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A GUIDE TO THE CARE AND USE OF AUSTRALIAN NATIVE MAMMALS IN RESEARCH AND TEACHING

2014

| WORKING TO BUILD A HEALTHY AUSTRALIA |

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Glossary of terms and phrases

Altricial: young at an immature stage of development (helpless, naked and blind).

Back-young: refers to the offspring of marsupials at a stage of development prior to independence, when they are still being carried on their mothers back.

Capture myopathy: see Appendix IV.

Degloving: where the skin, including the fur (for example, on the tail) is stripped off from the underlying tissue.

Diapause: a state of ‘dormancy’ or suspended animation of the life cycle. As applied to mammals, it is a state of arrested development or dormancy of the blastocyst during early embryonic development. Occurs in marsupials, in particular macropodids, seals, bats and rodents.

Expert: person with extensive knowledge and experience relevant to the species being used, and with the procedure being undertaken with that species (for example, care and management, breeding, anaesthesia, blood collection). Experts may include zoo or wildlife veterinarians, or a wildlife expert with experience with a particular species.

Folivore/folivorous: species that feed primarily on foliage such as leaves of trees and shrubs, as opposed to species that include some foliage in a broader herbivorous diet.

Hibernation (natural hypothermia): a drop in the internal body temperature (cooling) of an animal below its normal operating (active) temperature that is a natural part of the ecophysiology of the animal. Under normal field conditions, species undergoing natural hypothermia are able to spontaneously rewarm and resume normal activity.

Hyperthermia: a rise in the internal body temperature of an animal beyond its normal operating temperature, that is, overheating. Prolonged hyperthermia cannot be tolerated and will result in extreme physiological distress and can result in death.

Hypothermia: a drop in the internal body temperature of an animal below its normal operating (active) temperature, that is, cooling. Prolonged hypothermia cannot be tolerated by most species and will result in loss of physiological function and can result in death, as the animal is unable to spontaneously rewarm.

However note that some species of mammal undergo natural hypothermia during torpor/hibernation.

IC: an intracardiac injection directly into the heart.

IH: an intrahepatic injection directly into the liver.

IM: an intramuscular injection into a muscle.

IP: an intraperitoneal injection directly into the peritoneal (body) cavity.

IV: an intravenous injection directly into a vein.

Morphometrics: typically measurements associated with body morphology, such as body weight, the length of the body, head and feet. This type of data is used for determining growth and condition.

Pes: hind foot.

PIT tag: passive integrated transponder that is inserted subcutaneously or into a body cavity of an animal and provides lifetime identification of the animal.

PO: *per os* indicates by mouth.

Pouch-young: refers to the offspring of marsupials at a stage of development prior to independence when they are still being carried in their mother’s pouch.

SC: a subcutaneous injection just beneath the skin. A subcutaneous injection is delivered just under the skin but not into or through the muscle below.

Sex determination: various ways in which the sex of an animal is determined. For example, sex is genetically determined in mammals by the chromosomes.

Social insects: insects such as ants, termites and bees that live in a colony, usually associated with a nest. Animals that specialise in feeding on social insects are called myrmecophages, and for these animals, social insects make up more than 75% of the diet.

Taxon: members of a particular taxonomic group, for example, a particular species, genus or family.

Torpor/hibernation (natural hypothermia): a drop in the internal body temperature or cooling of an animal below its normal operating (active) temperature that is a natural part of the ecophysiology of the animal. Under normal field conditions, species undergoing natural hypothermia are able to spontaneously rewarm and resume normal activity.

Zoonoses: pertaining to diseases or pathogens that can be transmitted from animals to humans.

Purpose of the guide

The aims of this document are to:

- promote a high standard of care in the use of Australian native mammals in scientific research and teaching
- provide general information and a network of expert contacts to assist those working with native species
- promote the accumulation of reliable scientific knowledge about Australian native mammals
- promote the use of that knowledge for the welfare, conservation and management of native mammal species.

This guide describes current knowledge sourced from experts in the particular taxa. A science-based document such as this will require updating to incorporate scientific advances in the field. Those who have not previously or recently worked with a particular taxon must seek expert advice from knowledgeable sources before obtaining animals or commencing research.

The use of native mammal species in Australia is governed by Commonwealth, state and territory legislation and supported in particular by the *Australian code for the care and use of animals for scientific purposes* (8th edition, 2013) (the Code; and subsequent iterations) and the *Guidelines to promote the wellbeing of animals used for scientific purposes : The assessment and alleviation of pain and distress in research animals 2008* (and subsequent iterations) that, along with this guide, cover all non-human vertebrate animal species used in research and teaching. Any person intending to use native mammals for scientific purposes must read the relevant legislation, guidelines and policies (see Appendices VII and VIII) in conjunction with this document. Those working with native species must be aware that legislation varies between states and territories.

The Code is variously enacted in all state and territory legislation. The Code provides an ethical framework and governing principles to guide decisions and actions of all those involved in the care and use of animals for scientific purposes. The Code details the responsibilities of investigators animal carers, institutions, animal ethics committees and all people involved in the care and use of animals. The Code also describes processes for accountability.

An obligation to respect animals underpins the Code. This obligation brings with it a responsibility to ensure that the care and use of animals for scientific purposes is ethically acceptable, balancing whether the potential effects on the wellbeing of the animals involved are justified by the potential benefits to humans, animals or the environment.

The use of animals for scientific purposes:

- must have scientific or educational merit
- must aim to benefit humans, animals or the environment
- must be conducted with integrity.

When animals are used, the number of animals involved must be minimised, the wellbeing of the animals must be supported, and harm, including pain and distress, in those animals must be avoided or minimised.

Contributors to this guide are listed in Appendix VI. A list of relevant government departments is present in Appendix IX. Relevant institutions and organisations can be found in Appendix X. Experts and institutions with relevant expertise are listed in the sections for each species. A list of experts who are not included in the sections for specific species can be found in Appendix XI.

This document is a guideline and is not intended to serve as an exhaustive source of information on the care and use of native mammals in research. All researchers must ensure that their knowledge of the species they intend to use for scientific purposes is as complete as possible. All researchers must also ensure they have training in the use of anaesthetic and other drugs and equipment for the species they intend to use.

General considerations

The biology of native mammal species is enormously variable and basic characteristics such as diet, physiology, reproductive mode, sociality, habitat requirements are highly diverse, and for some species, not well known. Thus, it is extremely important not to make assumptions about these when conducting research. Native mammal taxa have species-specific capture, transport, handling, housing and care (both husbandry and veterinary) requirements that differ from purpose-bred laboratory and domestic animals.

While some native mammals habituate well to captivity others do not, and wild animals may be highly stressed by simple procedures such as capture and handling. In addition, many research projects using native mammals are conducted on wild populations. Such studies impose a substantially greater responsibility on the researcher, and require experience with, and knowledge of, the chosen study species. Furthermore, researchers must be vigilant about animal welfare issues that may arise under field operating conditions. All proposals for research using native mammals must be approved by the animal ethics committee with which the project and the researcher(s) are associated.

When conducting research on live animals from or in the wild researchers must consider:

- replacement of animals with other methods
- reduction in the number of animals used
- refinement of techniques used, to minimise the adverse impact on individual animals and populations
- minimising impacts on non-target species or individuals.

General requirements and comments

Detailed requirements are provided for each taxon in its respective section. Researchers must ensure adequate housing, care and access to expert veterinary advice. Species-specific considerations for safe capture, handling, transportation, nutrition, shelter, behaviour, health, disease control and breeding, for example, must be addressed.

Different species respond differently to anaesthesia, analgesia and handling. Researchers must consult the specific sections for the relevant species or taxon. During a restraint procedure (either chemical or physical) particular attention must be given to species-specific risks, such as capture myopathy, shock, hypothermia and hyperthermia, respiratory failure, cardiac failure, trauma and secondary infection. In addition, during field research requiring sedation or anaesthesia it is imperative that subsequent release of animals back into the wild is conducted under appropriate protocols to avoid potential risks, such as impaired ability to climb competently and exposure to predators.

Suitability for research and teaching

Scientific issues

Scientific research using Australian native mammals is essential to our understanding of their biology. Successful strategies for the conservation and management of native mammals will rely on a thorough knowledge of their biology. Research on Australian native mammals also reveals some of the basic principles of mammalian biology and may advance biomedical science. Australia's native mammals have some special features that can aid research. For example, marsupials produce small neonatal young that are born at a very early stage of development (altricial young) and are readily accessible in the pouch.

Native mammal research has made many contributions to science including improving understanding of:

- determination of information critical to the conservation and management of individual species
- the genetic control of sex determination and the hormonal control of sexual differentiation and descent of the testes
- the control of embryonic diapause

- the influence of lactation on mammalian reproduction
- the mode of action of hormones in inducing gene expression for milk protein synthesis, using the particular properties of the marsupial mammary glands
- the differentiation and development of the nervous system, with potential for understanding and subsequently alleviating central nervous system disorders in humans and other animals
- improved methods of control of breeding in seasonally responsive species through understanding how photoperiod information is interpreted in the brain and transformed into hormonal responses
- evolution of endothermy in vertebrates
- understanding parasites, pathogens and disease for conservation outcomes as well as alleviating human morbidity, controlling human diseases and understanding zoonoses
- understanding of respiratory biology using marine mammals
- assisted reproduction and contraception of endangered and overabundant species.

Recognising that wild native species will often be less adaptable to captivity than domesticated or laboratory animals, wherever possible, native mammals should be replaced with traditional laboratory species if they will fulfil the research requirements for laboratory-based research.

Alternatively, researchers may be able to source native mammals from an established captive breeding colony. If free-living animals are being captured or killed for another reason, secondary use (such as acquiring tissues) is strongly encouraged. If possible, conduct research using native mammal tissues acquired from other projects or tissue banks, rather than acquiring additional animals.

The fate of animals at the completion of the research is determined by the relevant government agencies and conditions imposed by animal ethics committees. It is essential that the welfare, disease and quarantine issues inherent in the re-release of wild animals are considered after any substantial period of captivity. In particular, some native species may experience high mortality subsequent to release. Release of animals from captivity into the wild and translocation of wild animals also pose a risk of introducing pathogens or parasites into the recipient population. There are similar risks for the animals being released or translocated if they have not previously been exposed to the pathogens or parasites in the recipient population.

Sources

Before obtaining native mammals, researchers must be aware of the conservation status of a species under state, territory and/or federal legislation. Native mammals can be sourced from the wild but must not be taken from natural habitats unless animals bred in captivity and held in research units, zoos, museums, university departments and similar organisations are unavailable or unsuitable for the scientific purpose.

To determine a source of native mammals for research purposes:

- contact the state or territory wildlife authorities for information on native mammal species being studied by researchers or displayed by exhibitors
- contact the major institutions conducting native mammal research in the state or territory
- contact the major societies involved with native mammals, for example, the Australian Mammal Society, the Australasian Wildlife Management Society, the Australian Wildlife Diseases Association (see Appendix X).

Subclass Prototheria: Monotremes

Echidna

The short-beaked echidna is one of two extant species, along with the platypus, in the order Monotremata (egg-laying mammals) in Australia. The echidna is highly specialised and difficult to maintain in captivity. This species is very susceptible to heat stress, and extra care should be taken to avoid this. Details of the basic biology of the echidna are provided in Augee (2008).

Echidna – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Short-beaked echidna, <i>Tachyglossus aculeatus</i> .	Widespread and common.	Throughout Australia.	Social insects, including ants and termites, with more limited consumption (varying locally) of other invertebrates such as soil-dwelling beetle larvae and worms.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999 - List of Threatened Fauna*

It is very important to note that the body temperature of echidnas is lower than that of most other mammals (25 °C to 32 °C). They are unable to thermoregulate effectively at ambient temperatures above 32°C, thus handling should be avoided in hotter conditions. In the wild, echidnas minimise activity during hot weather and seek cool shelter sites or burrows. Echidnas also undergo hibernation during the colder months.

Capture, handling, marking for identification, transport

Echidnas are very strong, and wild animals will often dig in rapidly when disturbed. They can be dug up using the hands, but the use of shovels should be avoided because they may cause injuries. Echidnas can be caught by firmly grasping one ankle and sliding the other hand beneath their body and then lifting them off the ground while supporting them. Alternatively, an echidna can be caught by placing one hand on either side of the head, effectively under the shoulders, and rolling the animal backwards (towards the handler) then lifting as described above. During handling, rest the echidna across the handler's arm (or leg if in the squatting position) to support its body weight. Captive echidnas can be picked up (if the base of the pen is hard) by simply placing the hands under either side of the body.

Leather gloves or gauntlets are not always particularly useful for catching echidnas as they reduce handling dexterity; however light gloves (for example, MSA Flexi Fit, FullBoar Safety Pty Ltd, Australia) offer good protection against the spines for catching and during general handling.

Animals can be individually marked. Four or five coloured plastic beads or tubing can be attached by slipping the beads or tubing onto the echidna's spines and securing them with superglue. Echidnas should be held and transported individually in a sturdy crate (with no gaps, as they will tear the crate apart), plastic garbage bin or plastic stacker box. Stacker boxes must be at least 0.6 metres high and packed into the vehicle so that they cannot be tipped over. Crates, bins or boxes used for holding and transport should contain leaf litter, shredded paper or peat. At environmental temperatures of 30 °C or above, echidnas must be transported in a cool environment or an air-conditioned vehicle (at temperatures well below 30 °C).

