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Review

Standard Operating Procedures for aluminium box, wire cage, and pitfall trapping, handling, and temporary housing of small wild rodents and marsupials

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Abstract. Many researchers and educators need to provide Standard Operating Procedures (SOPs) to their Animal Ethics Committee (AEC) for the purpose of trapping, handling, and temporarily housing small mammals. We devised general SOPs that are compatible with most existing ones for Australia and had these SOPs reviewed by a panel of Australian experts. The SOPs may be used as guidelines by researchers who need to provide such protocols to their organisation or AEC, or in teaching.

Additional keywords: animal ethics, guidelines, protocols, small mammals, trapping wildlife.

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Introduction

Standard Operating Procedures (SOPs), in this case detailed instructions on how to achieve a task using widely accepted procedures at a point in time, are required by some Animal Ethics Committees (AECs) in Australia and elsewhere. SOPs ensure that animal users work within guidelines and legal obligations relevant to the conduct of research or teaching involving animals. Researchers and teachers are required to handle and house animals in accordance with the 7th edition of the 'Australian Code of Practice for the Care and Use of Animals for Scientific Purposes' (Australian Government 2004). This Code is now under revision. Of the 3 Rs (replacement, reduction, and refinement) underlying the Code, field mammalogists are most likely to be able to act on refinement (Lunney 2012) using technology (e.g. camera traps (Mccallum 2012), which may also be considered as replacement in certain cases) and improved trapping and handling. To standardise and improve efficiency in the process of refinement, AECs may require SOPs from researchers and teachers using animals for both field and laboratory conditions. Many AECs are unfamiliar with field techniques, but researchers have access to only one AEC (Lunney 2012), so a need exists for these committees to understand the range of acceptable options.

This paper provides SOPs for the capture and handling of small species of wild rodents and marsupials. They may be used by researchers and teachers using these animals during field work, where captivity will be temporary. Some agencies have already produced SOPs (e.g. Western Australia's Department of Environment and Conservation and the Department of Environment and Natural Resources (formerly Department for Environment and Heritage) in South Australia). The intent is not to duplicate their work, but to establish a basic set of practices deemed acceptable by a wide range of Australian experts and organisations, and that may help to avoid confusion when faced with a range of different protocols. Detailed information is valuable in teaching. These SOPs may be useful to universities and organisations that currently offer scant guidelines, including the 'Guide to the Use of Australian Native Mammals in Biomedical Research, Public Consultation Draft' (Australian Government 2012). The current paper is not meant to be prescriptive, but rather provides a basis for SOPs that may be adapted to a local situation or particular species and environmental conditions.

Projects requiring extensive captivity of animals would require additional SOPs. The focus here is on small mammal species (up to *Rattus* spp. size approximately) that are normally captured in aluminium box-style traps (e.g. Elliott brand), wire cages, or pitfall traps, and held in captivity for less than one day. However, it is important to note that non-target species may also be captured, and that field researchers should be prepared for this eventuality.

Methods

We initially reviewed literature including existing SOPs/guidelines/protocols from various organisations, and included our own experiences in capturing and handling small mammals in South Australia, New South Wales, and overseas. The SOPs presented here are compatible with those of Western Australia's Department of Environment and Conservation (Freegard and Richter 2009*c*; Richter and Freegard 2009*a*), Museum Victoria

(2009), the South Australian Government guidelines for vertebrate surveys (Owens 2000), and Queensland's Department of Environment and Resource Management (DERM 2009), but they provide more detail on suggested methods and synthesise the information. They are also in agreement with the guidelines of the American Society of Mammalogists (Sikes *et al*. 2011). We devised a relatively comprehensive text combining all key recommendations, including those from wildlife authorities with extensive field experience. These experts have worked in different states and types of organisations; we included one veterinarian and one consultant (Table 1). Their comments were then incorporated into the paper before it was submitted for publication. Not all experts agreed on every detail of each procedure, and not every comment could be incorporated into the paper, when mild conflicts existed between experts regarding specific procedures. We made the final choice on what is presented; however, the panel agreed that the procedures presented were considered ethical at the time of writing, and disagreement was mild. The text is written in the present tense of the indicative mood, so as to avoid the repetition of 'must' and 'should'.

Here we assume that the preparation required for any trapping has been conducted, including planning (e.g. Owens 2000), study design and methodology, evaluation of welfare issues, and arrangement for emergency procedures. This preparation is essential to conduct scientifically and ethically sound projects, with minimal impacts on animal populations and the natural environment, and to ensure that enough resources are available for the project to achieve its goals.

Precautions that trappers should take

General precautions

Ecologists trapping animals have four priorities: protect themselves, protect the animals, protect the environment, and protect the data. Trappers need to seek information on, and be aware of, health risks specific to the conditions and be prepared (e.g. wear appropriate clothing). Diseases may be transmitted by the trappers to the animals, and many documented cases of diseases transmitted to trappers by animals, or their parasites, exist. It is not the intent of this document to review the diseases concerned, but simple precautions minimise the risk of disease transmission. Trappers wash their hands thoroughly before and after handling mammals and, if used, their gloves are disinfected or discarded. Disinfecting hands with an alcohol-based sanitiser may also be helpful. Trappers are trained to minimise bite risks and are vaccinated against tetanus; in certain cases, other vaccinations may be necessary. The holding bags are washed after each trapping trip and between animals that appear unhealthy (those with a heavy parasite load or that exhibit abnormal behaviour). Handling animals quietly, using sound methods, and with confidence, minimises the risk to both trappers and animals.

Checking all traps

Times of opening and closing are recorded (usually for the first trap of each line) so that trapping effort may be calculated, and every trap checked is noted on a data sheet, to prevent any risk of leaving an unchecked trap in the field. This process is even more important when several people assist with trap-checking or if assistants are new to the trapping procedure and layout of traps. Project teams are trained and the project supervisor ensures that every trap has been checked at each session. Whether trapping by night or day, the trap-checking schedule is rigorous, so that animals are not left in traps for more than one night or a few hours. Upon completion of the work, the traps are counted during and after collection or closed (pitfall traps) to ensure that no trap is left (open) in the field. Any trap left unattended is likely to cause the death of the trapped animal.

