in 2002.

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1. INTRODUCTION

These guidelines have been written to be used as a best practice document and as a way of streamlining the animal ethics application process by providing standardised information to the Federation University Australia, Animal Ethics Committee (AEC).

All research conducted on native fauna by staff and students is to be undertaken in accordance with research permits issued under the *Wildlife Act 1975* and *National Parks Act 1975,* which require that such research is conducted in accordance with the Australian Code of Practice for the Care and Use of Animals for Scientific Purposes 2013 (the Code). This means that approval from the AEC is required for all scientific procedures that are undertaken on vertebrate fauna.

Researchers should pay particular attention to the wildlife section of the Code. The following principles are extracted from the Code and outline the general responsibilities and justifications for conducting animal research.

1. Scientific and teaching activities using animals may be performed only when essential to:
   - Obtain and establish significant information relevant to the understanding of humans or animals;
   - The maintenance and improvement of human or animal health and welfare;
   - The improvement of animal management or production; or
   - The achievement of educational objectives;
   - The achievement of environmental objectives.
2. Studies using animals may be performed only after a decision has been made that they are justified, weighing the scientific or educational value of the study against the potential effects on the welfare of the animals.
3. People who use animals for scientific purposes have an obligation to treat them with respect and consider their welfare as an essential factor when planning and conducting studies.
4. The acquisition, care and use of animals for all scientific purposes in Australia must be in accord with the Australian Code of Practice and with Commonwealth, State and Territory legislation.
5. Investigators have direct responsibility for all matters relating to the welfare of the animals they use.
6. Institutions are required to establish an animal ethics committee to ensure that all animal use conforms to the standards of the Australian Code of Practice.
7. Investigators must submit written proposals for all animal studies to the ethics committee which must take into account in the standard project application, the expected value of the knowledge to be gained, the justification for the study, and all ethical and animal welfare aspects.
8. Scientific and teaching activities must not commence until written approval has been given by the animal ethics committee.

AEC approval must be sought and obtained for all activities that involve the use of vertebrates. Activities that involve the use of invertebrates will not at this stage be subject to the formal approval of the AEC. However, such activities must still abide by the principles outlined in the Code.

Encapsulated in these principles is the need in scientific and teaching activities to consider:

- the ***replacement*** of animals with other methods;
- the ***reduction*** in the number of animals used; and
- the ***refinement*** of techniques used to reduce the impact on animals.

The Principal Investigator of each project is responsible for seeking AEC approval and ensuring compliance. If working in collaboration with other organisations, the Principal Investigator will need to obtain Animal Ethics Approval prior to work commencing, regardless of whether collaborating organisations are covered by Animal Ethics Approval from their own organisation.

1.1 Permit requirements

A valid research permit is required to live-capture and release small mammals (including bats), birds, reptiles and amphibians. All staff and students conducting research are required to carry a copy of the permit when in the field and have knowledge of the contents of the Code as required under the permit.

Research permits are issued by State Government departments. Researchers working in Victoria are required to hold a current permit issued by the Department of Environment, Land, Water and Planning

(DELW&P) under the *Wildlife Act 1975. An additional permit issued by Parks Victoria is required to undertake research in parks and reserves listed under the National Parks Act 1975 (Vic).* New South Wales research permits are managed by the National Parks and Wildlife Service, Department of Environment and Conservation.

Permits clearly indicate the requirement for researchers to gain permission from land owners/managers (e.g. Parks Victoria, Forests, private landowners) prior to commencing work.

Permits are granted with a set of restrictions and conditions that researchers must adhere to.

You are required to give five days notice to the local DELW&P office prior to visiting a site to undertake fauna survey work. A similar period of notice should be given to Parks Victoria, NSW Parks and Wildlife Service and other land managing authorities or property owners. Include in the notification the details of the location, date, time, vehicle registration number and proposed activities.

1.2 Borrowing of equipment

School of Applied & Biomedical Science

The School of Applied & Biomedical Science has a large store of wildlife research equipment, including Elliot traps, cage traps (several types), harp traps, hair tubes, pitfall traps and spotlights. Vertebrate trapping equipment will only be lent to researchers with a current research permit and AEC approval for the work they are undertaking. All traps must be labelled with the appropriate research permit number. Upon returning equipment, researchers must submit a trapping record, detailing the location of trapping sites and the species captured.

The equipment request form and trapping report form are provided in Appendix 1.

1.3 Reporting

There are several reporting requirements for researchers undertaking wildlife studies:

- It is a condition of Wildlife Research permits to submit the details of all captures to the appropriate state government department. Records of captures within Victoria must be submitted to the Department of Environment, Land, Water and Planning. The information is added to the Atlas of Victorian Wildlife, which is an important repository of wildlife distribution data within Victoria.
- Researchers must submit annual and final reports to the AEC, detailing trapping activity, captures and ethical issues.