Captive husbandry

Echidnas are extremely strong for their size and very determined, thus cages must be very robust to contain them. Straight-sided, smooth-walled pens (4 metres × 4 metres and at least 0.9 metres in height) constructed of sheet metal are appropriate for short-term holding for research, with an extra 2 metres × 2 metres for each additional animal. If cages are built from wire mesh, the lower part of the walls (to about 0.75 metres) should be covered with sheet metal, as echidnas can climb up, but cannot climb down and will fall and potentially injure themselves, especially on concrete floors.

For pens with earth floors, wire mesh (which needs to be very heavy gauge to prevent them from tearing it apart) should be buried to a depth of at least 50 cm below floor level to prevent them from digging out. The floors of pens, especially if concrete, should be covered with leaf litter, sand or peat to a depth of at least 20 cm to avoid abrasion of the feet and to allow some digging activity. Because echidnas are sensitive to high ambient temperatures, housing should be designed to accommodate this: internal holding pens should be air conditioned, and outdoor pens should include access to deep soil for burrowing, shade, and cold water for bathing or a sprinkler (see Jackson 2003). Echidnas will die if exposed to ambient temperatures of 38 °C or more. Provision of hollow logs, or purpose-built burrow boxes, with an internal dimension of more than 30 cm are recommended for use as shelters.

Diets for captive echidnas

Diets for captive echidnas have been specially developed and refined by zoological institutions that routinely hold these animals. Captive diets vary somewhat in different institutions. Appropriate zoos should therefore be consulted about diets before bringing animals into captivity. Some institutions (for example, Perth Zoo) use canned cat food as the main portion of the diet. Other institutions use high-quality minced beef or pollard and meat meal. Vitamins (for example, Pentavite, special echidna mix, omnivore pellets and, if available, live termites) should be included. In order to avoid spillage and food contamination, food bowls should be secured.

Routine sample/data collection

Body weight, sex (note: echidna testes are inside the body cavity) and presence of a spur are routine data to collect from echidnas.

Blood samples can only be taken from sedated or anaesthetised echidnas (see the 'Anaesthesia, analgesia and sedation' section below). Up to 0.5 mL can be readily drawn from the rostral sinus, on the dorsal surface of the beak, directly posterior to the nostrils, using a 26-gauge needle or butterfly catheter. The jugular vein is an alternative blood-sampling site for experienced people.

Faecal samples can often be collected directly from wild animals, because when echidnas are picked up they commonly projectile defecate (however, successful collection requires rapid and skilful placement of a standard 50 mL sample vial).

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Echidnas can be sedated using 1 mg/kg diazepam or anaesthetised with Zoletil® (tiletamine/zolazepam) using 3–10 mg/kg, delivered IM into the hind leg muscle. However, the best method, if possible, is gaseous anaesthesia in an induction chamber using isoflurane in oxygen (5% for induction, 1–2% for maintenance) with an oxygen flow rate of 1 L/minute. Echidnas have a resting respiratory rate of about 60 breaths/minute, and satisfactory anaesthesia for blood sampling is achieved when the respiratory rate is approximately one breath/10 seconds, which appears to be very slow, but is normal. However, it is very important to monitor the time period of anaesthesia and record the respiratory frequency. The slow respiratory rate means that induction is slow and may take 7–10 minutes.

Echidnas cannot be intubated (Middleton 2008). Immobilisation procedures should not be conducted in hot weather unless the animals can be kept cool. There have been no pharmacokinetic studies on any analgesics for this species, thus if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Echidnas can be humanely killed using an overdose of sodium pentobarbitone (150 mg/kg) injected IP or IC. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

While echidnas carry a range of external parasites (for example, ticks and fleas), internal parasites (for example, cestodes) and pathogens (for example, *Toxoplasma gondii*), diseases do not appear to be a major problem in wild animals. Parasites and pathogens in echidnas are extensively documented by Middleton (2008) and Ladds (2009).

Specific breeding requirements

Echidnas are difficult to breed in captivity, with few births ever recorded until recently. Perth Zoo has now bred a number of echidnas in captivity. Information on captive breeding is provided in Ferguson and Turner (2013).

Institutions and individuals with specialised expertise

Institutions

Dreamworld Gold Coast, Queensland Phone: (07) 5588 1111 Internet: http://www.dreamworld.com.au	Perth Zoo Perth, Western Australia Phone: (08) 9474 0444 Email: email@perthzoo.wa.gov.au Internet: http://www.perthzoo.com.au
Taronga Zoo Sydney, New South Wales Phone: (02) 9969 2777 Email: tz@zoo.nsw.gov.au Internet: http://taronga.org.au/taronga-zoo	Zoos Victoria Veterinary Departments: Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville

Individuals

Dr Stephen Johnston (reproductive biology) Senior Lecturer in Wildlife Reproduction Wildlife Science Unit School of Agriculture and Food Science University of Queensland Phone: 0408 280 963 Email: s.johnston1@uq.edu.au	Dr Peggy Rismiller (ecology) Pelican Lagoon Wildlife Centre Kangaroo Island, South Australia Email: echidna@kin.net.au Internet: http://www.echidna.edu.au/index.html
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Bibliography for echidnas

Augee, ML 2008, 'Echidnas', in S Van Dyck & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 36–9.

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Ferguson, A & Turner, B 2013, 'Reproductive parameters and behaviour of captive short-beaked echidna (*Tachyglossus aculeatus acanthion*) at Perth Zoo', *Australian Mammalogy*, vol. 35, pp. 84–92.

Griffiths, ME 1978, *The biology of the monotremes*, Academic Press, New York.

Griffiths, ME 1968, *Echidnas*, Pergamon Press, Oxford.

Jackson, S 2003, 'Echidnas', in S Jackson (ed.) *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 33–51.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Middleton, D 2008, 'Echidnas', in L Vogelneust & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 77–101.

Temple-Smith, PD & Grant, TR 2002, 'Uncertain breeding: a short history of reproduction in monotremes', *Reproduction, Fertility and Development*, vol. 13, pp. 487–97.

Platypus

The platypus is one of two only extant species, along with the echidna, in the order Monotremata (egg-laying mammals) in Australia. The platypus, in particular, is difficult to maintain in captivity. This species is very susceptible to heat stress, and extra care should be taken to avoid this. Information on the basic biology of the platypus is provided in Carrick, Temple-Smith and Grant (2008) and Grant (2007).

Platypus – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Platypus, <i>Ornithorhynchus anatinus</i> .	Widespread and common.	Eastern Australia in permanent freshwater habitats, including streams and lakes (will extend into estuaries) from north Qld to Vic., Tas. and on Kangaroo Island, SA.	Mainly aquatic invertebrates.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Platypuses are one of the most difficult species to maintain in captivity, because they are highly susceptible to pronounced (life-threatening) physiological stress (Handasyde, McDonald & Evans 2003) unless managed very carefully and with a high degree of knowledge and expertise. Any person intending to conduct research on this species must seek expert advice, particularly to conduct captive studies. Furthermore, establishing animals in captivity must not be attempted without a firm commitment to invest in the time, labour, facilities and money required for success.

It is very important to note that the body temperature of platypuses is lower than that of most other mammals (mean: 32 °C, range: 29 °C to 34 °C) (Grant & Dawson 1978). They are unable to thermoregulate effectively at ambient temperatures above about 30 °C (or even less if agitated) and handling should be avoided under such conditions. They should not be kept at temperatures higher than 25 °C. While an ambient temperature of 25 °C is within the thermoneutral ('comfort') zone of the species, a platypus with a body temperature of 25 °C is probably hypothermic. In the wild, platypuses are mainly nocturnal, sleeping in earth burrows during the day, which helps them to avoid heat stress. During their activity period at night, they occupy aquatic habitats.

Capture, handling, marking for identification, transport

Platypuses can be caught from the wild in fyke nets or mesh nets.

Fyke nets (designed for catching eels and fish) have two wings leading to an entrance that opens into a series of cone-shaped, non-return chambers or funnels. These nets are best set in shallow water and the base and wings must be securely weighted to the streambed so that animals cannot squeeze under them, and instead are directed to the net entrance. The cod end of the net should be very securely staked above water level, so that air spaces exist in each chamber of the net and that captured animals can breathe. Fyke nets should be checked at 1- to 4-hour intervals and must be removed immediately from the stream if a rise in water level is anticipated. Platypuses can become progressively hypothermic if confined in cold water and may become moribund if confined for long periods, particularly if their fur is disturbed such that the insulating layer of air is compromised. It is particularly important to check nets every hour in very cold conditions, and netting in winter should be avoided if possible.

Platypuses can also be captured using unweighted or lightly-weighted mesh ('gill') nets (with a mesh of 48–49 mm, flattened knot to knot). This design ensures that animals are able to swim to the surface quickly to breathe, as long as nets do not become snagged on submerged debris or obstructions or weighed down by debris or gilled large and/or schooling fish. Mesh nets must be kept under regular surveillance so that animals can be removed promptly to prevent them from drowning (platypuses are captured in mesh nets by becoming entangled, thus may tire when attempting to maintain their position at the surface). To confirm the entire length of the net is hanging freely and is not snagged, the entire length of the net should be lifted up by hand from the water at least every hour or more frequently depending on the numbers of fish or the amount of debris present at the netting site.

The use of mesh nets at sites where large numbers of fish occur should be avoided if possible. The presence of large numbers of fish can increase the probability of platypuses drowning and the activity involved in clearing fish from the net can reduce platypus capture rates. High mortality of native fish species can also occur.

Because platypuses are largely nocturnal, nets are best set in late afternoon and must be closely monitored from the bank, by briefly using a spotlight and/or night scope every 10–15 minutes. When an animal is captured it should be disentangled quickly from the net and placed in a hessian or cloth sack (inside these their fur should dry rapidly). The animals should then be transferred almost immediately into a dry cotton or calico bag (for example, a flannelette pillowcase).

To remove a platypus from a net or to transfer one from containers such as bags and pens, grasp and hold the animal firmly by the terminal half of the tail. The tail is the site of fat storage, therefore holding the tail firmly but gently for the minimum time required, with fingers flat in the shape of a paddle, is not painful or harmful to the animal. In addition, holding a platypus by the end of the tail means that the handler will avoid being spurred by adult males, which carry venomous spurs on the inner side of the hind ankle. Males sometimes attempt to spur quite aggressively, particularly in the breeding season (special care should be taken between late June–October). Handling time should always be as brief as possible, and wherever possible, the platypus should be held in a bag so that the eyes are covered during conscious procedures. This will minimise the amount of direct physical restraint required and the exposure of the platypus to stressful stimuli.

After capture, wild platypuses should be released back to the wild, at the site of capture, as soon as possible (ideally within two hours of being removed from a net). They should ideally be held at ambient temperatures below 25 °C and should never be held at temperatures exceeding 30 °C. They can be held safely in securely tied bags, in a quiet, dark location, such as a holding crate or inside a vehicle. During this time each animal should be checked at least every hour to ensure that it is quiet and comfortable. Any animal showing signs of agitation (continuous scratching, failure to settle) or discomfort should be given priority for processing and/or immediate release. Special attention should be given to any animal showing lethargy after capture. A small number of individuals showing this behaviour have suddenly died of causes not yet identified in detailed post-mortem examinations. Any animals showing lethargy within the first 30 minutes to 1 hour of capture should be released immediately.

Animals can be individually marked permanently using microchips inserted under the skin between the scapulae, or for short-term purposes (lasting several months, depending on the stage of moult) by clipping distinctive patterns in the fur on the end of their tail. Radio tracking can be successfully conducted on platypuses. A small area of fur on the dorsal surface of the rump (the size of a 10-cent piece) is shaved bare and a small radio transmitter attached to the skin surface using tissue cement. Before deploying radio transmitters, expert advice should be sought on the appropriate size and shape and on safe, effective attachment procedures.

Platypuses should be held and transported individually in secure crates (ideally while also held inside a light cloth bag). It is particularly important that they are transported at a temperature of less than 25 °C. Transport should be as short as possible, ideally requiring animals to be held less than 4 hours from the time that they are retrieved from a net. Information on methods for transport of platypuses is provided in Jackson, Serena & Middleton (2003). Expert veterinary advice may also be sought (e.g. experienced wildlife veterinarian).

Captive husbandry

Platypuses require specialised captive facilities, and expert advice on husbandry and facility design must be sought before embarking on captive studies. Platypuses must be provided with a tank, as they feed exclusively in water. Tanks should have a minimum area of 6 m² with a ramp providing access to a tunnel leading to a nest box with absorbent bedding such as peat (Jackson, Serena & Middleton 2003). Platypuses are surprisingly strong for their body size and they are also quite good climbers. In addition, they are very determined and will escape from pens that are not sufficiently well constructed.

Platypuses can rapidly die if exposed to ambient temperatures above 30 °C. Because of their sensitivity to high ambient temperatures, the air and water temperature in their housing facilities should be kept below 25 °C. If the temperature of the water in their tank rises above this temperature on hot days, ice can be added to reduce the temperature.