Safeguarding environmental integrity and data

Old bait is not discarded at the trapping site, and trappers 'clean up' the site after trapping by removing the traps and flagging tape if applicable, filling the holes used for pitfall traps, and replacing the soil and vegetation as far as practicable. Seeds are removed from clothing, shoes, camping and trapping equipment, and the

Table 1. Panel of experts who reviewed the draft document

Not all experts may have agreed with all statements presented in this article, although the agreement was generally very strong

Experts	Organisations
Dickman, Christopher	University of Sydney (NSW), Professor in Terrestrial Ecology. Many awards including C. Hart Merriam Award for outstanding research (American Society of Mammalogists); has been member of many committees including the IUCN Species Survival Commission (Australian marsupials and monotremes); numerous publications on Australian wildlife; supervision of many research students.
Gill, Sarah	Earlville Veterinary Surgery (Qld), veterinarian. Masters of Veterinary Studies in Conservation Medicine with experience working on various mammal research projects; was locum at the Australian Wildlife Health Centre at Healesville Sanctuary, Zoos Victoria; attends to injured and ill wildlife in private practice.
Lunney, Daniel	Office of Environment and Heritage (NSW); Senior Principal Research Scientist. Long-term experience in the ecology and conservation of forest fauna and human dimensions of wildlife management, including animal ethics; numerous publications on all these topics.
Medlin, Graham	South Australian Museum, Honorary Research Associate. Council Member, Field Naturalists Society of South Australia (SA); Editor, South Australian Naturalist; numerous mammal surveys and reports.
Pavey, Chris	CSIRO Ecosystem Sciences (Alice Springs, NT); Research Group Leader within Ecology Program. Numerous publications on Australian vertebrates and author of several national recovery plans; member of many committees; research student supervision.
Thompson, Graham	Terrestrial Ecosystems (WA), Principal Zoologist. Former academic position; vertebrate surveys and environmental impact assessments, management plans, translocations; numerous publications on Australian wildlife and survey methods; research student supervision.

equipment that has been in contact with soil also may need to be sprayed with methylated spirits to reduce the risk of spread of *Phytophthora* in certain areas. Traps that are removed from the site are washed using detergent such as Napisan (Reckitt Benckiser (Australia) Pty Ltd, West Ryde). It is important to collect rigorous data and to file the records safely, because trapping not only represents a lot of work, but also incurs stress to animals; the effort and impact on animals must not be wasted. When trapping in a public area, researchers ensure that the traps cannot be taken by the public, respect the public's sensibility, and sometimes need to explain the study's objectives (Animal Care and Use Committee, undated).

Training personnel/students

People involved in trapping are thoroughly trained in SOPs and the purpose of the project is explained to them; everyone works with an expert before trapping alone. Vogelnest and Woods (2008) provide information on handling different species. Small stuffed toy animals available at stores are very useful to train beginner trappers to remove animals from box traps; we have been using this method successfully for many years in the courses Ecology and Wildlife Ecology at the University of South Australia. We have also found that the methods need to be repeated again and again in the field; the knowledge and memory of trappers must be developed over many thousands of trap-nights/days of experience. Details such as removing the lid of a pitfall and not placing it upside down on the ground (where it will collect humidity, which will get into the trap when it is closed), removing the stick (see below), or thoroughly checking the sand for scorpions and centipedes are often worth mentioning at each trapping session. The checking of the traps is supervised by an experienced trapper at the beginning of each session when relatively inexperienced trappers are assisting. Training also covers the scientific and ethical outcomes of a study. AECs require information on the necessity of the proposed work and whether such studies have already been conducted. It is important to publish the trapping information in order to avoid repeating a study and to facilitate the improvement of future work, which may not happen if a study is undocumented.

Standard Operating Procedures: aluminium box traps

Choice of traps

Box traps include, but are not limited to, Elliott traps (Elliott Scientific, Upwey, Vic.), Sherman traps (Sherman Traps Inc., Tallahassee, FL), and Longworth traps (Longworth Scientific Instruments Co., Oxford, UK). Traps of a suitable size and design are chosen for the target animals (Freegard and Richter 2009*c*); trap design can affect capture success and animal survival (e.g. Jacob *et al*. 2002; Anthony *et al*. 2005). Most small terrestrial mammals that can be attracted with baits can be trapped in these types of traps, but Tasker and Dickman (2001) noted that certain animals (such as *Cercartetus nanus*, *Sminthopsis leucopus* and *Sminthopsis murina*) are more effectively trapped in pitfall traps. Traps that are too small for an animal may cause the skin of its tail to be stripped by the door as it tries to escape.

Trap placement and weather protection

Factors such as spacing, placement, habitat structure, social behaviour, and odour affect trapping success and are considered before the trapping program commences (e.g. Dickman and Woodside 1983; Read *et al*. 1988; Tasker and Dickman 2001; Cunningham *et al*. 2005), as are trap type and trap combination (Garden *et al*. 2007). Box traps in good working order are labelled with sequential numbers and are placed in a consistent fashion that allows the research objectives to be attained. They are typically arranged as parallel lines in the field, usually on flat areas on the ground so that they do not wobble and scare the animals (they can also be mounted on trees and placed at numbered sites). Ant mounds and other dangers (such as areas that may flood) are avoided. Traps are placed in order by number so that when they are checked or collected, any missed trap can be noticed immediately. The advantage of permanent grids or trap lines is that the numbers are already in place. Flagging is normally used to indicate the location of individual traps, and the start of each line is marked with a GPS; in some cases it is necessary to mark all traps and some landscape features in this fashion (G. Thompson, pers. comm.). The visibility of traps to humans can be a concern because traps are occasionally stolen (G. Thompson, pers. comm.).

If traps are open during the day, they are placed in the shade or under shade covers and checked sufficiently frequently so that the temperature does not become uncomfortable to any captured animal. Heat is often more of an issue than is cold, particularly in open areas. Cold temperatures (particularly at night) may require frequent checking of traps as well, shortened trapping times, or the cancellation of the trapping session. The definition of 'cold' varies depending on location and species; for example, 5°C may be considered cold on the coast but acceptable in the high country or tablelands (C. Dickman, pers. comm.). Insulation such as paper towels or cotton wool is ordinarily placed inside the traps to increase the comfort of the animal; however, such absorbent bedding may be inappropriate in wet conditions (DERM 2009; Animal Ethics Infolink, undated) and may influence capture rate (G. Thompson, pers. comm.). Green and Osborne (1981) used coconut fibre very successfully to provide insulation for trapped small mammals; the animals' feet are checked for fibre before release. Adding leaf litter from the vicinity of the trap is a suitable alternative since it does not introduce an alien substance to the trap (D. Lunney, pers. comm.). To protect the traps from rain, plastic sleeves are sometimes used, but they tend to increase condensation in the traps; a 10% slope for the traps allows drainage when it rains (DERM 2009; Animal Ethics Infolink, undated). If the traps are tilted, then the door can be the highest point so that the bait will stay in the back of the trap (G. Medlin, pers. comm.). Tilting the trap downwards can also be effective if the bait is wedged with the bedding at the back of the trap (C. Dickman, pers. comm.). Whatever method is used, the material in the trap must not prevent the mechanism from operating. Generally, little rain penetrates traps that are kept flat, particularly if they are covered in dense vegetation. A shelter above the traps may be necessary in heavy rain.