The AEC must submit annual and final reports to the University, and the University must report to Government departments regarding the operation of the AEC, research activity and animal welfare issues.

School of Biomedical Science

The purpose of the trapping record form (Appendix 1) is to prompt researchers within the School of Biomedical Science to provide the necessary information to satisfy these reporting requirements. While researchers are each individually responsible for reporting to the AEC and the Department of Land, Water and Planning (Vic), the information supplied to the School of Biomedical Science on the trapping record form can form the basis of these reports.

Data submitted on the trapping record forms are entered into a Microsoft Access database managed by the school of science technical support group, and the database can produce formatted reports suitable for inclusion in an AEC report or a report to the DELW&P.

2. OBSERVATION AND TRAPPING TECHNIQUES

2.1 General considerations

Skills

All staff and students undertaking independent wildlife research must have demonstrated animal handling skills, identification skills and trapping experience. Knowledge of target species' ecology and behaviour is also important. Researchers require approval from the AEC prior to undertaking any trapping program.

Inexperienced students and staff may also participate in animal trapping and handling, but they must work under the direct supervision of experienced teachers or researchers.

Bait

There is a range of different baits used for attracting animals into traps. 'Standard' bait for ground-dwelling mammals is a mixture of golden-syrup, rolled oats and peanut butter. A variety of nuts are also used. Imitation pistachio essence may be added to attract fungus-feeding mammals such as bandicoots and potoroos. To target carnivores use chicken or fish, or a mixture of sardine, flour and tuna oil. These baits may also be useful for some lizards.

Selection and management of traps

The type of trap should be appropriate to the target species. Ensure that all traps are checked to be in good working order prior to use and that traps are secured to reduce the chance of a trapped animal rolling the trap. Use flagging tape or some other clear indicator of trap positions so they can be readily located, minimising the time spent by animals in the traps. Exercise caution in regard to placement of flagging tape near public areas so that undue attention is not drawn to the trapping site. Traps and flagging tape should be numbered and laid out sequentially at regular intervals, along recorded compass bearings in either single transect or grid patterns. The spacing between traps should be such that the next trap (or flagging tape) is clearly visible from the previous position. Suggested spacings include 5-10m for areas with dense understorey (eg. open forest, heathlands) and 10-20m in more open habitats (eg. grasslands and woodlands). If different kinds of traps are mixed within a trap site, such as Elliott traps, cage traps and hair tubes, consideration should be given to establishing a regular site layout and the arrangement of traps should be carefully recorded.

The start and end points of trapping lines should be recorded using a Global Positioning System (GPS), both to aid in re-finding the traps and meeting reporting requirements.

Ensure that all traps are located when checking and at the end of the survey, they are all removed or closed and locked. Traps must be thoroughly cleaned between trapping periods.

Timing and duration of trapping studies

Consideration must be taken when trapping during breeding seasons to minimise stress to animals. If possible, breading seasons should be avoided.

The AEC have expressed concern that individuals may spend several consecutive nights in traps (recaptured animals). It is usually necessary to trap over several nights, as many species (particularly small terrestrial mammals) need time to become accustomed to the presence of traps before being bold enough to enter the trap and be captured. Generally trapping should be limited to three consecutive nights at each site. If the study does not require animals to be marked, it may be desirable to mark individuals by painting a spot of fur or skin using a non-toxic paint. This would provide information on the short-term recapture rate, which should be presented to the AEC in annual and final project reports.

Protection of trapped animals from adverse weather conditions

Researchers must also be aware of potential adverse weather conditions, particularly frost, snow or heavy rain, and be prepared to suspend trapping by closing or removing traps. Traps should be located where they provide some protection for captured animals (eg. in dense shrubbery, or within grass tussocks), away from public view. Protection from rain and wind should be provided for animals held in cage traps, potentially by using plastic covers (see section 2.11). It is recommended to use insulating material inside mammal traps, and in the bottom of pitfall traps. Ensure that the material, such as sawdust, shredded bark or leaf litter, does not interfere with the trap mechanism or hold human scent.

Traps should be closed at the beginning of any day where the temperature is forecast to exceed 30°C, or if the day is declared a day of total fire ban within the region. Traps can be reopened in the evening, provided the temperature drops below 30°C. During cold weather trapping must be cancelled in the minimum temperature is forecast to drop below 0°C, or if heavy rain and/or high winds are expected.

Checking traps

For nocturnal animals the traps must be checked as soon as practicable after sunrise to reduce stress and risk of exposure to extremes of temperature and predation, both in the trap and on release. For diurnal animals, the traps should be checked once in the morning and once in the evening before sunset. Permits clearly indicate the need to check traps or nets at intervals not exceeding 18 hours.

General handling procedures

When handling animals, keep noise and movements to a minimum. Remove animal from trap as soon as practical and place in a cloth bag (except frogs, use plastic) and keep it securely closed. Try and keep the animal's eyes covered as much as possible as this can reduce movement and stress. The material strength and size of the bag used should be appropriate for the species, such that the animal is easy to control but cannot easily escape.

