General Guidelines for Wildlife Capture and Handling

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DOCUMENT STATUS AND AIMS

The primary purpose of the document is to provide guidance to staff, students and associates 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 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 reviewed by all staff, students and associates prior to their undertaking fieldwork involving wildlife survey. Students, associates 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 gaining AEC project approval 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.

Version number	Author	Reviewed by	Approved by Fed Uni AEC
2008	Matthew Gibson & Dr Grant Palmer	UB AEC	
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1. INTRODUCTION

These guidelines have been produced 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) and to staff, students and associates that may be involved in wildlife capture and handling.

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 8th (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 as identified in these guidelines. 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. The Principal Investigator should be affiliated with Federation University Australia.

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, students and associates 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 (DELWP) 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, Office of Environment and Heritage.

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 at least 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, field contact number and proposed activities.

1.2 Borrowing of equipment

School of Health and Life Sciences

The School of Health and Life Sciences at Federation University Australia has a large store of wildlife research equipment, including Elliot traps, cage traps (several types), harp traps, hair tubes, pitfall traps, remote cameras 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 when deployed in the field.

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 annually. Records of captures within Victoria must be submitted to the Department of Environment, Land, Water and Planning. The information is added to the Victorian Biodiversity Atlas, 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.

2. OBSERVATION AND TRAPPING TECHNIQUES

2.1 General considerations

Skills

All staff, students and associates 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, associates and staff may also participate in animal trapping and handling, but they must work under the direct supervision of experienced teachers or researchers who are named on the AEC project approval.

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 may 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. The nature of the bait to be used must be detailed in the AEC project application.

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 individual 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 (e.g. open forest, heathlands) and 10-20m in more open habitats (e.g. 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 the breeding period of any target species or potential by-catch species, to minimise stress to animals. If possible, breeding 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 permanently marked, it may be desirable to mark individuals temporarily by painting a spot of fur or skin using a non-toxic paint, dye, texta or liquid paper. 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 heat, 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 (e.g. in dense shrubbery, or within grass tussocks), away from public view. Added protection from rain and wind should be provided for animals held in cage traps or Elliott traps, potentially by using plastic covers (see section 2.11). It is recommended to use insulating material (e.g. Dacron filling is widely considered an appropriate material to use) inside mammal traps, and a piece of canvas, or other shelter material, in the bottom of pitfall traps. Ensure that the material 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 (generally) 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 the animal from the trap as soon as practical and place in a cloth (e.g. calico) 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.

Releasing animals

When releasing animals, animals should be released at the point of capture, or as near to as possible. Care must be taken when releasing animals to ensure that they have ready access to appropriate shelter. Care must be taken to ensure released animals are not targeted by predators in the vicinity (for example, currawongs have been known to immediately target released animals on occasions).

Nocturnal animals removed from traps during daylight hours should be held appropriately during the day for release that night.

Any departure from these release guidelines, must be justified in the AEC project application.

2.2 Bird observing

Observing free-flying and free-roaming birds, nests or roost sites in the natural environment with or without the aid of equipment, including binoculars, spotting scopes and cameras. Observations may include visual and audio observations.

Target groups:	Diurnal and nocturnal 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 BirdLife 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

Observing free-roaming animals, nests or roost sites in the natural environment at night with the aid of a spotlight or torch.

Target groups: Nocturnal arboreal and terrestrial mammals, birds, reptiles, frogs
Ethics approval required: Yes
Research permit required: No – Yes if being conducted in parks or reserves

At all times avoid overexposing animals to the light and keep noise to a minimum. Be aware that nocturnal animals may have eyes that are sensitive to light. A light with a low wattage should be used (30-50 W). Where circumstances require (such as in tall forest) a narrow beam of brighter light may be briefly used to focus the search area and to enable identification of animals. A red filter, or dimmer light, should be used to reduce the intensity of light being directed at an animal once it has been located. If observations greater than 30 seconds are required, a red-filtered light must be used. 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

Opportunistic observation is the general observation of animals that are encountered in the wild without use of active searching (see 2.9 Active searching), traps or other capture devices.