Bait

The bait consists of items that are not stale, for example peanut butter and oats, uncontaminated by pathogenic microorganisms (honey is not used by some agencies because it may contain such microorganisms), animals, or chemicals. Some baits (including honey) attract large numbers of ants (see below). Unused bait is refrigerated, kept in the shade, or discarded in a rubbish bag (not in the field) if it is rancid or soiled. Discarding bait in the field feeds animals unnecessarily and may attract predators, with possible repercussions on trapping rates and the safety of trapped animals. Bait that can germinate is rendered inert before use to minimise the risk of plant invasion. Bait needs to attract the target animals, but may also replace the food and energy that the animals would have been able to consume, had they not been captive (DERM 2009; Animal Ethics Infolink, undated), and needs to be biologically appropriate.

Trapping times

For night trapping, it is preferable to wait until after dark to open traps, especially when some bird species, such as Australian magpies, currawongs, and corvids are present, because they can take the bait and close the traps before dark, resulting in reduced trapping effort. This situation tends to occur in open habitats only. Australian ravens (in our experience) and currawongs may remove pins from traps, rendering them useless and/or releasing the animal inside. Placing a small piece of masking tape over the pull-out hook or placing the pin backwards and putting the back of the trap into a shrub helps to prevent the pin from being removed. C. Pavey (pers. comm.) has also observed Australian ravens taking animals out of traps early in the morning and uses ‘corvid-proof’ cages over traps on gibber plains. Foxes and a range of other animals can also set off the traps (G. Medlin, pers. comm.) or disturb them in other ways.

Traps are cleared at least twice a day when open continuously. During extremely hot or cold weather, traps are checked more often or closed. If weather conditions worsen during trapping, researchers close traps if it is safe for them to do so. Researchers consider closing traps in response to forecasts for inclement weather (for example, severe thunderstorms with strong wind and heavy rain or during bushfire alerts); this response is a precaution against unnecessary animal deaths, but also reduces risk to researchers.

Checking the traps

Traps are checked quietly. Open traps are preferably left untouched, but missing bait is replaced. Each trap with a closed door is examined quickly for its contents by opening the door slightly. The trapper keeps his/her face away from the door in case the trap contains a snake. Snakes or large animals may be removed by dislodging the pin that keeps the trap closed, while the trap is on the ground. The trap may be placed in a calico bag or sturdy container, before the pin is removed, if the animal is to be kept. If the trap contains a small mammal, it is turned upside down slowly, so that the trigger mechanism now lies on the roof of the trap and the animal rests on a flat surface. The trap is kept approximately horizontal, with the entrance facing away from the trapper. A calico bag is placed very tightly around the trap’s entrance and any loose section folded tightly against the trap, so that the animal does not escape through a gap. If the inner seams of the bag are loose and protrude, the bag is turned inside out, to prevent vertebrates’ claws from becoming tangled, which adds time to the handling of the animal and can result in injury (seams are better placed on the outside in any case). The trap is opened and at least one finger stays at all times on the door to keep it open, in case the mechanism suddenly releases the door. Holding the back of the trap against one’s stomach makes the manipulations easier. If the animal does not run into the bag (which is held horizontally and supported) on its own, one or two hard downward shakes of the trap with the open door facing down may be necessary. Another method is to blow lightly at the back of the trap – animals tend to fall into the bag (C. Dickman, pers. comm.); however, it may not be safe for a person to blow on the trap if pathogenic microorganisms are present. The trap is not shaken unnecessarily and care is taken not to let the bag hit against the ground when shaking the trap. If the animal does not fall out of the trap after one or two shakes, it is advisable to check that its tail or foot has not been caught inside the trap. When the animal falls into the calico bag, it is quickly prevented from running back into the trap by one hand closing the throat of the bag above the animal. The trap then may be shut until the next trapping session or rebaited and left open. Alternatives to this common method and restricted to experienced field researchers include the catching of a non-biting animal directly in the trap (but there is a chance that the animal will slip past the arm of the trapper), or shaking the trap gently with the door facing upwards, so that when the trap is inverted, the animal will tumble into the bag because it is braced in the wrong direction (D. Lunney, pers. comm.). Sturdy transparent plastic bags may also be used instead of calico bags; the advantage is that the animal can be identified and released promptly. This method may also facilitate the ‘scruffing’ of large rodents such as *Rattus* sp. If the animal is to be kept, capture details are recorded on the bag (for example, date, time, site, trap number, and species – we use masking tape on each bag) so that the animal can be returned to its correct location after processing and the correct data may be matched to each animal.

Traps between trapping sessions

Depending on conditions, it is sometimes preferable to place the closed traps on bushes to diminish infestation by ants until the next trapping session, because ants may be attracted by the bait. In any case, ants are monitored so that they do not affect trapped animals (see also section on pitfall trapping). Traps soiled with urine and faecal matter can affect the future capture of animals (positively or negatively) and may also introduce pathogens; such traps may have to be cleaned between trapping sessions (unless very lightly soiled). Traps are thoroughly washed and sterilised (e.g. with methylated spirits, dilute bleach, or a detergent such as Napisan (Reckitt Benckiser (Australia) Pty Ltd, West Ryde, NSW) after each field trip to decrease the risk of pathogen transfer; special care is taken in areas infected with *Phytophthora* or other pathogens, so that infection is not spread. Chapman *et al*. (2011) provide detailed information on how to clean traps, bags, and other equipment.

Standard Operating Procedures: cage trapping

Cage traps come in different sizes and are used similarly to box traps. Like box traps, cage traps are used to capture small mammals, including several rat species. The advantage of cage traps is that, covered with shelter appropriate for the location (e.g. canvas in case of rain or cold, hessian bags or shade cloth in some climates), they tend to keep the animals dry and make them less prone to extreme heat, whereas animals can get wet from condensation and their own urine in box traps and overheat when the temperature is high. Covers also prevent attacks from birds of prey (G. Medlin, pers. comm.). Covers are secured (for example with rocks or sand) so that an animal will not remove them (Freegard and Richter 2009*a*). The same precautions taken with box traps (placement, bait, time of use, data recording) are taken with cage traps. Traps are not lifted. A bag is positioned around the door of a small trap and the door is opened. The animal is encouraged to move into the bag by positioning one's body at the back of the cage and possibly blowing gently (Freegard and Richter 2009*a*). A large trap is tilted so that the door opens upward. The animal is then removed as if from a pitfall trap (see below). Traps, bags, and covers are cleaned and disinfected, as are other traps. Chapman *et al*. (2011) recommend washing cage traps in a 1% bleach solution.