2.2 Bird observing

Target groups:	Diurnal birds
Ethics approval required:	No
Research permit required:	No

Do not cause stress to the birds or expose them to danger by approaching too closely or interfering with their natural behaviour. Try not to put resting birds to flight and keep habitat disturbance to the minimum, especially near nests, eggs, display areas and roost sites. Approach nests carefully and do not stay nearby for long periods as this may attract predators to eggs or young. Continued observer presence may also drive parents away from young. Visits to nests should be as brief as possible, and preferably timed to coincide with periods when adult birds are absent. The Birds Australia code of practice for the Nest Record Scheme should be consulted for recommendations on minimising researcher impact.

Do not harass rare or vagrant birds by long periods of intense observation as they may be in strange territory and vulnerable to exhaustion and predation.

2.3 Spotlighting

Target groups:	Nocturnal arboreal and terrestrial mammals, birds, frogs
Ethics approval required:	Yes
Research permit required:	No

At all times avoid overexposing animals to the light and keep noise to a minimum. Observers should keep eyes as close as possible behind the light source to aid in detecting the reflected eyeshine of an animal. Poor weather conditions, such as high winds and wet conditions can impede detection of an animal's eyeshine and movement.

2.4 Opportunistic observation

Target groups:	Mammals, birds, herpetofauna, fish
Ethics approval required:	No (yes if animals are handled)
Research permit required:	No (yes if animals are handled)

Researchers generally note species observed opportunistically. Opportunistic observation is often the only way that many species are recorded in inventory studies, particularly species such as macropods, wombats, koalas, platypus, echidnas, snakes and large skinks. The presence of the animal should be recorded, and the animal allowed to continue normal activity with minimal disturbance from the observer. If it is necessary to capture and handle animals for positive identification, ethics approval and a research permit is required.

2.5 Observation of scats, tracks and signs

Target groups:	Mammals, birds, some reptiles
Ethics approval required:	No
Research permit required:	No

Animal scats, tracks, burrows and other signs such as diggings and tree scratch marks are often distinctive and can be used to indicate the presence of species not detected by other methods. A useful reference text for Australian mammals is 'Tracks, Scats and Other Traces - A Field Guide to Australian Mammals' by Barbara Triggs. Predator scats may also contain identifiable hair and bone fragments of prey species (see below).

2.6 Predator scat collection

Target groups:	Mammals, birds, herpetofauna
Ethics approval required:	No
Research permit required:	No

Predator scats and owl pellets potentially contain hair and bone fragments of native and introduced prey species. Experience is required to positively identify prey species from these samples, and professional identification services are offered by some organisations and individuals (in particular Barbara Triggs).

Scats should be collected in paper bags, clearly labelled with the location and date. Preferably, scats should be collected without touching the hand, and hands should be thoroughly washed and disinfected. If the scats are to be processed by an outside organisation, it may be possible to partially process (eg. washing and drying) the scats at UB to reduce the cost.

2.7 Call playback

Target groups:	Owls, arboreal mammals, cryptic bird species, frogs
Ethics approval required:	Yes
Research permit required:	No

Researchers must be capable of aurally identifying bird, mammal or other species that are likely to respond to the call tape. The volume of the playback will depend upon the quality of the recording, weather and habitat condition and should be adjusted accordingly. Generally, the volume should not be more than approximately 20% above natural volume. Avoid excessive use of playback at single sites, as repeated playback can cause significant disturbance to resident birds.

2.8 Nest boxes

Target groups:	Arboreal mammals, hollow nesting birds, microbats
Ethics approval required:	No (Yes if animals are to be handled)
Research permit required:	No (Yes if animals are to be handled)

Many community groups and private land holders use nest boxes to provide habitat for hollow dependent species in areas with a deficiency of natural hollows. Nest boxes may also be an effective method of detecting the presence of some cryptic species (such as Feathertail Gliders and microbats), and may be a useful tool for medium-long population monitoring studies. Designs are available for 'research' nestboxes that target specific animal groups and can be easily inspected or opened. Nest boxes should only be considered for projects spanning three or more years, and it is important that boxes be regularly checked and repaired when necessary. Researchers must obtain permission from land managers before erecting boxes and AEC approval and a permit is required if animals are to be handled.

Researchers must consider OH & S standards when using ladders to inspect boxes.

2.9 Rock rolling, searching litter and debris (systematic searching)

Target groups:	Herpetofauna and small mammals
Ethics approval required:	Yes – if animals are to be handled
Research permit required:	Yes – if animals are to be handled

This technique involves searching fallen timber, under rocks, under bark or within leaf litter for small cryptic animals. The level of disturbance should be minimised, and the habitat should be returned to its original state if possible. Never reach into or under objects when searching, as they may contain venomous snakes or other dangerous fauna.