Unless it is essential to answer specific research questions (for example, on reproduction), adult platypuses should not be brought into captivity in the breeding season, as during this period they exhibit elevated levels of stress hormones and are more susceptible to mortality (see Handasyde, McDonald & Evans 2003). Also, in the breeding period, males are more aggressive and females may be carrying eggs or have dependent young left in nesting burrows. Where possible, juvenile platypuses should be selected to bring into captivity. At this stage many would be dispersing from their natal area, and are probably more likely to adapt to captive conditions. Furthermore, this avoids the removal of established breeding adults from wild populations.

Diets for captive platypuses

In the wild, platypuses feed in water with their eyes closed, using electrical and mechanical receptors in their bill to locate prey such as freshwater crayfish and other aquatic invertebrates. Their diet in captivity should include a high proportion of appropriate live aquatic prey. This is particularly important in the initial stages of captivity, as it may take animals some time after arrival to adjust to feeding on dead prey and some individual animals are likely to always demand that live prey be provided. Platypuses consume large amounts of food for their body size, thus zoos with experience in platypus maintenance must be consulted about appropriate diet and volume of food before bringing animals into captivity. Further information on diets for captive platypuses is provided in Jackson, Serena & Middleton (2003).

Monitoring the health of captive animals is very important, because their health can decline rapidly. Expert advice should always be sought. Information is also provided in Booth & Connolly (2008), Jackson, Serena & Middleton (2003). Animals in poor condition are especially at risk. Platypuses can vary substantially in body size or weight in different stream systems even on a local scale, thus weight alone is not a good indicator of condition. The amount of fat stored in the tail is a useful way to monitor body condition (see Booth & Connolly 2008).

Routine sample/data collection

Body weight, tail fat index, body length (bill tip to tail tip), bill width and length, sex, spur morphology (see Grant 2007) and evidence of moult are routine data to collect from platypuses.

Blood samples of 1.0 mL can be taken from the rostral sinus, at the tip of the bill, using a 25 - or 26-gauge needle in conscious animals. Animals are best restrained wrapped inside a soft cotton bag, with a hole in one corner just large enough for the bill, but not the whole head, to protrude. The animal should be pressed firmly down onto a thick foam pad by one handler to restrain it (platypuses are very strong and difficult to hold still otherwise) while the blood sample is drawn.

Faecal samples can often be collected directly from wild animals because when they are picked up, platypuses commonly projectile defecate (however successful collection requires rapid and skilful placement of a standard 50 mL sample vial).

DNA can be obtained from the follicles of tail hairs, which are easily plucked out using small forceps. Tissue samples for DNA can be taken from the large area of webbing on one of the front feet or from the smaller areas of webbing on the rear feet. To do this, a small area of the leading edge of the webbing is selected and gently cleaned with a 70% alcohol swab. A few drops of bupivacaine local anaesthetic (0.05% aqueous solution) should be applied at the site to control any pain response. Using sharp scissors, a small piece of web (2 mm²) can be removed. Post-sampling bleeding is rare and there is usually little, if any, response from the animal.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Platypuses can be sedated using diazepam (0.5–1 mg/kg, IM). Gaseous anaesthesia can be achieved using isoflurane in oxygen (5% for induction, 2% for maintenance) with an oxygen flow rate of 1 L/minute and a typical induction time of 1–2 minutes. Further information on anaesthesia of the platypus is provided in Booth & Connolly (2008). Immobilisation procedures should not be conducted in hot weather unless the animals can be kept cool, and such procedures are not recommended in the field. There have been no pharmacokinetic studies on any analgesics for this species, thus if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Platypuses can be humanely killed using an overdose of sodium pentobarbitone (150 mg/kg) injected IP or IC. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior sedation (e.g. using diazepam) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

The major health issue for platypuses is their very substantial, often life-threatening, physiological stress response (see Handasyde, McDonald & Evans 2003). This is of particular importance for management of captive platypuses, but should also be taken into account during field research, for example when deciding how long animals can be held before release. One clear indication that a platypus is experiencing an extreme stress response is the failure of the animal's fur to dry out rapidly after removal from the water. In the event of any concerns over the health of animals being used in research, seek immediate expert veterinary advice (e.g. zoo or wildlife veterinarian).

While platypuses carry a range of external parasites (for example, ticks), internal parasites, and pathogens (for example, *Toxoplasma gondii*), diseases do not appear to be a major problem in wild animals. However, large ulcers resulting from *Mucour amphibiorum* infection occur in animals from a number of Tasmanian populations. Disease and health issues for platypuses are extensively documented in Booth & Connolly (2008), Jackson, Serena & Middleton (2003) and Ladds (2009).

Specific breeding requirements

Platypuses are difficult to breed in captivity; however, Taronga Zoo in New South Wales and Healesville Sanctuary in Victoria have successfully bred platypuses a number of times.

Institutions and individuals with specialised expertise

Institutions

<p>Taronga Zoo Sydney, New South Wales</p> <p>Phone: (02) 9969 2777 Email: tz@zoo.nsw.gov.au Internet: http://taronga.org.au/taronga-zoo</p>	<p>Zoos Victoria Veterinary Departments:</p> <p>Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au</p> <p>Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville</p>
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Individuals

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<p>Dr Melody Serena and Mr Geoff Williams (ecology, management) Australian Platypus Conservancy</p> <p>Phone: (03) 5157 5568 Email: platypus.apc@westnet.com.au Internet: http://www.platypus.asn.au</p>	<p>Associate Professor Peter Temple-Smith Department of Obstetrics and Gynaecology Southern Clinical School Monash University, Clayton, Victoria</p> <p>Email: peter.temple-smith@monash.edu.au</p>

Bibliography for platypuses

- Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.
- Booth, R & Connolly, J 2008, 'Platypus', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 103–32.
- Carrick, FN, Grant, TR & Temple-Smith, PD 2008, 'Family Ornithorhynchidae: Platypus'. in S Van Dyck & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 32–5.
- Grant, T 2007, *Platypus*, 4th edn, CSIRO Publishing, Melbourne.
- Grant, TR & Dawson, TJ 1978, 'Temperature regulation in the platypus, *Ornithorhynchus anatinus*, maintenance of body temperature in air and water', *Physiological Zoology*, vol. 51, pp. 1–6.
- Grant, TR 1983, 'Body temperature of free-ranging platypuses, *Ornithorhynchus anatinus*, with observations on their use of burrows', *Australian Journal of Zoology*, vol. 31, pp.117–22.
- Grant, TR & Temple-Smith, PD 1998, 'Growth of nestling and juvenile platypuses (*Ornithorhynchus anatinus*)', *Australian Mammalogy*, vol. 20, pp. 221–30.
- Grant, TR & Temple-Smith, PD 1998, 'Field biology of the platypus: current and historical perspectives', *Philosophical Transactions of the Royal Society B: Biological Sciences*, vol. 353, pp. 1081–90.
- Grant, TR & Temple-Smith, PD 1983, 'Size, seasonal weight change and growth in platypuses, *Ornithorhynchus anatinus* (Monotremata: Ornithorhynchidae), from rivers and lakes of New South Wales', *Australian Mammalogy*, vol. 6, pp. 51–60.
- Griffiths, ME 1978, *The biology of the monotremes*, Academic Press, New York.
- Grigg, G, Beard, L, Grant, T & Augee, M 1992, 'Body temperature and diurnal activity patterns in the platypus (*Ornithorhynchus anatinus*) during winter', *Australian Journal of Zoology*, vol. 40, pp. 135–42.
- Handasyde, KA, McDonald, IR & Evans, BK 2003, 'Plasma glucocorticoid concentrations in free-ranging platypuses (*Ornithorhynchus anatinus*): response to capture and patterns in relation to reproduction', *Comparative Biochemistry and Physiology – Part A: Molecular & Integrative Physiology*, vol. 136, pp. 895–902.
- Jackson, S, Serena, M & Middleton, D 2003, 'Platypus', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 1–32.
- Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.
- Temple-Smith, PD & Grant TR 2002, 'Uncertain breeding: a short history of reproduction in monotremes', *Reproduction, Fertility and Development*, vol. 13, pp. 487–97.

Subclass Theria: Marsupials

Dasyuridae

The order Dasyuromorphia represents the carnivorous marsupials. It is divided into three families, Thylacinidae that contains the extinct thylacine, Myrmecobiidae that contains the numbat (see section on the ‘Numbat’) and Dasyuridae that contains the remaining 66 species. This group includes the Tasmanian devil, quolls, antechinuses, dunnarts, planigales, phascogales and the kowari.

The following table shows species that are commonly worked with in research.

Dasyuridae – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Tasmanian devil, <i>Sarcophilus harrisii</i> .	Endangered.	Tas.	Primarily macropods, possums, wombats and carrion.
Spotted-tailed quoll, <i>Dasyurus maculatus</i> .	Endangered.	East coast of Australia from Far North Qld to Tas.	Invertebrates, reptiles, birds, mammals and carrion.
Western quoll, <i>Dasyurus geoffroii</i> .	Vulnerable.	South-western WA.	Small mammals, lizards, frogs, insects, and freshwater crustaceans.
Northern quoll, <i>Dasyurus hallucatus</i> .	Endangered.	Multiple separate populations spread across northern Australia from the Pilbara in WA to southern Qld.	Omnivorous diet including invertebrates, fruit (especially the wild grape, <i>Ampelocissus asetosa</i>), small vertebrates, bird eggs and nectar.
Eastern quoll, <i>Dasyurus viverrinus</i> .	Not listed.	South-eastern Australia, common in Tas., extinct on mainland.	Small mammals, birds, lizards, insects and fruit (for example, blackberries).
Red-tailed phascogale, <i>Phascogale calura</i> .	Endangered.	South-western WA.	Insects, spiders, small birds and mammals.
Brush-tailed phascogales, <i>Phascogale tapoatafa</i> and <i>P. pirata</i> .	Not listed.	Widespread, along east coast of Australia from far North Qld to Vic and SA, south-western and north-western WA Australia.	Invertebrates, small birds and mammals.
<i>Antechinus</i> spp.	Some widespread, others with limited distributions.	Throughout Australia or localised distribution, depending on species.	Generally feed on invertebrates.
Dibbler, <i>Parantechinus apicalis</i> .	Endangered.	Fitzgerald River National Park, Stirling Range National Park, Peniup Reserve, Boullanger Island, Whitlock Island and Escape Island in WA.	Insects and nectar.
Carpentarian pseudantechinus, <i>Pseudantechinus mimulus</i> .	Vulnerable.	Sir Edward Pellew Group of islands in the Gulf of Carpentaria, NT; Mt Isa, Qld.	Invertebrates.
Crest-tailed mulgara, <i>Dasyercus cristicauda</i> .	Endangered.	Simpson, Tanami and Strzelecki Deserts, WA rangelands.	Invertebrates, lizards and small mammals.
Kowari, <i>Dasyuroides byrnei</i> .	Vulnerable.	South-western Qld and northern SA.	Insects, arthropods and small vertebrates.
Fat-tailed dunnart, <i>Sminthopsis crassicaudata</i> .	Not listed.	Southern and Central Australia.	Insects and other invertebrates.

Dasyuridae – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Kangaroo Island dunnart, <i>Sminthopsis aitkeni</i> .	Endangered.	Kangaroo Island, SA.	Spiders, ants, beetles and scorpions.
Julia Creek dunnart, <i>Sminthopsis douglasi</i> .	Endangered.	North-west of Queensland.	Insects and other arthropods.
Butler's dunnart, <i>Sminthopsis butleri</i> .	Vulnerable.	Bathurst and Melville Islands, NT.	Invertebrates.
Kultarr, <i>Antechinomys laniger</i> .	Not listed.	Southern and central Australian arid zones.	Insects and small mammals.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Small dasyurids can be caught by hand from their nest box or with Elliott or pitfall traps. In the field, environmental thermal extremes must be taken into consideration. The number of traps set should always be limited to ensure that they can be cleared in a timely manner, so that animals do not remain in traps for extended periods and become hyperthermic or hypothermic. In extremely cold weather, especially when heavy rain or snow is forecast or during extremely hot weather, traps should be closed, as small animals can succumb rapidly. At sites where field work is conducted frequently, some species (such as *Antechinus* spp.) may become habituated to entering traps. If individuals are captured on two consecutive nights, it is recommended that traps in the vicinity be closed on the following night. This allows the animals time to forage, especially during periods when females are carrying large pouch-young and are under high lactational load or during periods of extreme weather conditions such as very hot, very dry, very wet or cold conditions.

Elliott traps constructed of light aluminium sheet are baited with peanut butter and rolled oats and additional components, such as golden syrup can be added to the baits. The traps are set one to two hours before dark and cleared commencing after dawn on the following morning. Elliott traps must be covered with plastic bags to provide protection from rain. In moderate to cooler areas, traps must also contain bedding material to keep animals warm. A handful of futon filling, **not** traditional shredded paper, is recommended because once wet, paper does not keep animals warm. This is essential because the very small body size of many dasyurids means that they become chilled easily when sitting overnight on the metal base of the trap. If an animal is hypothermic, place it into a light cotton bag and warm it with your hands or against your body, but handle the animal very gently and avoid excess movement. In hotter weather, Elliott traps should be placed to ensure they will not be exposed to the sun.