Standard Operating Procedures: pitfall trapping

Setting pitfall traps

Pitfall traps are made of PVC pipe or rolled plastic sheets with a screen at the bottom, or consist of plastic or metal buckets; the effectiveness of different trap types varies for different species (Thompson and Thompson 2007). Traps are inserted into holes that allow their edge to be level with the ground; the lip of the bucket is not visible. Traps are generally positioned into lines and the drift net (usually held in place by pegs) passed over the centre of each pitfall trap, in a continuous line. For drift-net fencing, aluminium fly wire has the advantage of standing up without the need for many pegs; it is also very resistant to tearing (C. Dickman, pers. comm.). Plastic sheeting and shade cloth may also be used (G. Thompson, pers. comm.). A drift fence seems to have a positive impact on capture rates in most cases (e.g. Briese and Smith 1974; Friend *et al*. 1989; Moseby and Read 2001), but not for the western pygmy-possum (*Cercartetus concinnus*) (Pestell and Petit 2007). The depth and width of traps affect the taxa captured (Morton *et al*. 1988; Friend *et al*. 1989; Catling *et al*. 1997; Thompson *et al*. 2005; Pestell and Petit 2007) and can be chosen so as to minimise non-target captures. Funnels or cones can increase trapping success of some animals (hopping mice, for example) by preventing them from jumping out. Trap lines and individual traps are generally marked with flagging tape, their location recorded with a GPS, and numbers written in the traps for recording purposes. Reflective tape on trees or on flagging tape is very useful in locating traps at night, particularly when a drift fence is not used. If few trees or shrubs are available (as may be the case in some desert environments), reflective tape can be attached to bamboo poles or metal star droppers, positioned near trap lines.

Bucket-style traps are pierced with small holes to allow water drainage during rain. However, these holes are not always fully functional depending on the soil quality and the intensity of the rain, so traps are monitored when it rains. They tend not to drain well in clay-dominated soils. The holes in the base of pitfall traps are small enough so that small animals do not get trapped under the buckets or escape; they may be covered with fine mesh. Floating objects such as flat cork discs or Styrofoam 'meat' trays can be used, giving animals a refuge from accumulating water (DERM 2009; Animal Ethics Infolink, undated; Thompson and Thompson, undated), although gut obstruction could occur if the animals ingest Styrofoam (S. Gill, pers. comm.), so the risk is evaluated. In harsh environments, pitfall traps may also be accommodated with small roofs during inclement conditions to protect animals from sun or rain. However, shade covers may affect capture rates; regular trap checking is the best way to avoid temperature-related deaths (Hobbs and James 1999). Although checking traps at night is difficult, increased trap-checking frequency can decrease trap deaths dramatically. It should be noted, however, that some species are short-lived and an occasional trap death may not be due to poor technique (D. Lunney, pers. comm.). Ideally, traps are closed if rain becomes heavy or threatens to isolate the site. Wet rags can be provided in hot weather to cool the animals and the traps are checked frequently. The time at which traps are checked is very important to avoid lethal temperatures (Thompson and Thompson 2009).

Shelters provide some protection to trapped animals from inclement weather (e.g. Thompson and Thompson 2009). Shelters in the traps are often used by trapped animals (e.g. Pestell and Petit 2007) and may consist of PVC tubing, transparent plastic tubing, cardboard rolls, or natural materials. In our studies, we have found that the use of tubing saved a considerable amount of time because natural materials did not have to be found at the time of set up. Cardboard absorbs water and tends to promote fungal growth very quickly in humid conditions. Other artificial shelters can be cleaned and reused, and do not introduce invertebrates into the traps, as natural materials sometimes do. They may be stuffed with paper towelling at one end to provide insulation to the animals.

Ants may be present in vast numbers and cannot always be removed every time the traps are checked. Traps are kept closed if they are located in the vicinity of meat ants (*Iridomyrmex* spp.). Insecticide is discouraged because it can affect vertebrates and impacts on native fauna are poorly known (Story and Cox 2001; Khan and Law 2005), but it may be appropriate under particular conditions (DERM 2009). C. Pavey (pers. comm.) has used the pyrethroid Coopex® (Bayer Environmental Science, East Hawthorn, Vic.) along with manual removal at some sites to reduce ant infestation; however, pyrethroids can affect reptiles and amphibians (e.g. Khan 2003; Khan *et al*. 2003; Talent 2005). C. Dickman (pers. comm.) has found no evidence of ill effect of Coopex® on trapped frogs and reptiles when it was sprinkled lightly around the traps, but ants were effectively deterred. Data on invertebrates are often useful to characterise environmental conditions (e.g. indicator species) and food sources for small mammals. Preventing animals from eating each other in traps is difficult, particularly when carnivorous marsupials such as mulgaras (*Dasycercus* spp.) are present, since they will go into the traps deliberately to eat other animals (C. Dickman, pers. comm.). In other cases, the provision of broken-up egg cartons can help the animals to find shelter from each other. Venomous animals such as small snakes, scorpions, and centipedes may also be present; we have observed centipedes killing lizards in pitfall traps.

Trapping times

As with aluminium box traps, opening times are important. Traps are opened after dusk and normally checked before dawn when nocturnal animals are targeted; they are checked several times a day when targeting diurnal animals, unless an appropriate microclimate can be maintained in the trap (e.g. with shading). Predatory animals such as currawongs, magpies, goannas, snakes, dingoes, foxes, or cats take captured animals by following trap lines, so monitoring is necessary. This problem can be solved by the use of narrow and long pipe traps (16 × 60 cm: C. Dickman, pers. comm.), if such traps are suitable for the targeted animals. Plywood covers on short stilts can minimise the impact of large predators as well as hot weather (DERM 2009; Museum Victoria 2009; Animal Ethics Infolink, undated).

Checking the traps

After checking and removing the shelter, each trap is examined for the presence of animals. Head torches are used to check the bottom of the traps during the night; during the day, especially early morning, the shiny metal lids of the traps can be used as mirrors to shine light down the traps (C. Dickman, pers. comm.). Tongs can be used to remove shelters or debris safely from deep pitfall traps. If soil is placed at the bottom of traps, scorpions and small lizards often hide in it and are difficult to find, so the substrate is examined very carefully. Ladles, other scoops, and battery-powered insect aspirators can assist with the daily removal of invertebrates.

Mammals can be captured with gloved hands (gloves may reduce tactile sensitivity of the handler, however) and may be covered with a calico bag as they are being removed from a trap to minimise their stress and the risk of escape via the handler's arm. Mammals that are not processed immediately are placed in calico bags, which are labelled appropriately.