2.10 Dip netting

Target groups:	Fish and frogs (adults and tadpoles)
Ethics approval required:	Yes
Research permit required:	Yes

After capture and identification, ensure that tadpoles and fish are put in water as soon as practical or held in appropriate water from their habitat. Return the tadpoles and fish to the point of capture.

2.11 Cage traps

Target groups:	Small to medium mammals
Ethics approval required:	Yes
Research permit required:	Yes

Select appropriate cage size, according to maximum size of target species. Place a waterproof cover around the trap (without impeding any trap-tripping mechanisms) for protection and insulation e.g. black plastic concrete-slab underlay is quite durable. Be aware of the space between the door and the trap as the force of the door closing can damage and break tails. To avoid this, use cage traps that have a door closing mechanism that allows animal tails to drag into trap without getting caught. Also ensure when setting, that the door and locking mechanisms are clear of vegetation and can operate freely.

2.12 Elliott traps

Target groups:	Small mammals and some reptiles
Ethics approval required:	Yes
Research permit required:	Yes

These aluminium traps should be provided with insulation in cold temperatures, using material such as wood, wool or dacron (cushion-filling) or grass and leaves from surrounding vegetation. If possible, place the trap in a sheltered and protected area. If conditions are likely to be wet, place the closed end of the trap in a small plastic bag. It is important to ensure that when placing traps they are sheltered from the sun as much as practical, as in high temperatures animal can suffer heat stress and may die.

2.13 Vertebrate pitfall traps

Target groups:	Small mammals, frogs and reptiles
Ethics approval required:	Yes
Research permit required:	Yes

When installing the buckets, place some cover (e.g. twigs or grass) in the bottom. On the first day of setting pitfall traps, a small amount of water should be added to the trap to provide moisture for trapped animals. Check buckets soon after sunrise. If diurnally animals are likely to be present, particularly reptiles, the traps should be checked again in the late afternoon. Ensure that buckets have drainage holes at the base and that the holes are mesh-covered. In wet conditions, such as during rain or high ground water habitats, when there is a risk of buckets filling with water, pieces of foam or some other floating platform should be placed in buckets to protect animals from drowning. Use battery-operated pumps, a container or sponge to empty water out of buckets if necessary.

2.14 Hairtubes

Target groups:	Small to medium mammals
Ethics approval required:	Yes
Research permit required:	Yes

The most commonly used hairtubes are the Faunatech Funnel, Scotts and Craig tube, Handyglaze Tunnel and PVC pipe tubes. It is important that the surface of the hairtube is clean and devoid of hair before use. Care must be taken when preparing tubes to avoid contaminating the adhesive with any hair or fibres. When placing the tube, angle it to enable drainage in wet weather. Ideally, the hair sample should be removed from the tube in situ, so that the sample isn't contaminated from elsewhere (eg. rucksacks, hairs off your clothes, etc). Recommended sampling period for ground-dwelling mammals is 14 nights. Note that the adhesive can vary in strength and overly sticky sample surfaces have a higher risk of capturing herpetofauna, which may consequently perish or be injured (e.g. lose limbs/digits/tails) when pulling themselves free. Where possible, deploy in colder months to reduce impact to herpetofauna.

2.15 Ultrasonic detection

Target groups:	Microbats
Ethics approval required:	Yes
Research permit required:	Yes

Anabat detectors are commonly used to detect the presence and activity levels of microbats. Positive identification of bat calls requires a comprehensive regional reference call collection covering the study area. To record reference calls, bats must be captured using either harp traps, mist nets or trip lines. All trapping requires AEC approval and a research permit.

Bats captured for reference calls should be released at dusk, just before other bats are observed flying in the area. Recording reference calls generally requires two to three people, one to release the bat, one to follow the bat with a spotlight, and one to record the call on an Anabat detector. Bats cleared from traps at night should be released and recorded immediately, despite the risk of contamination of the recording from other free-flying bats.

More information regarding bat trapping and handling is provided below.

2.16 Harp traps

Target groups:	Microbats
Ethics approval required:	Yes
Research permit required:	Yes

Harp traps should be set in protected positions and guy ropes should be tied securely to prevent the trap falling over. If ants are a problem in the survey area, the legs of the trap and guy ropes can be sprayed to prevent access into the bag. Ensure that no spray is applied directly to the bag. Inspection is recommended during the evening, at a minimum once (perhaps at about 10pm) after the major period of bat activity in the early evening. Traps should be checked and bats removed by mid-morning at the latest. If hot weather is likely, traps should be checked earlier. This is particularly important when females are lactating. Bats captured during the night should be released immediately.