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

Observing secondary signs to determine animal presence, including tracks, scats, scratches, burrows/tunnels/run-ways, feeding signs, hair/feathers, and bones/carcasses. This does not involve any capture or handling of animals. Note, materials such as bones and feathers cannot be removed without a valid scientific permit.

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

Collection of predator scats for the purpose of investigating the contents of the scat to determine potential prey items.

Target groups: Mammals, birds, herpetofauna
Ethics approval required: No
Research permit required: No (yes if collected from public land)

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 (e.g. washing and drying) the scats in the laboratory to reduce the cost.

2.7 Call playback

The call(s) of the target animal, or other animal, is broadcast to encourage a response, either a call back or the direct approach of the target animal.

Target groups:	Owls, arboreal mammals, cryptic bird species, frogs
Ethics approval required:	Yes
Research permit required:	No (yes if conducted on public land)

Researchers must be capable of aurally identifying bird, mammal or other species that are likely to respond to the broadcast call. 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. Calls should be broadcast in short bursts (i.e. 10-30 seconds of call followed by 2 minutes rest) Avoid excessive use of playback at single sites, as repeated playback can cause significant disturbance to resident birds. No more than two 15 minute sessions per night should be used in an individual's home range. Use of call playback should be during the breeding period of the target animal.

2.8 Nest boxes (and natural hollow inspection)

Hollow-using animals, such as arboreal mammals, bats and birds, can be monitored by erecting nestboxes and making observations of their use. Observations of animal use of nestboxes may be made from a distance (i.e. observing animals entering or exiting the nestbox) or by direct checks of the nestbox contents. Direct checks may be done by accessing the box directly and viewing its contents through an opening (i.e. hinged lid), or by viewing the contents by directing a camera (e.g. pole-mounted nest view camera) into the nestbox entrance to gain a 'live feed' of the contents.

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

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. Where possible, checks of nest boxes should avoid key breeding times for target species or other species potentially using nest boxes.

Researchers must obtain permission from land managers before erecting boxes and AEC approval and a permit is required if animals are to be handled or if direct checks of the nest box contents are to be undertaken.

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

2.9 Active searching - Rock rolling, searching litter and debris (systematic or targeted searching)

This technique involves searching fallen timber, under rocks, under bark or within leaf litter for small cryptic animals. The habitat features targeted are determined by the habitats used by the target species. If searching litter, a rake may be used to carefully shift litter.

Target groups:	Herpetofauna and small mammals
Ethics approval required:	Yes – if animals are to be handled or if habitat is to be disturbed during the search
Research permit required:	Yes – if animals are to be handled or if habitat is to be disturbed during the search.

The level of disturbance should be minimised, and the habitat should be returned to its original state if possible. Care must be taken when moving objects (e.g. logs or rocks) not to crush or otherwise injure animals that may be underneath. Never reach into or under objects when searching, as they may contain venomous snakes or other dangerous fauna.

2.10 Dip netting

This technique involves using a net on the end of a pole to manually capture aquatic animals, such as fish and frogs (adults and tadpoles). The net is moved through the water to capture animals that may or may not have been observed by the net operator above. The mesh size of the net is determined by the size of the animal targeted.

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

Cage traps, constructed of a metal frame and covered by wire mesh, are used for the live capture of small to medium sized animals. There are a range of trap sizes and designs available. They operate using a treadle plate mechanism, which operates a spring release, shutting the door when triggered. Animals are attracted into the trap and on to the treadle plate by a bait placed at the enclosed end of the trap. Cage traps are typically placed on the ground to target ground-based animals, but may be adapted for tree use using a suitable bracket.

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. Cage traps will also catch a range of non-target species, such as bird and reptiles and this must be considered when establishing appropriate inspection prototcols.

2.12 Elliott traps (or similar aluminium, enclosed box trap designs)

Elliott traps (or similar aluminium, enclosed box trap designs) are used for the live capture of small to medium sized animals. There are a range of trap sizes available, but the most commonly used is the version designed for small mammals (up to 250 g) (trap dimensions 90 mm x 100 mm x 330 mm). They operate using a treadle plate mechanism, which operates a spring release, shutting the door when triggered. Animals are attracted into the trap and on to the treadle plate by a bait placed at the enclosed end of the trap. Elliott traps are typically placed on the ground to target ground-based animals, but may be adapted for tree use using a suitable bracket.