Traps between trapping sessions

When the traps are unused during trapping sessions, they are kept closed with well fitting lids that may be additionally secured with rocks or thick layers of soil. A stick placed in the trap and reaching the top of the trap may serve as an escape route if the lid is broken and a climbing animal trapped inadvertently. If the traps are not removed at the end of the trapping period, but are used regularly, a long stick is placed in the trap after it has been completely emptied of animals, the lid is secured tightly, rocks are placed on the trap, and dead branches are placed on the top if available. In addition, traps may be filled with soil or rocks. A voluminous cover of dead branches or other items deters large animals, such as emus and kangaroos, from walking on the lids. Plastic lids can break and tend to tear after long-term use in the field and so need to be protected from large animals; they can also be protected from sun damage by being covered with soil. Should the lid break, the soil can also be used as an escape ramp by any animal falling into the trap. Unused traps are marked with metal or wooden stakes high enough to remain visible among growing vegetation. A permanent pitfall trap line requires regular maintenance, especially if it has a permanent drift fence. As with box-style traps, detailed records ensure that every trap has been checked.

Standard Operating Procedures: recaptures

Minimising recaptures and weather extremes can reduce death rates during trapping (Lemckert *et al*. 2006). Because recaptured animals can be exposed to increased stress, trapping is ceased temporarily after periods of 3–4 days when recaptures are common (Tasker and Dickman 2001); this recommendation is confirmed by C. Pavey (pers. comm.), who has observed an increased number of deaths when animals get recaptured frequently. However, G. Thompson (pers. comm.) traps for longer periods because mammal recaptures are uncommon at his sites. Moseby and Read (2001) also noted that survey accuracy was greater with a larger number of trapping nights; rare species were often not detected in four nights. This period is too long for small lactating dasyurids if traps are not checked at night. A record of captured animals, carried in the field, can allow the researchers to release recaptured animals immediately if data from these individuals are not needed and the conditions are adequate. Anecdotally, releasing recaptured vertebrates on the side of the drift fence that is opposite to their original release location may prevent them from being captured for a third time (i.e. because they are not trying to get back through the fence to a shelter, burrow, or territory). To avoid 'trap-happy' animals (animals that choose to go in a trap again to eat the bait), it may be possible (depending on the project) to open only half of the traps at one time, using random numbers to determine which traps are to be open (D. Lunney, pers. comm.), or to close specific traps involved in retrapping. When an animal has been recaptured in the same trap within a session, closing this trap gives the animal some distance over which to forage, even if it is going to be recaptured in another trap (C. Dickman, pers. comm.). Impacts on trapping effort and survey implications are considered every time a trap is kept closed.

Standard Operating Procedures: animal handling and short-term housing

Mammals known to be lactating (and without young in the pouch for marsupials) are released at the time of capture if it is safe to do so. Checking the traps during times of reproduction generally takes place before daylight for nocturnal animals. In some cases animals can be released safely during the day, for example in dense forest, where animals may not be exposed to increased risk of predation as they would in an open habitat. Small mammal species are unlikely to eject pouch young, but certain species of the Macropodidae, Potoroidae and Peramelidae do (procedures for dealing with this situation are presented in Freegard and Richter 2009*b*). Mammals held in calico bags may be transported in those bags in a hard container (plastic aquaria with lids work well). Rodents may chew through the bag, so it is wise to transfer them to a hard carrying container as soon as possible. The top of the bag is typically twisted and folded over before being closed tightly with a strong string. This method ensures that the mammal does not risk sticking its head through a partially open top and suffocating. Torpid animals that cannot be watched or warmed until they are aroused are held until the following evening. They can be stored in a bag placed in a polystyrene cooler, where they can warm up slowly. Each calico bag is labelled with the trap number and time and date of capture. Record-keeping is facilitated by this method and animals are kept with their bags and released at the exact location of their capture, normally within 24 h, at a time when they are active. Bags are normally washed with detergent between individuals to ensure they are free of pathogens and parasites.

All materials are ready before an animal is handled, to minimise handling time and associated stress. The animal is weighed before being removed from the bag; the bag is weighed separately upon each release, in case debris or bait from the trap accompanies the animal. A small mammal may be held through the bag in one hand and removed from the bag with the other hand. Rats and other animals that can bite through the bag are better handled on a hard surface. They are restrained just enough so that they cannot move too much, and are 'scruffed' from as near as possible to the top of the head down to the back (plenty of skin is available to hold). The handling of the skin from the top of the head down the back is important because it does not allow the animal to turn around and bite or escape from the handler. Although this method may appear dramatic, many animals are safely handled in this fashion, with minimal risk of injury to the handlers and to the animals. Species and reproductive status can be determined very quickly when the animal is handled properly. 'Scruffing' is not necessary for all small mammals, but minimising movement and the risk of injury to animal and handler is important; appropriate handling methods for various animals are provided in Vogelnest and Woods (2008) and in Richter and Freegard (2009*b*). Curiously, in view of their success dispersing in many environments, house mice (*Mus musculus*) and native rodents such as Forrest's mice (*Leggadina forresti*) tend to be fragile in traps and during handling; they may go into shock and die if cold, hot, or if handled too tightly (Petit *et al*. 2012). Some mammals tend to struggle less if their eyes are covered (Gannon *et al*. 2007). Some rodents are not held by the tail because the skin may be stripped off (degloving) if the animal struggles to escape (Breed and Eden 2008). Handling cones are useful for larger mammals (see Koprowski 2002). Different species have different particularities and the scope of this paper is not to explore them. Informed, experienced, and calm handlers minimise stress to animals.

Mammals are measured, marked, sexed, and otherwise examined before being released at the place of their capture, or if it is not possible (for example, if it is daytime and the animal is nocturnal or if scats must be collected), they are placed back in their bag and in an appropriate holding cage. The bag is left open and water and appropriate food (e.g. sugar water with an adequate proportion and type of sugar for nectarivores) are provided; Vogelnest and Woods (2008) give information on the diet of several species of small mammals. Feeding takes place after scats are collected, if needed (unless it is sugar water). The holding cages are protected from direct sun, heat, drought, and cold. Researchers are responsible for minimising impacts on dependent offspring or social groups by reducing captive time (Animal Care and Use Committee, undated). The duration of captivity is minimised because it can lead animals to abandon their territories or home ranges, modify their foraging and their social structures (Canadian Council on Animal Care 2003). Captivity can also cause a considerable amount of stress to wild animals (Langkilde and Shine 2006), so it is preferable to release animals immediately after processing whenever possible.