Bats removed from traps in the morning should be held during the day in cloth bags, hung in a cool, dark, quiet place with ample ventilation. The bags should have no loose threads on the inside (i.e. be turned inside out or double hemmed). Multiple individuals can be held together but overcrowding should be avoided. All bats are to be released at the point of capture. Bats should be released that night on dusk, after being warmed up to ensure they fly off readily. Every attempt should be made to return bats to the capture site at dusk, or soon after. However, if this is not possible, bats can be released into tree hollows or under bark during the day, although this increases the risk of predation. During the period when females have dependent young (November – January), all traps are to be checked at least once during the night and any lactating females released immediately. At other times of the year it is preferable, but not essential, to also check traps during the night and release bats soon after.

2.17 Trip Lining

Target groups:	Microbats
Ethics approval required:	Yes
Research permit required:	Yes

Trip lining involves setting up a grid of fine fishing line over small water bodies. When bats fly in for a drink, they hit the lines and fall into the water. The bats then swim to the edge where they are collected. Water bodies with trip lines must be monitored constantly from dusk to dawn, and depending on the size of dam, several people may be needed.

Trip lining for bats should only be undertaken by people experienced in the technique, or under direct supervision of an experienced person. The bats fur should be allowed to dry before the animal is released. The technique should only be used on bodies of water with surface dimensions less than 50 metres in any direction.

2.18 Mist and cannon netting

Target groups:	Birds and microbats
Ethics approval required:	Yes
Research permit required:	Yes

Mist nets should be set in sheltered positions and securely anchored. Traps must be checked at least every 25-30 mins, with more frequent checks during hot or cold weather. Mist nets should only be operated during favourable weather conditions and periods of rain or excessive heat should be avoided. Birds and bats caught in nets should be carefully extracted from the net with care taken to limit the amount of stress experienced by the animal (for a detailed description of extracting birds from mist nets see *The Australian Bird Bander's Manual* Lowe 1989). Captured animals extracted from mist nets should be placed in individual calico bags and transported to a safe site (i.e. banding station) for processing (i.e. banding or measuring). Every effort should be made to limit the amount of time between capture and release.

Mist netting for birds and bats should only be undertaken by people experienced in the technique, or under direct supervision of an experienced person. Cannon netting can only be undertaken under the supervision of a suitably qualified person.

2.19 Electrofishing

Target groups:	Fish
Ethics approval required:	Yes
Research permit required:	Yes

For operation, safety and ethical considerations please refer to The Australian Code of Electrofishing Practice. NSW Fisheries (1997) Australian Code of Electrofishing Practice. NSW Fisheries Management Publication No. 1.

Researchers must complete an accredited training course, involving a theory exam, a period of field experience under the supervision of a fully accredited mentor, and a suite of health checks.

Electrofishing may have impacts on platypus because of their electro sensory organs. There are no records of mortality or harm to platypus during electrofishing studies, but precautions are advised. We recommend that researchers maintain a high visibility while walking along the reach and assessing hazards prior to electrofishing, providing this is compatible with the study objectives. This should cause any nearby platypus to retreat into their burrows or to other reaches. Stopper nets subsequently placed at the upper and lower boundaries of the reach will discourage re-entry of platypus during electrofishing. Studies conducted during the day are less likely to encounter platypus, which are more active at dusk, dawn or night.

2.20 Netting

Target groups:	Fish
Ethics approval required:	Yes
Research permit required:	Yes

When using Mesh or Gill nets, it is prohibited to set them overnight without supervision. The nets should be checked every 10 minutes overnight and when setting nets in waterbodies where platypus are present. Fyke and Larval nets should be set with the cod or bag end out of the water. This enables a platypus to get to the surface to breathe if trapped. It is important that all nets are in good working order.

3. SEDATION, TAGGING AND TISSUE SAMPLING

Ethics approval is required for all sedation, tagging, collaring and tissue sampling activities. The techniques outlined below are not generally part of 'routine' wildlife studies. Staff or students intending to use these techniques must have relevant experience, or seek further training and veterinary advice or assistance.

3.1 Sedation

Mammals

The amount and type of sedative depends upon the species involved and its size and weight. Use a new needle for each injection and administer intramuscularly. After the injection, rub the area for a few seconds to increase the circulation. Make sure you remain quiet while the sedative takes effect, try and keep the animal as quiet and calm as possible. For some species, noise and movement can inhibit the action of the drug. When processing is complete, it is important that the animal is placed back into the trap or bag and allowed to fully recover from the effects of the sedative before being released. Recovery time will vary depending on the sedative and the species. In some cases more than 2 hours may be required. Food can be placed into the trap/ bag as some species will eat while recovering.

Fish

When sedating fish, handling should be kept to a minimum. Clove oil is one of the easiest sedatives to use. The amount used depends on the amount of water the fish is held in. While fish are recovering, place them in well-oxygenated water by using an aerator or fresh and flowing water. When surgical procedures are undertaken, fish are anaesthetised using 0.2 grams/Litre of Alfaxan-CD, either via immersion or for large fish by passing of solution over the gill membrane. The procedure is usually completed within 3 minutes.