Pitfall trapping is the most common trapping method for small dasyurids in more arid habitats. Pitfall traps are made of polyvinylchloride (PVC) pipe of approximately 20–30 cm diameter and up to 60 cm in length, with wire mesh at the bottom. Alternatively, 10 L plastic or tin buckets with drainage holes drilled in the base can be used. These are sunk into the ground so the top of the trap is flush with the surrounding ground. Pitfall traps should contain some sand or soil, leaf litter and small sticks or bark for shelter and protection from predators. A fence line (approximately 30 cm high) of fine mesh is placed vertically across the opening of the trap to channel animals into it. Depending on habitat and target species, long drift fences (tens of metres) with numerous pitfalls along their length, or individual traps with approximately 2.5 metres of drift fence either side of the trap are used. No bait is required. Traps need to be deep, otherwise animals will escape.

Animals can be removed from Elliott or pitfall traps by reaching in with a bare hand and either grasping the scruff of the neck (using the little finger to hold the base of the tail, as close to the rump as possible with the tail held against the palm of the hand) or cupping the animal in the hand (again using the little finger to hold the base of the tail, as close to the rump as possible with the tail held against the palm of the hand), and transferring it into a bag. Alternatively, the operator's hand can be covered with a calico bag and the animal grasped through the bag. The animal can then be removed from the trap and the bag inverted to contain the animal. It is important not to clasp too firmly, as suffocation is possible. Small dasyurids, such as antechinus,

dunnarts and phascogales should never be restrained by the tail alone, as severe degloving injuries (where the skin is stripped from the tail) and tail fractures can occur.

For most procedures in the field, including general handling and weighing, animals should be placed into individual soft cotton bags. Physical restraint is stressful and should only be used for minor procedures such as marking, measuring, checking pouch-young or to induce general anaesthesia.

In captivity, larger dasyurids such as quolls and the Tasmanian devil, can be caught in a net or grasped and restrained by the base of the tail (close to the rump), then lowered into a bag. They should not be carried long distances while being held only by the base of the tail. It should be noted that some quolls are agile enough to turn and potentially bite the handler. In the wild, they can be captured in larger wire mesh cage traps that are covered to provide protection from adverse weather conditions. Meat, such as chicken, is generally used for bait; however, advice should be sought from experts for individual species.

Bags are useful to transport dasyurids for short distances. These bags range from calico bags or pillow cases for smaller animals up to large hessian sacks for Tasmanian devils. When transported in this manner, Tasmanian Devils must be closely monitored to ensure that they do not chew through the hessian bag.

Animals can be marked for identification using ear punches, tattoos or microchips.

Captive husbandry

Because Tasmanian devils and quolls will climb, a smooth metal skirt around the enclosure and at least 1.2 metres high is necessary to stop them escaping. Small dasyurids can be maintained in plastic tubs such as standard laboratory mouse cages or glass-fronted enclosures made of solid wood at least 10 mm thick to deter chewing. Wood or metal nest boxes filled with shredded paper, paperbark, seagrass or non-toxic wood shavings should be provided. Arboreal dasyurids such as antechinus and phascogales require roofed enclosures with a maximum gap in the wire of 2–10 mm, depending on the species.

Because the optimum temperature range is 15 °C to 30 °C, supplemental heating may be necessary. Dasyurids do not hibernate but during colder weather and in the arid zone, many undergo shallow periods of daily torpor where the body temperature drops to varying degrees, depending on species (Geiser 2008, Körtner & Geiser 2011). Torpor bouts can last up to 19 hours but usually extend from 2–8 hours. Enclosures should be spot cleaned daily and small enclosures should receive a full substrate change at least weekly.

Diets for captive dasyurids

In captivity small dasyurids such as planigales, dunnarts, antechinus, phascogales and kowaris have been successfully maintained on a mixed diet containing dog chow, hard boiled eggs, cheese, day old chicks and mice. This can be supplemented with assorted invertebrates such as mealworms, earthworms, fly pupae, crickets and moths. A commercial insectivore mix can also be used, and pollen grains and blossoms can be fed when available. Tasmanian devils and quolls are fed dog chow and whole animals, such as guinea pigs, rabbits, rats, mice, pilchards and day old chicks. Quolls can also be supplemented with mixed invertebrates and may enjoy a small amount of soft fruit such as bananas, grapes and melon.

Routine sample/data collection

Standard data collection should include sex, body mass, head length, hind foot length and, if possible, the sex, head length and number of pouch-young.

Anaesthesia is usually required for successful blood collection in dasyurids. Blood sampling techniques require significant skill and appropriate training. The ventral coccygeal, femoral, medial metatarsal, cephalic and jugular veins are all suitable sample sites. No more than 10% of total blood volume (approximately 1% of body weight) should be collected at any one time.

- Access to the ventral coccygeal vein is by inserting the needle perpendicular to the tail, in the ventral midline, and advancing it until the vertebrae are reached. Withdraw the needle slightly and blood should enter the needle hub. This vein is useful for smaller dasyurids.

- Access to the femoral vein or artery is gained by directing the needle at the pulse felt in the groin. Arterial blood is often obtained and digital pressure is required to prevent the formation of a haematoma.
- The medial metatarsal vein is a small vein running along the inside of the hind leg.
- The cephalic vein is present on the dorsal surface of either foreleg. This vein is useful for gaining blood samples from Tasmanian devils and quolls.
- The jugular vein is suitable for blood sampling in all dasyurids.

Blood collection from the orbital sinus is not acceptable because of the adverse impacts of this procedure on animal wellbeing. Any proposal to collect blood from the orbital sinus would require specific justification to the animal ethics committee.

Anaesthesia, analgesia and sedation

Sedation and anaesthesia should only be performed by trained personnel. Sedation for transport is generally not required but very nervous animals may benefit from the administration of 1–2 mg/kg diazepam given IM. Effects should be obvious after approximately 20 minutes.

Gaseous anaesthesia using isoflurane in oxygen (5% for induction) at a flow rate of 200 mL/kg/minute with a minimum of 1 L/minute is the method of choice. Induction is via a mask placed over the face, either directly or through the bag, or by placing the animal in an induction box. Maintenance generally requires 2% isoflurane in oxygen, but this varies between species and individuals.

If gaseous anaesthesia is not possible, injectable agents can be used. Zoletil® (tiletamine/zolazepam) can be given at 10 mg/kg, IM.

While no pharmacokinetic or efficacy studies on the use of analgesics in dasyurids have been published, butorphanol (0.1 mg/kg), buprenorphine (0.01 mg/kg) and meloxicam (0.2mg/kg) have been used with no negative side effects.

Humane killing

Conscious dasyurids can be killed humanely by carbon dioxide inhalation. If the animal is already anaesthetised, then it is acceptable to administer an overdose of sodium pentobarbitone (150 mg/kg) injected IV, IP or IC. Methods that are acceptable, with reservation, in anaesthetised dasyurids are cervical dislocation or delivery of a physical blow to the skull.

In general, carbon monoxide, freezing or hypothermia or thoracic compression are not acceptable. However, in very small, early stage pouch-young, killing by hypothermia may be acceptable (see information on hypothermia of pouch-young under 'Anaesthesia, analgesia and sedation' for macropodids in this guide).

Health issues, disease control and zoonoses

A range of disease conditions have been reported for dasyurids (see Holz 2008, Ladds 2009), including mycobacteriosis, toxoplasmosis, ectoparasites and internal parasites. All dasyurids are commonly affected by a wide range of tumours, including the devil facial tumour in Tasmanian devils. Aging quolls and Tasmanian devils frequently suffer from intervertebral disc disease, which results in paralysis.

Several diseases that affect dasyurids are also potential zoonoses (see Appendix IV).

Specific breeding requirements

Many dasyurids have a very short, highly-synchronous breeding season, sometimes only a few weeks long. This imposes a very narrow window of opportunity for research on the reproductive biology of these species. Synchronous mating and, in general, complete post-mating male mortality occurs in the antechinus, phascogales and the kultarr with some mortality reported in mulgaras, northern quolls, dibblers, and ningauis.

Dasyurids are polyovular and most are monoestrous, except quolls, kultarrs, kowaris, dunnarts, ningauis, and planigales, which are polyoestrous. The oestrous cycle ranges from 24 days in the fat-tailed dunnart up to 60 days in the kowari. Oestrous usually lasts 2–3 days but can persist for up to 14 days in the brown antechinus. Ovulation is spontaneous. Gestation length ranges from 13–31 days. Post-partum oestrous and ovulation are suppressed during lactation. Females will mate with more than one male, multiple paternities having been demonstrated in the agile antechinus and brush-tailed phascogale. Mating lasts from 1 hour in Tasmanian devils up to 12 hours in the agile antechinus.

Male and female dasyurids are generally maintained separately outside the breeding season. Smaller dasyurids, such as kowaris, gain weight as oestrous approaches. The pouch skin of quolls, antechinus and planigales becomes moist and reddened and the vulva oedematous. The Tasmanian devil pouch becomes deep, wet and glandular. The cells of the urogenital sinus become predominantly cornified. These can be detected by inserting a swab into the urogenital sinus. In species too small to swab, urine is usually released when the animal is picked up. These samples can be examined microscopically. A predominance of cornified epithelial cells is indicative of oestrous in quolls, dibblers, kowaris, phascogales and Tasmanian devils. Successful mating can be determined by the presence of sperm in the urine of the female.

A variety of strategies have been used to successfully breed dasyurids in captivity. A male quoll placed adjacent to the female's enclosure will elicit a clucking response from the female if she is in oestrous. He can then be released into her enclosure. Breeding kowaris have been maintained successfully in pairs, trios, (one male and two females) and larger groups. Males often need to be removed following the birth of young because of aggression from the females. A phascogale male can be placed together with a female and left until pouch-young are observed. Alternatively, a round robin system can be used, with males rotating among females on a biweekly basis. Tasmanian devils should be paired at weaning. If this is not possible, individuals should be gradually introduced with visual and olfactory contact for at least 7 days before having physical contact. Males should be removed when pouch-young are detected. Dasyurids need to be closely monitored and the male removed either following mating to prevent serious and possibly fatal injury or after the arrival of the pouch-young to prevent infanticide. Siblings should be separated at the time of dispersal or prior to their first breeding season for the same reason.

Because many of the smaller species such as kowaris and phascogales breed for only one or two seasons, captive spaces rapidly fill up with reproductively senescent individuals necessitating rehousing or culling to keep the colony reproductively active.

Institutions and individuals with specialised expertise

Institutions

<p>Perth Zoo Perth, Western Australia</p> <p>Phone: (08) 9474 0444 Email: email@perthzoo.wa.gov.au Internet: http://www.perthzoo.com.au</p>	<p>Taronga Zoo Sydney, New South Wales</p> <p>Phone: (02) 9969 2777 Email: tz@zoo.nsw.gov.au Internet: http://taronga.org.au/taronga-zoo</p>
<p>Zoos Victoria Veterinary Departments:</p> <p>Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au</p> <p>Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville</p>	

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<p>Dr Julie Old (dunnarts, red-tailed phascogales, kultarrs: immunology, disease and nutrition) Water and Wildlife Ecology Research Group School of Science and Health University of Western Sydney</p> <p>Phone: (02) 4570 1283 Email: j.old@uws.edu.au</p>	

Bibliography for dasyurids

- Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.
- Geiser, F 2008, 'Ontogeny and phylogeny of endothermy and torpor in mammals and birds', *Comparative Biochemistry and Physiology – Part A*, vol. 150, pp. 176–80.
- Holz, P 2008, 'Dasyurids', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 359–82.
- Jackson, S 2003, 'Carnivorous marsupials', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 53–98.
- Körtner, G & Geiser, F 2011, 'Activity and torpor in two sympatric Australian desert marsupials', *Journal of Zoology*, vol. 283, pp. 249–56.
- Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.
- Lambert, C, Gaikhorst, G & Matson P 2011, 'Captive breeding of the sandhill dunnart, *Sminthopsis psammophila* (Marsupialia: Dasyuridae): reproduction, husbandry and growth and development', *Australian Mammalogy*, vol. 33, pp. 21–7.
- Van Dyck, S, & Strahan, R (eds) 2008, *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Numbat

The numbat is a small (up to 750 g) diurnal marsupial. They have a highly-specialised diet feeding only on termites. Once widespread, it experienced a major decline in distribution after European settlement and is now confined to restricted sites in the forests and woodlands of south-western Western Australia. Research on this species should not commence without considerable consultation with experienced wildlife researchers and zoo experts from Western Australia. Further information on the basic biology of this species is provided in Friend (2008).

Numbat – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Numbat, <i>Myrmecobius fasciatus</i> .	Vulnerable.	South-western WA.	Highly-specialised diet of termites.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Numbats are not easy to catch using conventional trapping methods, and expert assistance or guidance must be sought. One approach is to scan for animals using binoculars, pursue them on foot until they enter shelter such as a hollow log and then retrieve them by hand. They should be placed in soft calico bags or pillow cases for handling and transport over short distances. Some animals may be particularly nervous, thus handler training and experience is important. For longer distance transport they should be placed in their holding bag, into wooden transport boxes (350 mm × 400 mm × 200 mm) with adequate padding, such as seagrass, and with holes in the sides to facilitate air flow. Transport should be at night or in the early morning to avoid heat stress. If temperatures are high, an air-conditioned vehicle is required. Numbats should not be transported for periods in excess of 24 hours. It is very important to seek expert advice before commencing any studies on this species.

Numbats have individual banding patterns on their dorsal surface that can be used for identification. Microchips (for example, passive integrated transponder tags; 'PIT' tags) implanted subcutaneously between the scapulae are also suitable for marking individuals.