Marking small mammals

Animals may be marked in several ways (Thompson and Thompson, undated). Temporary marking (e.g. shaving a small area, or marking the base of the tail or head with a permanent marker) is appropriate for short-term studies. For long-term studies, a permanent and unique mark is needed to identify each animal. Common permanent marking methods include PIT (Passive Integrated Transponder) tags for animals that are large enough and ear notching (Richter and Freegard 2009*c*). Ear tags are used on larger animals (Richter and Freegard 2009*d*), but are rarely appropriate for small animals. For fragile animals, we have also used a less invasive method involving tail tattooing with UV-fluorescent ink (Petit *et al*. 2012). Studies involving toe-clipping, tail-clipping, ear-notching, branding or other mutilation need to demonstrate to their ethics committee that there is no alternative (The Province of British Columbia 1998; Canadian Council on Animal Care 2003); in Australia, the October 2011 *Draft Code of Practice for the Care and Use of Animals for Scientific Purposes* states that toe-clipping may be used only if tissue samples are needed. Whether or not this statement appears in the next version of the Code, it indicates an intention to move away from mutilation as a method for marking animals. Reticence is felt by some people who have employed such methods for years because a new method could introduce error from loss or misinterpretation of marks; it is thus essential that any new method be tested to ensure that it achieves the desired results for the sake of long-term studies (D. Lunney, pers. comm.). Marking is done with clean/disinfected equipment in conditions that maximise speed and minimise stress to the animals. Sharp tools (e.g. scissors, needles) are used after disinfection with alcohol; numbing of the body part to be marked can be achieved with Xylocaine 2% jelly (lignocaine hydrochloride: AstraZeneca Pty Ltd, North Ryde, NSW), EMLA® dermal cream (lignocaine and prilocaine: AstraZeneca Pty Ltd, North Ryde, NSW), or other appropriate anaesthetic. All equipment is ready before handling the animals and the environment is quiet. White and Garrot (1990) recommended that radio-transmitters be restricted to 5% or less of the animal's body mass. Bradshaw and Bradshaw (2002) found that all three honey possums (*Tarsipes rostratus*) they had fitted with radio-transmitters that exceeded 10% of their mass died. Although mass is an important consideration, it is associated with other factors such as duration of the study and the methods of attachment. For example, some radio-transmitters are mounted so they can break away at the end of their useful life (Canadian Council on Animal Care 2003) (such as with an adhesive that deteriorates over time). The shape of the device, its position and method of attachment, the length of time of the study, and the ability to monitor the welfare of the animal are presented to an AEC along with the ratio of transmitter mass to animal mass (D. Lunney, pers. comm.).

Trapping effort

The time of opening and closing of traps is recorded for calculating trapping effort. The duration of 'trap time' may vary depending on season and environmental conditions. Trap-nights (the number of nights when one trap was open), for example, may be shorter in winter to minimise risk to the animals when the nights are too cold. Often, the quality of the data increases as the number of animals trapped increases (e.g. collection of scats for dietary studies), so it is important to maximise trapping effort by using good methods and monitoring the traps. The accurate recording of trapping effort is particularly important when trapping is conducted to evaluate species richness (e.g. Thompson *et al*. 2007) or estimate population characteristics. For example,

Table 2. Form for Standard Operating Procedures: trapping of small rodents and marsupials with aluminium box or pitfall traps
Sample data are in italics

	Aluminium box traps	Pitfall traps
Dates	*5–10 July 13*	*5–10 July 13*
No. of lines	*8*	*8*
No. of traps per line	*10*	*12*
No. of days open	*0*	*0*
No. of nights open	*4*	*4*
Trap checking schedule	*Midnight, 4 a.m.*	*5 a.m.*
Trap size	*A*	*40 L*
Trap type (e.g. bucket)	–	*Bucket (small holes at bottom)*
Permanent line (yes/no)	–	*Yes*
Permanently closed how?	–	*Stick, rocks on lid, veg.*
Drift fence	–	*Yes*
Bait	*Peanut butter and oats*	*No bait*
Shelter types	–	*PVC tubes*
Paper towel (yes/no)	*Yes*	*Yes*
Other insulation	*Square of woollen carpet*	*No*
Plastic sleeves	*No, weather monitored*	–
Covers over traps	*No, weather monitored*	*No, weather monitored*
Ant infestation	*Traps closed if too many ants*	*Traps closed if meat ants*
Animals processed	*Small mammals and reptiles*	*Western pygmy-possums only*
Mammal marking method	*Tail tattoo*	*Tail tattoo*
Reptile marking method	*Body tattoo*	*n.a.*
Voucher killing method	*n.a.*	*n.a.*
Holding time	*<5 min (released at capture)*	*<24 h*
Holding cage type	*n.a.*	*plastic aquaria*
Type of food provided (species)	*n.a.*	*Sugar water, 32° Brix (C. concinnus)*
Phytophthora spraying	*Yes*	*Yes*

Elliott traps that are found shut in the morning and do not contain an animal after being open overnight, are deducted from the total number of effective trap-nights. It is recommended that weather (e.g. temperature, humidity, rainfall, cloud cover) be recorded since it can affect trapping success (Read and Moseby 2001). It is important to understand that modifications to the trapping schedule are going to affect trapping effort, with implications for the results of the survey or research project if comparisons are going to be made over time. Thompson and Thompson (2010) discuss several factors that affect trapping effort.

Vouchers

If it is necessary to keep some animals as vouchers, or if an animal is seriously injured, suitable killing methods are used. Examples are provided in Owens (2000), Reilly (2001), AVMA (2007), Sharp and Saunders (2004), and Chapman *et al*. (2011). Students and researchers are trained (Canadian Council on Animal Care 2003) in appropriate methods and animals are killed away from other animals (Sikes *et al*. 2011). The local museum is contacted for information on appropriate preservation methods and what data must be recorded.

Conclusion

Capture is a stressful experience for most wild mammals, and it can affect their health. Under Australian law, an AEC is required to give approval for the use of animals in research and teaching. The purpose of this paper is to provide general SOPs for the trapping of small mammals to AECs, with enough flexibility to allow for differences in conditions among sites, species, and objectives of the study. It is the concern of the AECs to examine what the risks are to the animals and what precautions have been taken to minimise those risks. Ethics application forms cover many aspects of the research or teaching project, including housing of animals and number of animals. For what is relevant to trapping, we propose a trapping summary form that can be attached to these SOPs and replace part of the ethics application (Table 2), to indicate what methods presented in this paper are being used by people involved in trapping small rodents and marsupials for a particular project. This paper may be updated as new information becomes available. Ideally, there should be no injury or trap death during trapping; injuries and deaths must be reported and methods need to be adjusted to reduce the risks (Canadian Council on Animal Care 2003). Little information is available on trap death statistics in Australia. We suggest that the *Australian Journal of Zoology* could be an excellent medium to report findings relevant to animal ethics. Important information in this context, and too brief to compose a full article, could be published as a short communication. Information improving ethical standards would then be available quickly to all readers, for the benefit of animals. Improved standards for animal welfare should always be sought (Iossa *et al*. 2007).