3.2 PIT tagging

A (PIT) tag is a small inert passive integrated transponder. PIT tags use the same technology as tags for pets implanted by veterinarians. The tags are encapsulated within inert glass, weigh less than 1 gram and are approximately 10mm long. Veterinary advice and assistance must be sought by any researchers intending to use PIT tags on any species.

Mammals

Ensure that the animal is immobile and kept very still. Lift the skin between the animal's shoulder blades (dependent upon animal) and keeping the needle parallel to the spine insert under the skin. Make sure the tip of the needle does not pierce the muscle layers under the skin. Depress plunger to insert the tag. When withdrawing the needle pinch the skin around the insertion site to ensure the tag doesn't come out as the needle is withdrawn. Rub the area for a few seconds to ensure the tag it is under the skin and away from the insertion point. You may want to swab the area of insertion with a disinfectant. Before and after insertion ensure that you can read the PIT tag.

Bats

PIT tagging has been conducted on macrobats by vets, but has not been used with microbats within Australia. Veterinary advice must be sought.

Fish

Sedate fish first (see technique above) and ensure that the fish is immobile and kept very still. There are two places to insert the PIT tag in fish and it depends upon the size and species. Generally, in small fish the tag is placed into the gut cavity and in larger fish it is placed into the cheek or the dorsal muscle. Firstly, swab the area of insertion with a disinfectant (e.g. Betadine) and depress plunger to insert the tag. After withdrawal, lightly coat the incision area with a flexible cover of surgical adhesive (Vetbond) to further promote healing. After the operation place the fish in an aerated disinfectant salt bath to recover. Return the fish to point of capture. Before and after insertion ensure that you can read the pit tag.

Herpetofauna

Seek veterinary advice.

3.3 Other marking techniques

Toe clipping

Toe-clipping involves the removal of unique combinations of toes from frogs and lizards in order to identify them upon recapture. A maximum of three toes should be removed, as studies have shown negative impacts on amphibians if more are removed. Toes are removed by cutting at the base of the terminal phalange on lizards and the base of the penultimate phalange on frogs. Sterile high quality surgical scissors must be used for this procedure and then sterilised between each procedure by flaming with ethanol and rinsing in betadine. A mild antiseptic, such as betadine can be applied to the wound. Bleeding may occur with larger lizards after toe clipping, in which case surgical glue (eg. Vetbond) may be applied to the wound.

Bird banding

The banding of wild birds in Australia can only be carried out under a project approved by the Australian Bird and Bat Banding Scheme (ABBBS) using approved bands issued by the ABBBS. Bird banders must also hold a current ABBBS Bird Banding Authority to band birds in Australia. The levels of authority are dependent on the experience of the bird bander.

Bird bands are to be placed on the tarsus of the bird with care taken to ensure that the right type and size of band is placed on the bird (refer to *Recommended Band Size List: Birds of Australia and its territories* ABBBS 2000).

The process for banding birds is briefly summarised below (from Lowe 1989):

1. Determine the bird species and refer to the current List of Approved Band Sizes for the correct band size and metal type to use.
2. Record the band number and select the appropriate banding pliers.
3. Carefully close the band around the bird's leg using the correct sized ABBBS pliers and check to ensure it is applied correctly and that there are no sharp edges.
4. Recheck that the correct band number has been recorded.
5. Release the bird near the site of capture.

The colour banding of birds using anodised aluminium or stainless steel, darvic or celluloid bands can only be undertaken under a current Colour Marking Authority approved by the ABBBS.

Ear tagging and collaring

A variety of ear tags and collars are available and it is important to use the most suitable type for a particular species. For medium-sized ground-dwelling/arboreal mammals, numbered metal 'fingerling' type tags may be suitable. These tags are fitted with a specialised tool resembling pliers. Ensure that you practise with them to become familiarised with their operation before using on an animal. Ensure that the tag is fitted to a strong part of the ear (e.g. the thick front as opposed to the thinner top of the ear). Check that blood vessels are avoided. Have antiseptic/alcohol on hand to swab the area before and after fitting. Ideally, tags of the same number should be fitted to both ears.

Fingerling tags should not be used on small mammals with delicate ears (such as *Antechinus*, *Sminthopsis* and *Acrobats*). Many of these species claw at the tags until they rip out, seriously damaging the ear.

Ear tattooing

The animal should be anaesthetised, and the person well trained. Place the tattoo pin at shortest length to ensure ear does not tear. The ear should be swabbed with alcohol before and after procedure.

Radiotagging and collaring

Radiotags can be attached to an animal in a number of ways. The type and weight of the radio tag depends on the species, however the general rule is that the tag should be less than 7% of the body weight. If the tag is to be administered internally or the animal is stressed and cannot be calmed, the radiotags should be fitted while the animal is under sedation. Researchers should refer to Kenward (1987) for a wide-ranging discussion of techniques of radiotelemetry. Investigators new to the techniques of transmitter attachment should also refer to literature related to the target animal group, and the internet, for the latest developments in telemetry techniques. The guiding principle is to minimise the impact on the movement and behaviour of the animal and to avoid short-term and long-term injury resulting from the transmitters.