Captive husbandry

Numbats are not easily kept in captivity because of their highly-specialised diet of termites and expert advice must be sought. In addition, numbats are regarded as solitary in the wild, and should be held individually (or in pairs for breeding) unless very large enclosures are available.

Numbats can be held in wire (25 mm × 12 mm weldmesh) enclosures measuring 5 metres × 3 metres × 2 metres high, but these must have a fully enclosed roof as the animals climb very well and will escape. To allow the animals to burrow, the mesh floor should be buried beneath soil of a depth of approximately 1 metre. Enclosures should have sun exposure to allow the animals to bask, but should also have adequate shade, such as 90% shade-cloth, to protect them from heat extremes. Numbats become inactive at temperatures over 30 °C.

Numbats retreat to den sites when it is cold and wet. Hollow logs and nest boxes should be provided as den sites, along with suitable nesting material. Visual barriers are important for this species so, wherever possible, additional cover should be provided on lower sections of the wire of cage walls using materials such as branches, tussocks and shade cloth. Enclosures should be cleaned regularly. Further information is provided in Power & Monaghan (2003), however before considering research on this species, expert advice should be sought (e.g. wildlife experts and Perth Zoo).

Diets for captive numbats

Numbats are not easily kept in captivity because they must be supplied with live (or frozen) termites to keep them in good health. Further information on captive diets is provided in Power & Monaghan (2003). Expert advice should also be sought from Perth Zoo.

Routine sample/data collection

Because collection of blood samples may be difficult, expert veterinary advice must be sought (e.g. Perth Zoo veterinarians). Standard data collected should include body mass to the nearest gram, sex, coat condition, general behaviour (activity and demeanour), approximate age based on morphometrics and reproductive status based on pouch condition and the number and size of pouch-young.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Gaseous anaesthesia can be achieved using isoflurane in oxygen using a face mask (5% for induction, 1.5–3% for maintenance), with an oxygen flow rate of 1L/minute. Zoletil® (tiletamine/zolazepam at 5–9 mg/kg body weight, injected IM) can be used in the field, however ‘topping-up’ with Zoletil® is not recommended. There have been no pharmacokinetic studies on any analgesics for this species, thus if pain relief is required, you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Numbats can be humanely killed using an overdose of sodium pentobarbitone (150 mg/kg) injected IP or IC or IV. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Very few serious disease issues have been identified in numbats. Further information is provided in Vitali & Monaghan (2008).

Specific breeding requirements

Numbats do not breed well unless specific, appropriate diet and captive husbandry conditions are provided. Expert advice should be sought, in particular, from Perth Zoo.

Institutions and individuals with specialised expertise

Institutions

Perth Zoo

Perth, Western Australia

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Bibliography for numbats

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Bester, A & Rusten, K 2009, 'Trial translocation of the numbat (*Myrmecobius fasciatus*) into arid Australia', *Australian Mammalogy*, vol. 31, pp. 9–16.

Friend, JA 2008, 'Numbats', in S Van Dyck & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 162–65.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Power, V-L & Monaghan, C 2003, 'Numbats', in S Jackson (ed.) *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 99–125.

Vitali, S & Monaghan, C 2008, 'Numbats', in L Vogelneust & R Woods R (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 383–94.

Marsupial moles

The marsupial moles are small (up to 70 g), extremely cryptic species with a burrowing lifestyle. They rarely come to the surface. They occupy remote arid zone areas of north-western and Central Australia. As a result, their biology is very poorly understood. These species are very difficult to maintain in captivity. The limited information available on the general biology of these species is provided in Benshemesh (2008) and Benshemesh & Aplin (2008).

Marsupial moles – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Southern marsupial mole, Itjaritjari, <i>Notoryctes typhlops</i> .	Endangered. Abundance unknown.	Sand dune deserts of Central Australia, the arid zone.	Small subterranean prey, including insects (especially ants and termites), spiders and geckos.
Northern marsupial mole, kakarratul, <i>Notoryctes caurinus</i> .	Endangered. Abundance unknown.	Sand dune deserts of the northern WA arid zone, including the Great Sandy Desert and Little Sandy Desert.	Poorly known, probably similar to the southern marsupial mole but may be more restricted to invertebrate larvae.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Due to their fossorial lifestyle and remote distribution in arid Australia, the capture of wild marsupial moles is extremely difficult, being possible only on the rare occasions when the animals are observed at the surface. The animals may appear at the surface more often after rain. Limited information suggests that marsupial moles might habituate to gentle handling in captivity, but do not tolerate firmer physical restraint. Marsupial moles must not be subjected to cold or very hot temperatures during transport (see 'Captive husbandry' below).

Captive husbandry

Marsupial moles are extremely difficult species, with no success in keeping these animals alive in captivity for any length of time. However, the lack of knowledge on captive maintenance is compounded by the fact that very few animals have ever been brought into captivity. The few that have been brought in from the wild have generally survived for 10 weeks or less, suggesting that husbandry conditions are not well enough understood to ensure survival (Howe 1975, Jackson 2003).

Some evidence indicates that marsupial moles will die if exposed to cold temperatures (Howe 1975), which is not surprising for a specialist subterranean animal with small body size and thermolabile body temperature (Withers, Thompson & Seymour 2000). They shiver at temperatures below 16 °C, and it has been suggested that housing should provide a thermal gradient from 22 °C to 32 °C. In the wild, tunnels are mainly 20–60 cm beneath the surface, thus deep sand should be provided for burrowing. However, because marsupial moles tunnel through lightly-cemented sand in the wild (Benshemesh 2008), replicating this in captivity is likely to be difficult and preparation of a suitable substrate in a captive facility might take several weeks or months.

There is no information on the social behaviour of marsupial moles to inform whether they should be held singly or in groups in captivity, but they are assumed to be solitary (Johnson & Walton 1989, Benshemesh & Johnson 2003).

Diets for captive marsupial moles

The diet provided to the small number of animals that have been held in captivity includes invertebrates, insect larvae, ant eggs and larvae, mealworms and geckos (Howe, 1975, Benshemesh & Johnson 2003, Jackson 2003). While animals were fed on these dietary items, many still lost condition and died. Whether this was the result of an inappropriate diet or other factors relating to inadequate husbandry conditions, is not known.

Routine sample/data collection

Due to the paucity of information on this species, ensure that body mass, head and body length, tail length and sex is always recorded for any marsupial mole that is encountered in the field.

Samples for DNA analysis should also be collected from hair with follicles or from a mouth swab. These samples should be stored in 70% ethanol or dried and kept sterile, as they will provide important information for any future studies including distribution and population connectivity. Scat samples should be collected if available or possible, as these are very valuable for studies of the diet of marsupial moles.

Anaesthesia, analgesia and sedation

Approaches to anaesthesia for this species are unknown, and if attempted, should only be performed by trained personnel. Marsupial moles have a low metabolic rate and a variable body temperature (see Withers, Thompson & Seymour 2000) that should be considered if administering anaesthetics. Likewise, there have been no pharmacokinetic studies on any analgesics for these species. Thus if pain relief is required, you must seek veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Marsupial moles can be humanely killed using an overdose with sodium pentobarbitone (150 mg/kg) injected IP or IC. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Due to the fossorial lifestyle and remote distribution of marsupial moles in arid Australia, there is no information available regarding health issues, disease control and zoonoses.

Specific breeding requirements

Due to the fossorial lifestyle and remote distribution of marsupial moles in arid Australia, virtually nothing is known about the reproductive biology of marsupial moles.

Individuals with specialised expertise

Individuals

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Bibliography for marsupial moles

- Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.
- Benshemesh, J & Johnson, K 2003, 'Biology of marsupial moles (*Notoryctes*)' in M Jones, CR Dickman & M Archer (eds), *Predators with pouches: the biology of carnivorous marsupials*, CSIRO Publishing, Melbourne.
- Benshemesh, J 2004. *Recovery plan for marsupial moles Notoryctes typhlops and N.caurinus, 2005–2010*. Northern Territory Department of Infrastructure, Planning and Environment, Alice Springs.
- Benshemesh, J & Aplin, KP 2008, 'Kakarratul, *Notoryctes caurinus*', in S Van Dyke & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 410–11.
- Benshemesh, J 2008, 'Itjaritjari, *Notoryctes typhlops*', in S Van Dyke & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 412–13.
- Howe, D 1975, 'Observations on the behaviour of a captive marsupial mole (*Notoryctes typhlops*)' *Australian Mammalogy*, vol. 1, pp. 361–65.
- Jackson, S 2003, 'Carnivorous marsupials', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 53–98.
- Johnson, KA & Walton, DW 1989, 'Notorictidae' in DW Walton & BJ Richardson (eds), *Fauna of Australia: Vol 1B Mammalia*, Australian Government Printing Service, Canberra, pp. 591–602.
- Thompson, GG, Withers, PC & Seymour, RS 2000/2001, 'Blind diggers in the desert' *Nature Australia*, Summer 2000/2001, pp. 26–31.
- Withers, PC, Thompson, GG & Seymour, RS 2000, 'Metabolic physiology of the north-western marsupial mole, *Notoryctes caurinus* (Marsupialia: Notoryctidae)', *Australian Journal of Zoology*, vol. 48, pp. 241–58.

Bandicoots and bilbies

The extant bandicoots and bilbies are divided into two families, Peramelidae and Thylacomyidae, respectively, within the Order Peramelemorphia (Paull 2008; Lynch 2008). The family Peramelidae is divided into three genera: *Perameles* and *Isoodon* in the subfamily Peramelinae and *Echymipera* in the subfamily Echymiperinae. The family Peramelidae contains the seven extant bandicoot species in Australia, while the family Thylacomyidae contains the only extant species of bilby, the greater bilby (Paull 2008). The bandicoots and greater bilby are small- to medium-sized (190–3100 g) nocturnal, omnivorous animals occupying a wide range of habitats including tussock grasslands, heaths and open forests. Information on individual species is provided in van Dyck & Strahan (2008), Lynch (2008). These marsupials have a rapid reproductive rate and are generally short lived.

Bandicoots and bilbies – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Rufous spiny bandicoot, <i>Echymipera rufescens</i> .	Not listed.	Common in the rainforest areas of Cape York, Qld.	Diet includes invertebrates, fungi, fruit, plant materials and seeds.
Northern brown bandicoot, <i>Isoodon macrourus</i> .	Not listed.	Occurring along the east coast of Australia from the Hawkesbury River to Cape York and then across the NT to the north of WA. Also found on islands off the north and east coasts of Australia. Occurs in southern Papua New Guinea. The northern brown bandicoot is the common bandicoot of suburban gardens along the east coast of Australia, north of Sydney.	Omnivorous. Diet includes insects and other invertebrates such as earthworms, spiders, berries, grass seeds, subterranean fungi and plant fungi.
Southern brown bandicoot/ southern short-nosed bandicoot, <i>Isoodon obesulus</i> .	<i>Isoodon obesulus obesulus</i> is endangered. <i>Isoodon obesulus nauticus</i> is vulnerable.	South-eastern Australia, mainly Vic. and the south- eastern coast of NSW, south-eastern SA, southern WA and northern Queensland.	Diet includes invertebrates, including underground beetle larvae and fungal material.
Golden bandicoot, <i>Isoodon auratus</i> .	Vulnerable.	Tropical subhumid north-western Kimberley in WA, the Arnhem Land coast in NT, Qld. Thrives on Barrow Island, off the WA coast.	Diet of the north-western Kimberley population includes termites, insect larvae, centipedes and plant material. The Barrow Island animals eat termites, ants, moths, turtle eggs, small reptiles, the common rock rat, roots and tubers.
Western barred bandicoot. <i>Perameles bougainville</i> .	Endangered.	Limited to two islands off the WA coast (Bernier and Dorre Islands) and has been reintroduced to Heirisson Prong in Shark Bay, WA and to Roxy Downs, SA.	Diet includes insects, other small animals, berries and seeds.
Eastern barred bandicoot, <i>Perameles gunnii</i> .	Vulnerable in Tas. and endangered on the mainland.	Tas., particularly in the northern and eastern regions and in three small reintroduced colonies in south-western Victoria.	Earthworms, insects and their larvae, tubers, bulbs, underground fungi, berries and fruits.
Long-nosed bandicoot, <i>Perameles nasuta</i> .	Not listed.	Relatively common along the eastern coast of Australia, from the south-western coast of Vic. to Cooktown, Qld. Also found on the east coast of Cape York and on Fraser and Hummock Hill Islands, Qld.	Insects and other invertebrates, small vertebrates, lizard eggs, seeds, fruits, fungi and plant material.

Bandicoots and bilbies – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Greater bilby, <i>Macrotis lagotis</i> .	Vulnerable.	Isolated arid and semi-arid regions in WA (Pilbara and western Kimberley), NT (Tanami Desert) and south-western Queensland (Diamantina National Park, Astrebla Downs National Park). Reintroduced colonies in south-western Queensland (Currawinya National Park), NSW (Scotia Sanctuary, WA (Peron Peninsula and Dryandra Woodland) and SA (Arid Recovery near Roxby Downs, Yookamuura Sanctuary, Venus Bay Conservation Park, Thistle Island).	Invertebrates (such as spiders, ants, termites, beetles, insect larvae), fungi, fruit, plant roots and vegetative parts and seeds.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

General techniques, applicable across species in this grouping

The techniques used for capture, handling, sampling and health assessment are common to all bandicoot species and the bilby and, unless otherwise noted, are outlined below under 'Capture, handling, marking for identification, transport'. However, information on captive management of individual species is provided in Jackson (2003). In addition, researchers should consult experts in dealing with particular species and the appropriate literature.