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

These aluminium traps should be provided with insulation, particularly in cold environments, using material such as wool or Dacron (cushion filling). The old practice of using of grass and leaves from surrounding vegetation should be avoided. It is also recommended that the enclosed end of trap be covered by a plastic sleeve when trapping in damp environments or where there is a chance of rain during the trapping event. If possible, place the trap in a sheltered and protected area (e.g. against a log or under low vegetation). If conditions are likely to be wet, place the closed end of the trap in a small plastic bag so that the trap is effectively water-proofed by the plastic layer. 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 (here it is important that researchers consider the path of the sun during the day).

2.13 Arboreal pipe traps

Arboreal pipe traps are plastic pipes (generally 90 mm diameter), attached vertically to a tree trunk, with an open spout at the top and a sealed end (consisting of a screw cap) at the bottom. The depth of the trap is such that the target animals are unable to escape once they fall into the trap (typically around 700 mm). The traps are used to survey possums, gliders, phascogales and other arboreal mammals.

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

Arboreal pipe traps should be securely fixed to trees at a safe operating height (3-5 metres). Traps should be positioned such that they are as protected as much as possible from direct sunlight and prevailing weather conditions. Material such as wool or Dacron (cushion filling) should be included in traps to provide additional shelter for animals. For some attachment designs, it may be possible to remove the trap from the tree using a pole operated from the ground during inspections.

2.14 Vertebrate pitfall traps

Pitfall traps are plastic buckets or tubes dug into the ground so that the lip of the opening is flush with the ground surface. The depth of the trap is such that the target animals are unable to escape once they fall into the trap. Pitfall traps may be used in association with a drift fence that intercepts the path of the animal and funnels it towards the opening of the trap (this increase the potential capture area). The drift fence is typically aligned along a row of traps or as a series of 'arms' radiating out from a trap. A drift fence is typically around 30cm in height with several centimetres buried in the ground to improve its impenetrability. The drift fence is typically constructed from 1mm fly mesh and is held in place by metal or wood stakes.

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

When installing the buckets, place some cover (e.g. piece of canvas, 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 if they are open during the day period. 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 high risk of buckets filling with water buckets should be emptied and closed. Where there is a lower risk of buckets filling with water, pieces of foam or some other floating platform should be placed in buckets to protect animals from drowning should the buckets fill and hold water. If a bucket contains water it should not be opened until the water is removed. Use battery-operated pumps, a container or sponge to empty water out of buckets if necessary.

Animals trapped in pitfalls are vulnerable to being injured or consumed by other animals (particularly ants, but also centipedes and spiders) or preyed upon by other trapped animals (e.g. large reptiles, dasyurids).

2.15 Hairtubes

Hair tubes include small tubes or specifically designed apparatus, such as Faunatech Hair Funnels, lined with sticky tape or wafer and baited to attract small mammals. The animal enters the hair tube, brushing against the sticky tape and depositing a hair sample which remains on the tape. It is then possible to analyse the hair sample to identify the species involved. Hair tubes are a cost-effective, unobtrusive survey technique and may be useful for rare or trap shy mammals.

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. FedUni AEC encourages the use of Faunatech Funnels due to features which improve the welfare of animals, including a floor area free from adhesive, and specially designed structure and wafer tackiness that limits potential by-catch or entrapment of small vertebrates (e.g. skinks). 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 (e.g. 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. Hairtubes should be inspected daily to check for entrapped animals, where this is considered a potential issue. The type of hairtube to be used must be described in the AEC application for approval by the AEC.

2.16 Remote cameras

Remote cameras (also widely referred to as camera traps) are set in order to be triggered by animals moving in the vicinity through a motion or infrared sensor and resulting in the capture and storage of an image or sequence of images. Remote cameras may be set up to be passive (i.e. to simply record animals wandering past) or active (i.e. animals are attracted to the detection zone of the camera by use of a lure (either a bait, scent or sound recording)). Remote cameras allow for the observation and monitoring of cryptic species in a non-invasive way (i.e. there is no handling of animals involved and research personnel are not present).