Global Positioning System (GPS) collars

GPS collars have been used successfully by Federation University Australia researchers to study the movements of Feral Goats and Deer. The GPS receiver is attached to a leather collar placed around the animal's neck. The collar also houses a radio transmitter, batteries and data logger. The GPS can be set (before putting on the animal) to record the animals position at fixed intervals (eg. every 30 minutes), and the collar is set to fall off at a particular date and time, avoiding the need to recapture the animal. The radio transmitter provides a way of locating the animal while the collar is on, to check on the animal's location, behaviour and welfare. After the collar falls off, the researcher locates it by finding the radio transmitter. Unfortunately there is no way of knowing if the GPS is functioning correctly (ie. recording the animals position at regular intervals), until the collar is retrieved.

Other types of GPS animal monitoring systems are also available, involving the continuous transmission of the GPS position. These systems require a form of long-distance communication, either by radio or mobile phone. These systems are considerably more expensive, complicated and prone to failure, but may be the only option if the GPS data logger cannot be retrieved. These systems have been used with marine mammals and birds (Albatross).

The AEC must approve techniques used to capture, sedate and handle animals to be fitted with GPS collars.

3.4 Tissue and hair sampling

Tissue sampling can be done in a number of ways. Generally, sedation isn't required for small samples of tissue. Ensure that the animal is immobile and kept very still. Sampling can be taken from the ears of mammals, fins of fish, tail tips of reptiles and the toes of frogs. Hair samples can also be taken. Samples must be taken with sterile scissors and they must be sterilised with each procedure by bleaching and flaming with ethanol.

4. STUDIES OF DIET AND REPRODUCTION

4.1 Dietary analysis

Diet is an important component of ecology, and hence its consideration is often a vital aspect of wildlife research. Some methods of investigating diet are non-invasive (eg. observations of foraging, faecal analysis), but other techniques are variably discomforting or destructive. There is now no justification for killing native animals solely in order to obtain stomach samples.

Stomach flushing (typically using saline solution) is now used routinely for many vertebrates and is generally regarded as fairly benign, provided the operator is competent with the procedure.

The use of chemical emetics is considered more intrusive and risky. These have been used for dietary studies on birds, with patchy success and impacts.

4.2 Reproductive studies

Reproduction is a fundamental component of the biology of wild animals and hence of valid interest to researchers. However, investigation of reproduction may be a field particularly prone to detrimental impacts from the observer. The visits of researchers to nests, maternity roosts or other reproductive sites may lead to increased predation, short-term or permanent abandonment by parents, damage to eggs or dependent young, or premature departure or young. Impacts may be magnified where reproduction is concentrated in colonies.

For reproductive studies of birds, detrimental impacts can be minimised through the use of telescopes, hides, careful timing of visits, and gradual habituation. Maternity roosts of bats should not be disturbed.

5. HYGIENE, SPECIMENS, EUTHANASIA AND TRANSPORT

5.1 Safety and hygiene

Before commencing any fieldwork, staff and students must complete an OH & S risk assessment using the University's '*Hazard Identification, Risk Assessment and Control*' (HIRAC) approach.

After handling animals and scats, hands should be washed to reduce the chance of contracting zoonotic diseases. Hands should be routinely washed with a disinfectant such as hexol.

If bitten, first aid should be administered and medical assistance sought as appropriate. All staff should have current Level 2 first aid accreditation. Staff and students handling animals should have an up to date tetanus shot. Minor scratches and bites should be promptly washed in soapy water, and a tetanus booster should be arranged within a week of the injury.

Although Lyssavirus has not been confirmed from the majority of microbats in Victoria, there is the potential for it to occur in any species. Therefore, adequate precautions need to be taken when handling any bat. People regularly handling bats should have the pre-exposure rabies vaccinations. For people not vaccinated it is essential that they wear fine leather gloves to prevent being bitten. If bitten the wound should be washed for 10 mins in soapy water and then medical attention sought. This may include the series of post-exposure rabies shots.

5.2 Voucher and archival Specimens

The two types of voucher specimens are the specimen that you intend to kill and the non-target specimen. For euthanasia, see below. For the procedure for preserving and labelling dead specimens see the *Wildlife Act, 1975* and *National Parks Act, 1975*. The Department of Land, water and Planning research permits specify how accidental deaths should be preserved, labelled and stored.

5.3 Euthanasing animals

Projects involving the euthanasia of animals require AEC approval. .