Capture, handling, marking for identification, transport

For capture in the wild, wire-mesh cage traps (approximately 36 cm × 21 cm × 17 cm or larger) may be used (see Lynch, 2008). These should be covered, for example, with a heavy plastic bag, to protect animals from rain, especially in colder weather. In hotter weather, traps should be placed to prevent exposure to the sun after dawn. Traps must have Coopex® (or similar insecticide) applied near the trap to prevent ant problems.

Bandicoots and bilbies are mostly nocturnal. Traps should therefore be set around dusk and cleared commencing at dawn. Traps should be left closed during the day in hotter weather. For trapping, baits such as apple or carrot smeared with peanut butter and golden syrup, or bait balls of rolled oats, peanut butter and golden syrup are effective. To minimise the chance of panic in the animals, traps should be approached quietly and animals transferred quickly into a cotton handling bag. Because female bandicoots will sometimes drop and trample their pouch-young when captured in traps, trapping during periods when females will be carrying pouch-young should be avoided if not essential for the project.

Bandicoots and bilbies are delicate and timid and should be carefully handled. The animal should be held with one hand around the shoulders and back of the neck and the other around the hips and groin, thereby supporting the animal's weight. Bandicoots should never be held by the tail. Note that when held, the animal will often flex its body in a sudden and violent manner, raking at the handler with the long claws on its powerful hind feet. This can easily rip open the hand of the handler and hence animals should be handled with caution. Bandicoots are most easily handled in either a calico bag or a pillow case. Animals remain much calmer if their head is covered during handling. If captured during the day and kept in a quiet environment, animals often go to sleep and may be reluctant to wake when handled. Some species such as northern brown bandicoots are amenable to handling and if handled gently, may permit the examination of the pouch without sedation.

Animals should be transported in either a dark calico bag or a pillow case for short journeys. For longer journeys, the animal should be placed in an adequately-ventilated plywood box or a standard small animal transport container such as a 'Pet Pak', covered by a hessian bag to create a darkened environment. All animals should be transported in air-conditioned vehicles, particularly during the warm summer months.

Captive animals should be caught using a calico bag or a hand-held cloth net (50 cm in diameter), as trapping increases the risk that females may lose pouch-young. Females carrying pouch-young should not be caught unless essential for the project. Mesh nets should not be used as they increase the risk of injury to the animals. The cloth net should have a drawstring opening that facilitates transfer from the net to a cloth bag. An animal in a nest may be caught by placing a net over the animal's nest and then transferring the animal into a calico bag. Animals running free in the pen can also be caught using a hand net.

Bandicoots and bilbies are commonly marked for identification using a microchip (for example, passive integrated transponder tags; 'PIT' tags), which is inserted under the skin between the scapulae (Lynch, 2008). Ear tattoos may also be used, or small holes (for example, 2 mm in diameter) can be punched in a specific pattern adjacent to the ear margin. Refer to Lyne (1982) for a marking system. Care must be taken during this process, as bandicoot ears are delicate and susceptible to tearing.

Bandicoots do not tolerate collars, thus fitting radio transmitters to them is problematic and risky. Seek expert advice before commencing radiotelemetry studies.

Captive husbandry

Bandicoots and bilbies should be housed in fully-galvanised wire-enclosed cages to prevent predation by foxes, cats, dogs, owls and raptors, and to prevent young bandicoots from escaping. The floor of enclosures should have soil or sand to a depth of 10–20 cm, and leaf litter areas to allow animals to forage and dig nests. Galvanised wire mesh (1 cm × 1 cm) should also be placed under the floor base (30 cm depth) to stop animals digging out of the cage. Ideally, the wire-mesh cage walls should be cemented into the ground to a depth of 30 cm to prevent animals escaping.

The cage walls should consist of a smooth material such as plywood, up to a height of 1.5 metres to prevent animals catching their claws in the wire. The enclosure should include a grassed area and tussock grasses, hollow logs, branches and small shrubs, which the animals may use for shelter and nest building materials. Animals will generally build their own nests, but additional shelter should be provided. Plywood sheets may be attached at an angle to the cage walls, with straw bedding placed under the sheets. Nesting boxes should also be provided (for example, 50 cm high × 40 cm wide × 60 cm or more long). A hinged lid on top of the box allows access for capture.

Unlike bandicoots, bilbies in the wild dig burrows of up to several metres in which to shelter during the day (Lynch, 2008). Therefore, captive bilbies should be provided with an artificial burrow of plastic tubing 15 cm in diameter and several metres long as a means of access to the nesting box. Approximately one-third of the cage should be covered to provide shelter from the sun and rain. When the temperature is above 33 °C, some sort of cooling should be used, as bilbies and bandicoots prefer a cool and moist environment. This may include the misting of cages and the use of fans. Further information is provided in Jackson (2003). Unlike bandicoots, male bilbies are less aggressive and can therefore be housed together. An ideal pen size for a mating pair, or two females and a male is 8 metres × 5 metres.

Male bandicoots should generally be housed individually in small cages, because they invariably fight when housed together. For housing a breeding pair, a cage of 6 metres × 3.5 metres should be used to maximise breeding success. For larger groups of animals, seven adults (one male and six females) can be housed in enclosures of 8 metres × 10 metres. Animals should not be housed in conditions that cause obesity or nail problems. They should be housed on earth substrate with enough room to dig and exhibit normal behaviours. Lactating female bandicoots have also successfully raised litters in indoor pens 80 cm wide × 80 cm high × 120 cm long.

Diets for bandicoots and bilbies

The diet of captive bandicoots and bilbies may include commercial dried dog kibble, insects (such as crickets), other invertebrates (such as mealworms), seeds and small amounts of vegetables such as carrot, pumpkin, endive, mushrooms, sweet potato, sweet corn, broccoli and beetroot (refer to Jackson 2003 and Lynch 2008). Kangaroo cubes, lucerne chaff, cabbage, hard-boiled eggs, meadow hay and leafy branches may also be provided. Soft fruits such as bananas, citrus fruit and pears should **not** be given. Clean water should be provided ad libitum.

Hand rearing of bandicoot young using an artificial milk replacer has been successful. However, for some species, the milk replacer must be supplemented with lipid. Lynch (2008) provides details on hand rearing.

Routine sample/data collection

Standard data collection should include sex, body mass, head length, hind-foot length and, if possible, the sex, head length and number of pouch-young.

Blood is routinely collected from the femoral vein, which is often visible in the inguinal area. The cephalic vein, lateral tail vein, lateral saphenous vein and medial tibial artery can also be used (Lynch 2008). Blood collection from the orbital sinus is no longer acceptable because of animal health and welfare issues (see Lynch 2008).

The recommended maximum handling regime for animals is no more than twice daily for a maximum of 5–7 days (Lynch 2008).

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. The preferred method of chemical restraint is anaesthesia by inhalation of isoflurane in oxygen (5% for induction, 1–2% for maintenance) with an oxygen flow rate of 1L/minute administered via a face mask (Lynch, 2008). Bandicoots can become anaesthetised very quickly, often within 20–30 seconds, using isoflurane. Researchers need to reduce the isoflurane rate as soon as practical to 2%. The use of injectable anaesthetics is rare in bandicoots. Bilbies may struggle when anaesthetised using a face mask. Initial sedation with midazolam (0.1–0.2 mg/kg, IM) may therefore be necessary. Fasting is not required prior to anaesthesia. Intubation may be performed with a small-bladed laryngoscope and a 2 mm endotracheal tube (uncuffed). Animals should be placed in a nest box, carefully monitored and not released until they have fully recovered from anaesthesia.

There have been no pharmacokinetic studies on any analgesics for this species, thus if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Bandicoots and bilbies can be humanely killed using an overdose of sodium pentobarbitone (150 mg/kg) injected IV, IP or IC). If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Bandicoots and bilbies carry a large number of parasites (Lynch 2008). These may include nematodes, acanthocephalans, cestodes, protozoans and trematodes. Fleas, ticks and mites are commonly found on bandicoots. Ivermectin (10 mg/mL) given at 200 µg/kg, SC, can be used for tick and mite infestations, while fleas can be treated by fipronil (Frontline®, 2.5 g/L) sprayed over the coat, or imidacloprid (Advantage®, 100 mg/mL) given at 10mg/kg, percutaneously. Because fleas can be a problem, particularly in spring and summer, nest boxes should be dusted with bird flea powder or spray.

A comprehensive list of infections and medical drugs is provided in Lynch (2008). Some common problems of captive bandicoots include internal parasites and stress that may cause gastric ulcers and death. Bacterial septicaemia caused by infection of wounds is also common. Both captive and wild animals may also develop a papilloma-like condition with lesions developing on mucosal surfaces and skin, particularly on the face, feet and in the pouch. These masses may result in hair loss, skin thickening and nodular growths, with some growths becoming malignant. Affected animals may have to be killed humanely. Bandicoots and bilbies are also very susceptible to *Toxoplasma gondii*, which is transmitted from cats and results in death.

Captive animals are very susceptible to periodontal disease, which may be due to their diet. Their teeth should therefore be regularly checked. Tartar build-up may be high at two to three years of age, with animals experiencing loose teeth and bleeding gums. The provision of a predominantly dry food diet (such as dog kibble) and food supplements that the animals must chew (such as *Acacia* spp. gum placed in bamboo sticks) may alleviate this problem (Lynch 2008).

Sick or injured bandicoots may be temporarily housed in small cages with a nest box, with their bedding changed regularly. Animals kept in small cages (60 cm² or less) for long periods may become obese. If animals are housed in areas where they cannot dig, their nails may have to be clipped.

Specific breeding requirements

Information on breeding of bandicoots and bilbies is provided in Lyne (1982), Gemmell (1982) and Lynch (2008). Bilbies and bandicoots are polyoestrous, breed throughout the year in the wild and have a rapid reproductive rate. However, reproductive output may be affected by climate and food availability. For captive animals, the major breeding season is from May to December or January.

To maximise reproductive success, animals should be fed a good diet, provided with good social conditions and housed in cages that minimise stress. Bandicoots and bilbies have a gestation period of 12–16 days that is followed by a lactation period of 55–80 days. Loss of pouch-young in bandicoots is common, especially with large litters of five or six young. Bilbies tend to give birth to 1–3 young. Female young reach maturity at three months of age. The male is left with the female and her young. When the young become independent from their mother they must be removed because they may be injured by the adults.

Bandicoots and bilbies live for a maximum of 3.5 years in the wild, but up to 4 or 5 years in captivity and this must be considered when managing a self-sustaining population (Lynch 2008).

Institutions and individuals with specialised expertise

Institutions

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Bibliography for bandicoots and bilbies

- Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.
- Gemmell, RT 1982, 'Breeding bandicoots in Brisbane (Marsupialia: Peramelidae)', *Australian Mammalogy*, vol. 5, pp.187–94.
- Jackson, S 2003, 'Bandicoots', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 127–44.
- Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.
- Lynch, M 2008, 'Bandicoots and bilbies', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 439–64.
- Lyne, AG 1982, 'The bandicoots *Isoodon macrourus* and *Perameles nasuta*: their maintenance and breeding in captivity', in DD Evans (ed.), *The management of Australian mammals in captivity*, Proceedings of the Scientific Meeting of the Australian Mammal Society, Healesville, Victoria, February 1979, The Zoological Board of Victoria, Melbourne. pp. 47–52.
- Paull, D 2008, 'Order Peramelemorphia. Bandicoots and Bilbies', in S Van Dyck & R Strahan (eds), *The mammals of Australia.*, 3rd edn, Reed New Holland, Sydney, pp. 169–95.
- Pavey, C 2006, *National Recovery Plan for the Greater Bilby *Macrotis lagotis**. Northern Territory Department of Natural Resources, Environment and the Arts.
- Seebeck, JH, Brown, PR, Wallis, RL & Kemper, CM (eds) 1990, *Bandicoots and bilbies*, Surrey Beatty & Sons, Pty Ltd, Sydney.
- Southgate, RI, Christie, P & Bellchambers, K 2000, 'Breeding biology of captive, reintroduced and wild greater bilbies, *Macrotis lagotis* (Marsupialia: Peramelidae)', *Wildlife Research* vol. 27, pp. 621–8.
- Van Dyck, S & Strahan, R (eds) 2008, *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Koala

The koala is the only extant member of the family Phascolarctidae. There are three recognised subspecies, *Phascolarctos cinereus adustus* (Queensland), *P. c. cinereus* (New South Wales) and *P. c. victor* (Victoria). Koalas are strongly sexually dimorphic, with males substantially heavier than females. They exhibit a strong cline in morphological characteristics across their distribution, with animals from the north being substantially smaller (weighing between 4–9 kg) with shorter, paler fur, through to animals in the south which are larger (weighing between 7–15 kg) with longer, darker fur. Detailed information on the basic biology of this species is provided in Martin & Handasyde (1999) and Martin, Handasyde & Krockenberger (2008).