Target groups:	Mammals, birds, reptiles, frogs
Ethics approval required:	Yes – if a lure is used to attract animals to the vicinity of the camera
Research permit required:	Yes (if used on public land – parks and reserves)

Remote cameras have quickly become a commonly used technique to detect the presence of a wide range of animals, including ground-based mammals and birds. They are also used to target animals in specific situations, such as at nest sites, bait stations, along paths, shelter sites or carcasses. Remote cameras are generally operational for relatively long periods of time at a site (i.e. weeks). It is important that they are securely fixed to their attachment (examples of attachments include trees and posts). Remote cameras should be arranged in the field so as to improve the capability to record target animals. This includes considering the height of deployment, where the camera is focused, the range of the sensor, the timing of the camera operation and the sequences and quality of images per trigger. When using infrared cameras it is important to avoid pointing the camera into direct sunlight as this may contribute to false triggers. Similarly, for movement activated cameras it is important to prepare the detection zone so that pieces of moving vegetation do not lead to false triggers.

Lures, such as baits, are commonly used to encourage animals into the detection zone. The decision to use lures will depend on the purpose of the monitoring activity. Using lures will interrupt the natural behaviour of any animals using the area and may influence the activity rates (typically measured as the number of 'hits' (i.e. number of images) captured on the camera). Using bait could also attract potential predators to the vicinity. Careful consideration of the need to use lures is required.

2.17 Audio detection

Audio detection of animal calls using sound recorders, such as Song Meters and voice recorders, is a non-intrusive technique that is highly efficient and effective. Calls are recorded on devices placed in the field and then downloaded for automatic analysis using specialist software (e.g. Raven Pro) or manually by listening to the recorded calls.

Target groups:	Birds, frogs, vocal mammals
Ethics approval required:	No
Research permit required:	No (yes if used on public land – parks and reserves)

Sound recorders are commonly used to detect the presence and activity levels of animals. The identification of calls requires sufficient skills, but is increasingly aided by the development of automated call recognition software and applications. Positive automated identification of calls requires a comprehensive regional reference call collection covering relevant to the study area.

2.18 Ultrasonic detection

Ultrasonic detection of bat calls (i.e. echolocation) using specialist recorders, such as AnaBat and Song Meter, is a non-intrusive technique that is highly efficient and effective. Calls are recorded on devices placed in the field and then downloaded for automatic analysis using specialist software (e.g. AnaScheme).

Target groups:	Microbats
Ethics approval required:	No
Research permit required:	No (yes if used on public land – parks and reserves)

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.

Please note, where a suitable reference call collection does not exist, it will be necessary to catch local bats and record their call upon release. 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.

2.19 Harp traps

Harp traps consist of two banks of taut vertical fishing lines arranged within a frame that sits on a set of adjustable legs, usually arranged to catch microbats flying within a zone 1 to 4 metres above the ground. Flying microbats hit the lines and fall into a canvas holding bag at the bottom of the lines. Bats climb up the side of the bag and are protected under a sheet of weather proof plastic. A trap can catch multiple individuals at a time.

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 as soon as possible after sunrise, at least within 3 hours of sunrise. 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 at the location of capture, 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.

2.20 Trip Lining

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. This technique has now largely been superseded by use of Harp traps (see 2.18 Harp traps). This technique will not be approved for use by the AEC.

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

2.21 Mist nets

Mist nets are routinely used for the capture of birds, and to a lesser extent bats. Mist nets work by entangling flying animals is a fine mesh net strung between two poles and positioned across likely animal flight paths.

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

This technique also requires that researchers hold the appropriate ABBBS authority and endorsement(s) to conduct mist netting. To gain these endorsements, researchers must undertake extensive training in the techniques under the supervision of experts.

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.

2.22 Cannon nets

Cannon nets are routinely used for the capture of birds, in particularly flocking waders and shorebirds. Cannon nets operate by firing a net strung between two projectiles out and over a target group of birds. This technique takes a high level of skill and experience and must only be performed by experienced sufficiently licensed operators.

Target groups:	Birds, particularly waders and shorebirds
Ethics approval required:	Yes
Research permit required:	Yes

This technique also requires that researchers hold the appropriate ABBBS authority and endorsement(s) to conduct cannon netting. To gain these endorsements, researchers must undertake extensive training in the techniques under the supervision of experts.