Methods must be humane and produce a painless death as rapidly as possible. Methods which are acceptable are described in Reilly, J (2nd ed) "Euthanasia of animals used for Scientific Purposes" ANZCCART 2001 (Australian and New Zealand Council for the Care of Animals in Research and Teaching). In some instances it may become necessary to euthanase animals due to accidental injury etc. Appropriate practical and humane methods are to be employed wherever possible. The best course of action that minimises suffering should be determined. All euthanasia incidents are to be reported to the AEC.

All researchers should be familiar with the ANZCCART euthanasia guidelines. The guidelines list recommended euthanasia techniques for all groups of animals, and list techniques that are 'acceptable with reservations'. For most groups of animals the only recommended techniques involve the injection of chemicals such as pentobarbitone sodium. For some groups of species, no techniques are recommended.

Generally, wildlife research studies conducted by the Federation University Australia only involve the killing of introduced species (such as the House Mouse, Black Rat or Feral Cat) or the euthanasia of seriously injured animals. The table below lists euthanasia techniques considered 'acceptable with reservation' by the ANZCCART guidelines.

Species group(s)	**Acceptable euthanasia techniques**
Rats, Mice, small marsupials	Stunning followed by cervical dislocation
Microbats	Cervical dislocation
Small and medium birds (up to 150g)	Cervical dislocation
Reptiles (excluding crocodiles), Frogs, Fish	Stunning followed by destruction of the brain
Invertebrates	Cooling followed by freezing
All other groups – including Feral Cats and Dogs, large birds, medium and large marsupials, larger feral and domestic animals, macrobats.	Seek veterinary assistance

For some animals (eg. feral cats, large birds, macropods), shooting may be acceptable. This may be appropriate in some cases if the researcher has access to trained and licensed person. Generally, shooting will not be an option. Cats or dogs that are obviously pets (friendly animals or animals with collars and registration labels) should be taken to the local council pound or RSPCA animal shelter.

The ANZCART guidelines describe the way in which these euthanasia procedures should be carried out. Stunning and cervical dislocation are briefly described below.

Cervical dislocation of small birds up to 150g

This method involves the placement of the thumb and index finger on either side of the neck at the base of the skull, or, alternatively, a rod is pressed at the base of the skull. With the other hand, the base of the tail or hind limbs are quickly pulled, causing separation of the cervical vertebrae from the skull.

Stunning and cervical dislocation of small mammals up to 150g

Stunning is generally regarded as producing immediate unconsciousness. Small mammals may be stunned by holding the tail (mice) or hind quarters (rats) and swinging the animal down quickly so as to strike the back of the head on the edge of a bench or other suitable object.

Cervical dislocation involves holding the animal prostrate on a bench (or other flat surface) with the thumb and forefinger of the operator firmly squeezing the neck behind the head of the animal. The hindquarters of the animal are grasped firmly with the free hand and pulled caudally. An instrument such as the blades of a pair of scissors, or a firm steel rod can be used instead of the thumb and forefinger.

5.4 Care of injured animals

Captured native animals that have minor injuries should be taken to a veterinarian for treatment. Prior to lodging an application for AEC approval, researchers are required to obtain current contact details (including after-hours contact details) of veterinarians that operate in the particular area where the research is being carried out.

5.5 Transport

Guidelines for the transport of wildlife are described in Clause 3.2.5 of the Australian Code of Practice for the Care and Use of Animals for Scientific Purposes, 8th Edition 2013.

Key points include:

- The conditions and duration of the transportation must ensure that the impact on animal health and welfare is minimal.
- Containers must be secure and escape-proof. There should be adequate nesting or bedding material and animals must be protected from sudden movements and extremes of climate.
- Water must be provided when necessary.

5.6 Temporary storage

It may be necessary to hold animals for short periods following identification, prior to release at an appropriate time. For example, microbats cleared from traps checked in the morning should be held during the day and released at dusk. Animals should be held in a quiet, safe, dark, well ventilated area without extreme temperatures. There should be adequate nesting or bedding material and it may be necessary to provide water. Handling should be minimised, to allow animals to remain settled.

6. SOURCES OF FURTHER INFORMATION

- Australian code of practice for the care and use of animals for scientific purposes. Australian Government, National National Health and Medical Research Council. 8th Edition 2013.
- Reilly, J (2nd ed) "Euthanasia of animals used for Scientific Purposes" ANZCCART 2001.
- The Australian Code of Electrofishing Practice. NSW Fisheries (1997) Australian Code of Electrofishing Practice. NSW Fisheries Management Publication No. 1.
- Tracks, Scats and Other Traces - A Field Guide to Australian Mammals, by Barbara Triggs.
- Birds Australia Code of Practice for the Nest Record Scheme.

  Available online at: http://www.birdsaustralia.com.au
- Federation University Australia AEC website: http://www.federation.edu.au/research-and-innovation/research-support/ethics

  The AEC website provides a list of meeting dates and application due dates. Application and report forms can be downloaded in Microsoft Word format.