Koala – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Koala, <i>Phascolarctos cinereus</i> .	Vulnerable in Qld, NSW and ACT but not in Vic. or SA.	Widespread in eastern Australia, but population density is highly variable. Occurs from the southern end of the Atherton Tablelands in Qld to Vic., with a limited distribution in SA. Overabundant in some areas in south-eastern Australia, including Kangaroo Island, SA and a number of sites in Vic.	Specialist folivore. Locally, feeds principally on foliage of a limited number of eucalypt species, with substantial regional variation in preferred food trees.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Koalas should only be caught by people with considerable experience. Consideration of environmental conditions is very important when capturing wild koalas. They should not be caught and handled during wet and cold conditions. Handling them when they are wet disrupts the insulative properties of the fur. This can expose them to cold stress because wild koalas tend to retreat to the canopy after release, where they may experience high wind chill. Capture and handling under hot conditions should also be avoided as the animals can rapidly become heat stressed due to their thick fur. Seek local expert advice on appropriate catching conditions, as these will vary substantially across the very large distributional range of this species. For example, the Victorian Koala Management Strategy stipulates that animals should not be captured at ambient temperatures above 30 °C.

Slightly different capture techniques are used by different groups depending on frequency of capture and the constraints of the habitat and climate. In Victoria and South Australia, koalas are generally caught using a rope noose, fitted with a stop knot to prevent pulling too tightly around the animal's neck. This rope noose is dropped over the animal's head using an extendable pole. Once noosed, a flag is waved above the animal's head to encourage it to back down to the base of the tree. Other teams simply flag the animals to the ground, however without the use of a noose there is a risk that some animals might jump and fall. In taller forests, tree climbers or portable scissor lifts are deployed to gain access to the animals. A light-weight portable trap, which is set up around the base of the tree, has also been developed for capture (see Phillips 2011). This is suitable for some situations, but seriously limits the number of animals that can be caught during field work. When conducting research on wild populations, individuals should always be released at the base of the tree from which they were captured.

Once on the ground, koalas should immediately be placed individually into a large heavy hessian coffee sack, or similar, for handling. It is particularly important to use large sacks in Victoria and South Australia, as the males are too big for standard hessian sacks, such as those used for potatoes. If females are carrying young on the back, the young should be removed at capture and placed in a separate bag to avoid injuries and to facilitate safe sampling and collection of morphometric data.

Koalas can be held and carried or transported short distances in hessian sacks, provided it is not hot. In hot weather they should be transferred into appropriate well-aired crates. Back-young should be placed carefully onto the mother's back after she has been placed in the crate because occasionally confused angry females will bite or lash out at the baby. Animals exhibiting signs of heat stress, including salivating onto their wrists and panting rapidly, should be cooled by applying cold water over the wrists and on the side of the face, followed by immediate release, if possible. Wild koalas can also be picked up or carried by hand for short distances, by firmly grasping the fur between the shoulder blades and on the rump, and holding the animal in an upright position facing away from the handler. However, this requires experience and is not recommended, especially for wild adult male koalas from Victoria and South Australia, which are very strong and can be aggressive. Captive koalas become relatively tame and can be handled with relative ease. Some can simply be picked up from behind by placing a hand under the rump and grasping the fur on the back of the neck. Alternatively, smaller animals can be picked up by grasping the upper arms from behind.

For longer transport distances, koalas should be placed in special transport crates with good air flow in hot weather or, in cold weather, appropriate protection to prevent the animals from becoming chilled. The transport crates should have a slatted base so that the animals are not sitting in faeces or urine. Back-young should be placed carefully onto the mother's back after she has been placed in the crate.

Koalas can be individually marked by tattooing the ears, or by tagging ears with multiple (up to two per ear) plastic swivel tags designed for sheep (for example, Leader Products® tags) and applied with tagging pliers. Such tags can be colour-coded and numbered, allowing remote identification of individuals in the field without the need for recapture. Alternatively, koalas can be identified using a microchip.

Captive husbandry

Seek expert advice before bringing koalas into captivity. Koalas must be housed in pens of appropriate height with a minimum size of 2 metres × 2 metres × 2 metres for short-term research. A larger size pen is preferred for longer-term studies. The pens must have suitably sized and arranged tree branches for the koalas to climb and tree forks in which they can sit. Cages should be partially covered to provide shade and protection from rain.

Diets for captive koalas

Appropriate volumes, presentation and arrangement of fresh branches of eucalypt foliage are absolutely essential for maintaining captive koalas in good health as they have highly stereotyped feeding behaviour and will not feed from a tray or if food is presented inappropriately. Locally preferred food tree species should be identified, as preferences vary regionally. Larger branches of fresh leaves bearing a mix of mature and younger foliage from several of their locally preferred tree species should be offered to the koalas daily. The best indicator of general health is serial monitoring of body weight, which should be measured weekly. In addition, a body condition index, based on palpation of the muscle over the scapula, is very useful for monitoring condition in koalas (see Jackson et al. 2003). In addition, the approximate number of faecal pellets produced by each koala can be monitored daily. Significant decreases in pellet production can quickly reveal a decrease in food intake.

While they are renowned for rarely drinking in the wild, koalas should always have access to clean water in captivity. Extensive information on captive management of koalas is provided in Jackson et al. (2003).

Captive koalas require straightforward but meticulous care due to their dietary fastidiousness and specialisation on eucalypt foliage, which is of inherently poor quality in terms of energy and nitrogen. If koalas fail to feed properly, the energetic constraints of this diet can result in rapid loss of condition, hence the emphasis on seeking expert advice on appropriate feeding for this species in captivity. When bringing koalas into captivity for research, the use of old animals (see below for age determination) should be avoided. Old animals tend to suffer additional problems in maintaining body weight and general health due to their advanced tooth wear that makes mastication and digestion inefficient.

Routine sample/data collection

Standard morphometric data, including body weight, head length and wear patterns on the premolar teeth in the upper jaw that indicates age (Jackson et al. 2003) can all be recorded from animals handled in a sack.

Blood samples of up to 10 mL can be collected from conscious animals in the field from the cephalic vein in the forearm arm using a 21-gauge needle. The animal should be placed on the ground and the head should be kept covered by the handling bag while an assistant sits astride the koala without placing any weight on the animal. Then the koala's arm can simply be pulled out of the handling bag to enable collection of a blood sample.

Tissue for DNA analysis can be collected from towards the outer margin of the ear with a 3 mm diameter biopsy punch. If animals are being ear tagged, the small plug of tissue generated provides an excellent sample for DNA analysis.

Faecal pellets can often be collected fresh from the holding bag or from beneath the tree where the animal was caught.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Koalas can be sedated with Zoletil® (tiletamine/zolazepam 5–7 mg/kg body weight) or Alfaxan® (5 mg/kg body weight) injected IM into a hind leg. Gaseous anaesthesia can be achieved using isoflurane in oxygen (5% for induction, 1–2% for maintenance), with an oxygen flow rate of 1 L/minute. If anaesthetised or sedated in the field, koalas **must** be held until they are fully awake and competent to climb. There have been no pharmacokinetic studies on any analgesics for this species, thus if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Koalas can be humanely killed using an overdose with sodium pentobarbitone, (150 mg/kg) injected IV, IP or IC. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Due to the very large and complex literature in this area, expert advice should be sought on koala health and diseases. Information is also provided in Blanshard & Bodley (2008), Jackson et al. (2003), Ladds (2009).

A large number of pathogens and parasites have been recorded in koalas, many of which are widespread in wild populations. In part this is due to the very extensive research that has been conducted on the biology of the koala. Chlamydial infection is widespread in wild populations. A major consequence of this infection is female infertility, with eye and urinary tract disease also seen. Retroviruses have also been identified in koalas and are the subject of current research. *Mycobacterium ulcerans*, a potential zoonotic organism, has been recorded as a cause of ulcers in koalas from eastern Victoria.

Specific breeding requirements

Koalas are seasonal breeders and relatively easy to breed in captivity. Expert advice on breeding requirements should be sought from major zoos with captive colonies.

Institutions and individuals with specialised expertise

Institutions

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Bibliography for koala

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Blanshard, W & Bodley, K 2008, 'Koalas', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 227–327.

Jackson, S, Reid, K, Spittal, D & Romer, L 2003, 'Koalas', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 145–181.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Martin, RW & Handasyde, KA 1999, *The koala: natural history, conservation and management*, 2nd edn, UNSW Press, Sydney.

Martin, RW, Handasyde, KA & Krockenberger, A 2008, 'Koala', in S Van Dyck & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 198–201.

Phillips, S 2011, 'Development of a light-weight, portable trap for capturing free-ranging koalas *Phascolarctos cinereus*', *Australian Zoologist*, vol. 35, pp. 747–9.

Wombats

Wombats are large (up to 40 kg) semi-fossorial grazers. There are three extant species in the family Vombatidae, which belongs to the order Diprotodontia, suborder Vombatiformes (the latter also containing the koala). The common wombat occurs in eastern Australia in wetter environments, while the two species of hairy-nosed wombats occupy more arid regions. Detailed information on the three species and their basic biology is provided in Van Dyck & Strahan (2008) and Triggs (2009).

Wombats – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Common wombat, <i>Vombatus ursinus</i> .	Not listed, common, widespread. Status within states varies: vulnerable in Qld, rare in SA, low risk in other states. The subspecies on the Bass Strait islands, <i>V.u.ursinus</i> , is also considered vulnerable.	Widespread in eastern Australia, occurring from south eastern Qld to Vic., into Tas. and a limited distribution in far-eastern SA.	Herbivore/grazer.
Southern hairy-nosed wombat, <i>Lasiorhinus latifrons</i> .	Not listed. Common, limited.	Southern SA with a very limited distribution into south-eastern WA.	Herbivore/grazer.
Northern hairy-nosed wombat, <i>Lasiorhinus krefftii</i> .	Endangered. Rare, limited.	Currently known only from a single site at Epping Forest National Park, Qld.	Herbivore/grazer.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Wild wombats can be captured using anaesthetic darts loaded with Zoletil® (tiletamine/zolazepam) delivered IM at a dose of 3–5 mg/kg. At some sites, if the habitat is fairly open and the wombats are relatively habituated to people, they can be stalked and captured by hand using a large robust hoop net, mounted on a pole. They can also be captured using large (800 mm × 450 mm × 450 mm) heavy gauge, weld-mesh cage traps. These must be dug well into the burrow entrance and securely pegged to prevent the animals from pushing them aside (see Skerratt 2001). Because wombats sometimes refuse to emerge from the burrow into the trap, traps should not be left set for more than two or three consecutive nights.

Once inside a trap, wombats can be sedated with Zoletil® (3–4 mg/kg, IM) to facilitate handling, sampling and collection of morphometric data. Sedation is necessary because wombats are very strong and can be aggressive. Wombats in open habitat can be captured using a technique described as ‘stunning’ (see Taggart et al. 2003) where the wombat is ‘held’ in a spotlight beam. A sonic boom is then created by an experienced marksman firing a high-velocity bullet just above the skull of the wombat. Temporarily disoriented by the light and the sonic boom, the wombat often remains motionless long enough for a team of catchers to run and capture it in a hand net. Once restrained, it can immediately be sedated with Zoletil®.

Wombats can be held safely in large hessian sacks for recovery or short distance transport, however it is essential to ensure that they do not become too hot. This approach is not suitable for longer distances. Wombats can be very aggressive towards each other, hence it is essential to keep sedated animals in a secure location until they are fully recovered. Juvenile animals and tame captive individuals can be picked up from behind by a handler, who passes their arms under the animal’s armpits and locks their hands together over the chest of the animal.

Wombats can be individually marked by tattooing the ears, or tagging the ears with multiple (up to 2 per ear) plastic swivel tags designed for sheep (for example, Leader Products® tags) and applied with tagging pliers. Because the ear tissue is quite thick, it is usually necessary to make a hole in the ear first, using a biopsy punch (see 'Routine sample/data collection' below) or a sharp, sterile leather punch. Such tags can be colour-coded and numbered, allowing remote individual identification in the field without the need for recapture. Application of reflective tape to tags facilitates remote identification of individuals at night using a spotlight.

Fitting radio transmitters to wombats (see Evans 1997) is somewhat problematic as the diameter of the neck is very similar to the head, meaning that collars must be fitted quite tightly, and thus there is a risk of chafing. As wombats can be difficult to recapture this can be a serious issue. Alternatively, intraperitoneal transmitters can be implanted surgically (see Skerratt 2001). It is very important to seek expert advice before commencing radiotelemetry studies.

Wombats are powerful animals and for longer distance travel, they should be transported in very strongly-built crates with adequate ventilation. The animals do not tolerate high temperatures well and should be transported at less than 24 °C, with air conditioning in the vehicle recommended. Due to their low energetic needs, wombats do not need to be fed during shorter-term transport (up to 24 hours), but for longer-term transport they should be provided with food and water. If planning on transporting wombats, seek expert advice from a zoo with appropriate expertise.

Captive husbandry

Wombats generally adjust well to captive conditions. Because they are very powerful diggers and chewers, pens must be very strongly constructed and have an area suitable for the length of time that the animals are being held. Seek advice from major zoos with captive colonies. Pens should be well drained and provided with bedding such as clean straw, a substrate of soil or sand to allow natural digging behaviour.