Cannon netting is only suitable as a technique in very specific situations, such as open shorelines where waders and shorebirds congregate. There are not insignificant risks associated with the techniques potential to injure animals that must be considered in establishing a safe protocol for surveys. If considering use of this technique then researchers must seek advice of relevant organisation, such as Victorian Wader study Group.

2.23 Electrofishing

Electrofishing is a fish sampling tool that involves passing an electrical current through water, stunning fish so that they float to the surface, allowing netting and processing (identification, size measurement, etc.). Fish recover quickly and are released back into the water.

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

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.

For operation, safety and ethical considerations please refer to *The Australian Code of Electrofishing Practice*, NSW Fisheries (1997).

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.

2.24 Netting fish (gill nets, fyke nets, seine nets and similar)

A variety of netting approaches are used to capture fish. These nets may be actively used (i.e. dragged by a boat or from shore) or passive (i.e. left in situ). The mesh size of netting is determined by the target animals.

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 species which may be susceptible to drowning, such as 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.

Birds

Seek veterinary advice if intending to sedate birds as part of research.

Herpetofauna

Seek veterinary advice if intending to sedate herpetofauna as part of research.

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.

Birds

Seek veterinary advice if intending to undertake PIT tagging with amphibians.

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 if intending to undertake PIT tagging using herpetofauna.

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. Toe-clipping is no longer a technique that will be approved by the FedUni AEC and suitable alternatives must be used (e.g. PIT tagging or UV fluorescent marking).

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.

Visible Implant Elastomer (VIE) (including fluorescent ink use)

VIE is created by mixing a coloured elastomer and a curing agent. The mixture is injected under the animal's epidermis and solidifies to form a small rubber-like mark, which is visible externally. Different combinations of colours and locations on the body can then be used to identify individuals. The mark is long-lasting (>2 years). The technique has commonly been used for marking fishes, however more recently it has been applied successfully to reptiles and amphibians with good results. An alternative form is the use of fluorescent ink, which is injected in similar way and which can be detected later using an ultraviolet light.

The methods for these techniques is clearly described and discussed in detail in Waudby and Petit (2011) and Petit *et al.* (2012). While the application of the technique is considered straightforward, it requires that that researchers be adequately trained in the technique before its use.

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. Investigators new to the techniques of transmitter attachment should 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.

The AEC must approve techniques used to capture and handle animals to be fitted with radiotagging devices and to the attachment method to be used.

Global Positioning System (GPS) collars

GPS collars have been used successfully by Fed Uni 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, hair and DNA sampling

Tissue, hair and DNA 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. Any intentions to collect tissue, hair and DNA samples, and the procedure to be followed, must be clearly defined in the AEC project application and only conducted under AEC project approval.

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 a 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. During handling of wildlife, staff, students and associates should take particular care to avoid putting their hands near their mouth or eyes, which present high risk infection transmission sites.

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 (Fed Uni staff handling bats are required to be vaccinated). 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 Environment, 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. It is expected that all project personnel that may be required to euthanase animals is appropriately trained and experienced in the use of the technique required.

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 decision to humanely kill an animal must be made on a measured consideration of the perceived degree of suffering by the animal and the chances of recovery. If an animal is severely injured and is suffering, it must be relieved of its suffering as soon as possible. The decision to euthanise an animal must be supported by using a humane technique that produces a painless, rapid death, that does not unreasonably alarm or stress the animal. 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 a 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.
- BirdLife Australia Code of Practice for the Nest Record Scheme.
- Available online at: http://www.birdlife.org.au
- Federation University Australia AEC website: http://federation.edu.au/research/support-for-current-students-and-staff/ethics/animal-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.
- Waudy, H.P. and Petit, S. (2011). Comments on the efficacy and use of visible implant elastomer (VIE) for marking lizards. South Australian Naturalist 85, 7-13. (see https://search.informit.com.au/fullText;dn=251857167920976;res=IELHSS)
- Petit, S., Waudby, H.P., Walker, A.T., Zanker, R. and Rau, G. (2012). A non-mutilating method for marking small wild mammals and reptiles. *Australian Journal of Zoology* **60**, 64-71. (see http://www.publish.csiro.au/zo/pdf/ZO11088)