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References

Animal Care and Use Committee (undated). Guidelines for the capture, handling, and care of mammals as approved by the American Society of Mammalogists. Available at: http://www.mammalsociety.org/committees/commanimalcareuse/98acucguidelines.pdf (accessed 5 January 2011).

Animal Ethics Infolink (undated). Wildlife surveys. Industry and Investment NWS, Animal Research Review Panel Guideline 10. Available at: http://www.animalethics.org.au/policies-and-guidelines/wildlife-research/wildlife-surveys (accessed 5 January 2011).

Anthony, N. M., Ribic, C. A., Bautz, R., and Garland, T Jr. (2005). Comparative effectiveness of Longworth and Sherman live traps. *Wildlife Society Bulletin* **33**, 1018–1026. doi:10.2193/0091-7648(2005)33[1018:CEOLAS]2.0.CO;2

Australian Government (2004). 'Australian Code of Practice for the Care and Use of Animals for Scientific Purposes.' 7th edn. (National Health and Medical Research Council, Commonwealth of Australia: Canberra.)

Australian Government (2012). 'A Guide to the Use of Australian Native Mammals in Biomedical Research – Public Consultation Draft.' (National Health and Medical Research Council, Commonwealth of Australia: Canberra.)

AVMA (2007). Guidelines on euthanasia. American Veterinary Association. Available at: http://www.avma.org/issues/animal_welfare/euthanasia.pdf (accessed 5 January 2011).

Bradshaw, S. D., and Bradshaw, F. J. (2002). Short-term movements and habitat use of the marsupial honey possum (*Tarsipes rostratus*). *Journal of Zoology* **258**, 343–348. doi:10.1017/S0952836902001486

Breed, A., and Eden, P. (2008). Rodents. In 'Medicine of Australian Mammals'. (Eds L. Vogelnest and R. Woods.) pp. 503–526. (CSIRO Publishing: Melbourne.)

Briese, L. A., and Smith, M. H. (1974). Seasonal abundance and movement of nine species of small mammals. *Journal of Mammalogy* **55**, 615–629. doi:10.2307/1379550

Canadian Council on Animal Care (2003). 'Guidelines on: the Care and Use of Wildlife.' (Canadian Council on Animal Care: Ottawa.)

Catling, P. C., Burt, R. J., and Kooyman, R. (1997). A comparison of techniques used in a survey of the ground-dwelling and aboreal mammals in forests in north-eastern New South Wales. *Wildlife Research* **24**, 417–432. doi:10.1071/WR96073

Chapman, T., Sims, C., and Mawson, P. (2011). 'Minimising Disease Risk in Wildlife Management. Standard Operating Procedures for Fauna Translocation, Monitoring and Euthanasia in the Field.' 3rd edn. (Department of Environment and Conservation: Perth.)

Cunningham, R. B., Lindenmayer, D. B., MacGregor, C., Barry, S., and Welsh, A. (2005). Effects of trap position, trap history, microhabitat and season on capture probabilities of small mammals in a wet eucalypt forest. *Wildlife Research* **32**, 657–671. doi:10.1071/WR04069

DERM (2009). 'Standard Operating Procedure – Wildlife Surveys.' (Department of Environment and Resource Management: Brisbane.)

Dickman, C. R., and Woodside, D. P. (1983). A test of a competition model with reference to three species of small mammals in south-eastern Australia. *Oecologia* **60**, 127–134. doi:10.1007/BF00379332

Freegard, C., and Richter, V. (2009a). Cage traps for live capture of terrestrial vertebrates. Standard Operating Procedure No. 9.2. Department of Environment and Conservation, Perth.

Freegard, C., and Richter, V. (2009b). Care of evicted pouch young. Standard Operating Procedure No. 14.1. Department of Environment and Conservation, Perth.

Freegard, C., and Richter, V. (2009c). Elliott traps for live capture of terrestrial vertebrates. Standard Operating Procedure No. 9.1. Department of Environment and Conservation, Perth.

Friend, G. R., Smith, G. T., Mitchell, D. S., and Dickman, C. R. (1989). Influence of pitfall and drift fence design on capture rates of small vertebrates in semi-arid habitats of Western Australia. *Australian Wildlife Research* **16**, 1–10. doi:10.1071/WR9890001

Gannon, W. L., and Sikes, R. S. Animal Care and Use Committee of the American Society of Mammalogists (2007). Guidelines of the American Society of Mammalogists for the use of wild animals in research. *Journal of Mammalogy* **88**, 809–823. doi:10.1644/06-MAMM-F-185R1.1

Garden, J. G., McAlpine, C. A., Possingham, H. P., and Jones, D. N. (2007). Using multiple survey methods to detect terrestrial reptiles and mammals: what are the most successful and cost-efficient combinations? *Wildlife Research* **34**, 218–227. doi:10.1071/WR06111

Green, K., and Osborne, W. S. (1981). The diet of foxes *Vulpes vulpes* (L.), in relation to abundance of prey above the winter snowline in New South Wales. *Australian Wildlife Research* **8**, 349–360. doi:10.1071/WR9810349

Hobbs, T. J., and James, C. D. (1999). Influence of shade covers on pitfall trap temperatures and capture success of reptiles and small mammals in arid Australia. *Wildlife Research* **26**, 341–349. doi:10.1071/WR98032

Iossa, G., Soulsbury, C. D., and Harris, S. (2007). Mammal trapping: a review of animal welfare standards of killing and restraining traps. *Animal Welfare (South Mimms, England)* **16**, 335–352.

Jacob, J., Ylönen, H., and Hodkinson, C. G. (2002). Trapping efficiency of Ugglan traps and Longworth traps for house mice in south-eastern Australia. *Wildlife Research* **29**, 101–103. doi:10.1071/WR01048

Khan, M. Z. (2003). Effect of agricultural chemicals on reptiles: comparison of pyrethroid and organophosphate with phytopesticide on cholinesterase activity. *Pakistan Journal of Biological Sciences* **6**, 821–825. doi:10.3923/pjbs.2003.821.825

Khan, M. Z., and Law, F. C. P. (2005). Adverse effects of pesticides and related chemicals on enzyme and hormone systems of fish, amphibians and reptiles: a review. *Proceedings of the Pakistan Academy of Sciences* **42**, 315–323.

Khan, M. Z., Tabassum, R., Naqvi, S. N. H., Shah, E. Z., Tabassum, F., Ahmad, I., Fatima, F., and Khan, M. F. (2003). Effect of cypermethrin and permethrin on cholinesterase activity and protein contents in *Rana tigrina* (Amphibia). *Turkish Journal of Zoology* **27**, 243–246.

Koprowski, J. L. (2002). Handling tree squirrels with a safe and efficient restraint. *Wildlife Society Bulletin* **30**, 101–103.

Langkilde, T., and Shine, R. (2006). How much stress do researchers inflict on their study animals? A case study using a scincid lizard, *Eulamprus heatwolei*. *The Journal of Experimental Biology* **209**, 1035–1043. doi:10.1242/jeb.02112

Lemckert, F., Brassil, T., Kavanagh, R., and Law, B. (2006). Trapping small mammals for research and management: how many die and why? *Australian Mammalogy* **28**, 201–207. doi:10.1071/AM06028

Lunney, D. (2012). Ethics and Australian mammalogy: reflections on 15 years (1991–2006) on an Animal Ethics Committee. *Australian Mammalogy* **34**, 1–17. doi:10.1071/AM10010

Mccallum, J. (2012). Changing use of camera traps in mammalian field research: habitats, taxa and study types. *Mammal Review* doi:10.1111/j.1365-2907.2012.00216.xIn press

Morton, S. R., Gillam, M. W., Jones, K. R., and Fleming, M. R. (1988). Relative efficiency of different pit-trap systems for sampling reptiles in spinifex grasslands. *Australian Wildlife Research* **15**, 571–577. doi:10.1071/WR9880571

Moseby, K. E., and Read, J. L. (2001). Factors affecting pitfall capture rates of small ground vertebrates in arid South Australia. II. Optimum pitfall trapping effort. *Wildlife Research* **28**, 61–71. doi:10.1071/WR99058

Museum Victoria (2009). Manual of standard operating procedures (SOPs) for the ethical use of live terrestrial vertebrate animals by staff and associates of Museum Victoria. Available at: http://museumvictoria.com.au/pages/17688/museumvictoriaanimalethicscommitteesop-terrestrialvertebrates-vn11-12july2011.pdf?epslanguage=en (accessed 19 April 2012).

Owens, H. (ed.) (2000). 'Guidelines for Vertebrate Surveys in South Australia using the Biological Survey of South Australia.' (Department for Environment and Heritage: Adelaide.)

Pestell, A. J. L., and Petit, S. (2007). Methods and ethical considerations of pitfall trapping for the western pygmy possum (*Cercartetus concinnus* Gould) (Marsupialia: Burramyidae), with observations on capture patterns and nest sites. *Wildlife Research* **34**, 296–305. doi:10.1071/WR06090

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. doi:10.1071/ZO11088

Read, J. L., and Moseby, K. E. (2001). Factors affecting pitfall capture rates of small ground vertebrates in arid South Australia. I. The influence of weather and moon phase on capture rates of reptiles. *Wildlife Research* **28**, 53–60. doi:10.1071/WR99057

Read, V. T., Malafani, K. W., and Myers, K. (1988). A comparison of grid and index-line trapping methods for small mammal surveys. *Australian Wildlife Research* **15**, 673–687. doi:10.1071/WR9880673

Reilly, J. S., ed. (2001). .Euthanasia of Animals used for Scientific Purposes.' 2nd edn. ANZCCART. Available at: http://www.adelaide.edu.au/ANZCCART/publications/Euthanasia.pdf (accessed 5 January 2011).

Richter, V., and Freegard, C. (2009*a*). Dry pitfall trapping for vertebrates and invertebrates. Standard Operating Procedure No. 9.3. Department of Environment and Conservation, Perth.

Richter, V., and Freegard, C. (2009*b*). Hand restraint of wildlife. Standard Operating Procedure No. 10.2. Department of Environment and Conservation, Perth.

Richter, V., and Freegard, C. (2009*c*). Permanent marking of mammals using ear notching. Standard Operating Procedure No. 12.2. Department of Environment and Conservation, Perth.

Richter, V., and Freegard, C. (2009*d*). Semi-permanent marking of mammals using ear tags. Standard Operating Procedure No. 12.3. Department of Environment and Conservation, Perth.

Sharp, T., and Saunders, G. (2004). GEN001 methods of euthanasia. New South Wales Department of Primary Industries. Available at: http://www.environment.gov.au/biodiversity/invasive/publications/pubs/gen001-methods-euthanasia.pdf (accessed 5 January 2011).

Sikes, R. S., and Gannon, W. L. Animal Care and Use Committee of the American Society of Mammalogists(2011). Guidelines of the American Society of Mammalogists for the use of wild animals in research. *Journal of Mammalogy* **92**, 235–253. doi:10.1644/10-MAMM-F-355.1

Story, P., and Cox, M. (2001). Review of the effects of organophosphorus and carbamate insecticides on vertebrates. Are there implications for locust management in Australia? *Wildlife Research* **28**, 179–193. doi:10.1071/WR99060

Talent, L. G. (2005). Effect of temperature on toxicity of a natural pyrethrin pesticide to green anole lizards (*Anolis carolinensis*). *Environmental Toxicology* **24**, 3113–3116. doi:10.1897/05-053R.1

Tasker, E. M., and Dickman, C. R. (2001). A review of Elliott trapping methods for small mammals in Australia. *Australian Mammalogy* **23**, 77–87. doi:10.1071/AM01077

The Province of British Columbia (1998). Live animal capture and handling guidelines for wild mammals, birds, amphibians and reptiles. Standards for Components of British Columbia's Biodiversity No. 3. Resources Inventory Committee, Vancouver.

Thompson, G., and Thompson, S. (2007). Usefulness of funnel traps in catching small reptiles and mammals, with comments on the effectiveness of the alternatives. *Wildlife Research* **34**, 491–497. doi:10.1071/WR06081

Thompson, G., and Thompson, S. (2009). Comparative temperature in funnel and pit traps. *Australian Journal of Zoology* **57**, 311–316. doi:10.1071/ZO09052

Thompson, S. A., and Thompson, G. G. (2010). Terrestrial vertebrate fauna assessments for ecological impact assessment. Terrestrial Ecosystems, Perth.

Thompson, G., and Thompson, S. (undated). General terrestrial fauna surveys protocol. Available at: http://www.ecu.edu.au/GPPS/ethics/assets/General_Terrestrial_Fauna_Surveys_Protocol.pdf (accessed 19 April 2012).

Thompson, S. A., Thompson, G. G., and Withers, P. C. (2005). Influence of pit-trap type on the interpretation of fauna diversity. *Wildlife Research* **32**, 131–137. doi:10.1071/WR03117

Thompson, G. G., Thompson, S. A., Withers, P. C., and Fraser, J. (2007). Determining adequate trapping effort and species richness using species accumulation curves for environmental impact assessments. *Austral Ecology* **32**, 560–580.

Vogelnest, L. and Woods, R. (2008). 'Medicine of Australian Mammals.' (CSIRO Publishing: Melbourne.)

White, G. C., and Garrot, R. A. (1990). 'Analysis of Wildlife Radio-tracking Data.' (Academic Press: San Diego.)

Handling Editor: Paul Cooper