Pens should include a shaded, cool, dry denning area, such as suitably large concrete pipes, hollow logs or a chamber. If housed in outdoor pens, include a sprinkler system to help keep animals cool in hot weather. Be aware that common wombats do not tolerate temperatures above 24 °C well, while hairy-nosed wombats can deal with environmental temperatures up to about 33 °C.

Common wombats are solitary and can be aggressive, thus should be housed singly or at maximum in pairs, but only if pens are sufficiently large enough. Hairy-nosed wombats are more social and can be held in groups. Information on captive husbandry is provided in Jackson (2003). Expert advice should also be sought from zoos with experience with the relevant species.

Diets for captive wombats

The animals do well on a captive diet of lucerne hay, fresh oats and carrots.

Routine sample/data collection

Standard morphometric data can be recorded from animals handled in a sack or after sedation (see 'Anaesthesia, analgesia and sedation' below). Recorded data can include body weight, ear, head and hind foot length, reproductive status of females and testis length, width and depth for males. Any clinical signs of skin disease should also be recorded (see information on mange under 'Health issues, disease control and zoonoses' below).

Tissue samples for DNA can be collected from the margins of ears. For example, swab the site with 70% ethanol and use a 3–4 mm biopsy punch. Alternatively, if animals are being ear tagged, the small plug of tissue generated provides an excellent sample for DNA analysis.

Blood samples of up to 10 mL can be collected from sedated or anaesthetised animals from any of the femoral, radial, cephalic, brachial, or jugular veins using a 21- or 22-gauge needle. The femoral vein is the most effective site for sampling.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Wombats can be sedated with Zoletil® (tiletamine/zolazepam given at 3–5 mg/kg body weight) injected IM into a hind leg. Sedation can also be achieved with diazepam (0.5–1 mg/kg, IM). Gaseous anaesthesia can also be achieved using isoflurane in oxygen (5% for induction, 1–2% for maintenance), with an oxygen flow rate of 1 L/minute (see Bryant & Reiss 2008). There have been no pharmacokinetic studies on any analgesics for these species, and thus if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Wombats can be humanely killed using an overdose with sodium pentobarbitone (150mg/kg) injected IP or IC. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

The most widespread and significant disease of common wombats is sarcoptic mange, caused by infestations of the mite, *Sarcoptes scabiei* (see Skerratt, Martin & Handasyde 1998; Skerratt 2001). If the disease is not advanced and extensive, it can be treated with a course of readily available drugs such as ivermectin. Expert veterinary advice should be sought. Wombats also carry infestations of several tick species and a range of other parasites (see Bryant & Reiss 2008).

Specific breeding requirements

None of the wombat species breed particularly well in captivity. Information on breeding requirements of the wombat is provided in Bryant & Reiss (2008) and Jackson (2003).

Institutions and individuals with specialised expertise

Institutions

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Individuals

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Bibliography for wombats

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Bryant, B & Reiss, A 2008, 'Wombats', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 329–58.

Evans, MC 1997, 'A collar design for wombats', *Australian Mammalogy*, vol. 20, pp. 119–20.

Jackson, S 2003, 'Wombats', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 183–203.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Skerratt, LF 2001, *Sarcoptic mange in the common wombat, Vombatus ursinus (Shaw, 1800)*, PhD Thesis, Veterinary Science, The University of Melbourne.

Skerratt, LF, Martin, RW & Handasyde, KA 1998, 'Sarcoptic mange in wombats', *Australian Veterinary Journal*, vol. 76, pp. 408–10.

Taggart, DA, Finlayson, GR, Richings, N, Shimmin, G, Dibben, R, Adcock, J & Temple-Smith, PD 2003, 'Environmental factors affecting the capture of southern hairy-nosed wombats (*Lasiorhinus latifrons*) by stunning', *Wildlife Research*, vol. 30, pp. 539–46.

Triggs, B 2009, 'Wombats', 2nd edn, Australian Natural History Series, CSIRO Publishing, Melbourne.

Van Dyck, S & Strahan R (eds), 2008. *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Macropodoids

The superfamily Macropodoidea is divided into three families: Hypsiprymnodontidae (musky rat-kangaroo), Macropodidae (kangaroos, wallabies and relatives) and Potoroidae (bettongs, potoroos and rat kangaroos). This group contains Australia's largest marsupials (the big kangaroos) but also contains much smaller species such as the musky rat-kangaroo that weighs only approximately half a kilogram. The majority of the macropodoids are generalist grazers or browsers, whereas the potoroids and musky rat-kangaroo are more omnivorous.

Macropodoids – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Musky rat-kangaroo, <i>Hypsiprymnodon moschatus</i> .	Least concern.	Restricted to tropical rainforests of north-eastern Qld.	Omnivorous, consuming mainly fruits but also invertebrates, seeds and fungi.
Rufous bettong, <i>Aepyprymnus rufescens</i> .	Least concern.	The north-eastern coast of Australia, west of the Great Dividing Range, south of Cooktown, Qld to north of Sydney, with a smaller population around the centre of NSW/Vic. border.	Generalist herbivore predominantly feeding on herbs, grasses and tubers but may consume entire plants, underground fungi, and plant exudates. It may also consume bones of dead animals.
Long-nosed potoroo, <i>Potorous tridactylus</i> .	Least concern.	A patchy distribution along the south-eastern coast from south-eastern Qld, through NSW and Vic. to just over the SA border as well as in Tas.	Omnivorous, consuming fungi, arthropods, fleshy fruits, seeds and vascular plant tissue.
Bennett's tree-kangaroo, <i>Dendrolagus bennettianus</i> .	Near threatened.	Small population on the Cape York Peninsula, Qld.	Browser that feeds on the leaves of a small number of tree species as well as fruits and fern fronds.
Lumholtz's tree-kangaroo, <i>Dendrolagus lumholzi</i> .	Least concern.	Small population on the Cape York Peninsula, Qld.	Generalist browser.
Quokka, <i>Setonix brachyurus</i> .	Vulnerable.	South-western WA and Rottnest Island.	Primarily a generalist grazer.
Swamp (black) wallaby, <i>Wallabia bicolor</i> .	Least concern.	Along the eastern coast of Australia from western Vic., to the east coast of the Cape York Peninsula, Qld.	Primarily a generalist browser although will feed on grasses and fungi.
Spectacled hare-wallaby, <i>Lagorchestes conspicillatus</i> .	Least concern.	Inhabits tropical grasslands in northern Qld, NT and WA but is rare in coastal areas and absent from the northern-most parts of Cape York Peninsula, north-western NT and the Kimberly, WA. Abundant on Barrow Island, WA.	Generalist grazer.
Bridled nailtail wallaby, <i>Onychogalea fraenata</i> .	Endangered.	A small, free-ranging and self-sustaining population has recently been established within its former range in Idalia National Park in central Qld.	Generalist grazer and browser.
Northern nailtail wallaby, <i>Onychogalea unguifera</i> .	Least concern.	Found in northern Australia in Qld, NT and WA.	Generalist herbivore.
Tasmanian pademelon, <i>Thylogale billardieri</i> .	Least concern.	Currently restricted to Tas.	Generalist herbivore.

Macropodoids – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Red-legged pademelon, <i>Thylogale stigmatica</i> .	Least concern.	Patchy distribution along the east coast of Australia from north of Sydney in NSW to Cape York Peninsula, Qld.	Generalist herbivore.
Red-necked pademelon, <i>Thylogale thetis</i> .	Least concern.	Distributed along the east coast of Australia from southern NSW to southern Qld.	Generalist herbivore.
Brush-tailed rock-wallaby, <i>Petrogale penicillata</i> .	Threatened.	Small isolated populations in East Gippsland and the Grampians, Vic. and sparsely distributed along the east coast from the Kangaroo Valley in NSW to just over the Qld border and Stradbroke Island, Qld.	Mainly a generalist grazer but will feed on browse, seeds, fruit and flowers.
Black-flanked (Black-footed) rock-wallaby, <i>Petrogale lateralis</i> .	Near threatened.	Small isolated populations on the Islands of the Recherche Archipelago, Barrow Island, the Wheatbelt region, the mid-west region, the Gascoyne region and the west Kimberly of WA, the Investigator Group islands of SA and in the Anangu Pitjantjatjara Lands in north-western SA. A larger population is found in the Macdonnell Ranges of NT and another in the ranges of the north-west region of Qld.	Generalist herbivore predominantly feeding on grasses and succulents.
Yellow-footed rock-wallaby, <i>Petrogale xanthopus</i> .	Near threatened.	Isolated populations in the Ranges of the Adavale Basin in south-western Qld and the Coturaundee Range in western NSW, and a continuous distribution in the Flinders Ranges in SA.	Generalist herbivore eating grasses, browse and forbs with browse becoming more important in dry seasons.
Tammar wallaby, <i>Macropus eugenii</i> .	Least concern.	Small isolated populations on islands of the Recherche Archipelago, Houtman's Abrolhos and Garden Island in WA but a very large population on Kangaroo Island in SA, and a remnant mainland population in south-western WA.	Generalist grazer.
Parma wallaby, <i>Macropus parma</i> .	Near threatened.	Found in NSW from the Watagan Mountains to the Gibraltar Range.	Generalist herbivore.
Bennett's wallaby, <i>Macropus rufogriseus rufogriseus</i> .	Least concern.	Tas.	Generalist grazer.

Macropodoids – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Red-necked wallaby, <i>Macropus rufogriseus banksianus</i> .	Least concern.	The south-eastern coast of Australia from a small corner in south-eastern SA, through southern and eastern Vic., coastal NSW and southern Qld.	Generalist grazer.
Eastern grey kangaroo, <i>Macropus giganteus</i> .	Least concern.	Widely distributed throughout eastern Australia from the north and east of Tas., the south-eastern corner of SA, throughout almost all of Vic. and NSW and the majority of Qld with the exception of the far north and north-west.	Generalist grazer.
Western grey kangaroo, <i>Macropus fuliginosus</i> .	Least concern.	Widely distributed along Australia's south from western Vic. and NSW through southern SA to southern WA with a small extension into southern Qld.	Generalist grazer, but consumes some browse.
Agile wallaby, <i>Macropus agilis</i> .	Least concern.	Common along the northern coast of Australia.	Generalist herbivore.
Antilopine wallaroo, <i>Macropus antilopinus</i> .	Least concern.	Found in northern WA and NT as well as on the Cape York Peninsula, Qld.	Generalist herbivore.
Red kangaroo, <i>Macropus rufus</i> .	Least concern.	Found throughout inland Australia.	Generalist herbivore.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Some macropodoid species, including red and eastern grey kangaroos, agile, black-tailed, swamp (black) and red-neck wallabies, pademelons and potoroos, are susceptible to capture myopathy. Capture myopathy appears to result from stress combined with extreme exertion, such as when animals have been chased during capture. Particularly nervous animals may be sedated at a dose rate of 1mg/kg diazepam, IM. However, if animals are caught with long-handled nets quietly and one at a time, capture myopathy can be avoided. Capture myopathy is also much reduced or even eliminated in larger macropodids with darting. Capture myopathy may also occur after repeated handling and restraint events, after transport, after exposure to an unfamiliar environment or close confinement. Capturing animals in hotter ambient temperature increases the risk of capture myopathy. Clinical signs include abnormal gait and posture associated with stiffness, and pain. In particular, neck muscles may be involved, resulting in failure to hold the head erect. Signs may appear immediately or become apparent 1–2 weeks post-capture, with deaths occurring up to 4 weeks post-capture. If you are intending to conduct research on macropodids, seek expert advice (e.g. experienced researchers and zoo veterinarians). Further information is provided in Ladds (2009), Vogelnest & Portas (2008) and Appendix IV.

The smaller macropodids (such as rock-wallabies or the quokka), potorooids and the musky rat-kangaroo can be caught in cage treadle traps approximately 40 cm high × 40 cm wide × 70 cm deep. Larger wallabies, such as swamp (black) wallabies are often trapped in specially-designed soft-walled traps (Pollock & Montague 1991; Di Stefano, Moyle & Coulson 2005). Traps are set overnight and checked early in the morning. Traps should be stabilised and secured by either being pegged to the ground, if possible, or should otherwise be weighed down with rocks or a log. This prevents the trap rolling over when an animal is inside. Traps should

be well covered with a tarpaulin or plastic to provide shelter for trapped animals in the event of rain. Traps must be cleared early in the morning. In summer, extra care should be taken to avoid animals becoming overheated while in traps or while being processed after removal from traps.

Recommended baits include apples, oats and fresh white bread for the grazing macropodids. Potoroos and bettongs have been successfully trapped with a mixture of peanut butter, golden syrup and oats, sometimes with pistachio essence. Sweet potato and banana can be used for trapping the musky rat-kangaroos. Musky rat-kangaroos have been successfully trapped using fence traps and no bait to reduce unwanted species being attracted. Trapping success for potoroos and bettongs can be improved if traps are set along runs or natural features such as logs. In all cases, a period of free feeding from traps is recommended to increase trapping success. Animals are removed from traps by one of three methods:

- standing the trap up on end and reaching in via the door to grasp the animal by the base of the tail
- placing the animal straight into a sack
- placing a sack over the opening of the trap and tilting the trap to encourage the animal to enter the sack.

While the last option sounds more animal-friendly, it has a low probability of success. The double-layered trap design (Di Stefano, Moyle & Coulson 2005) allows the internal bag to be removed with the animal inside it, minimising the risk of escape or injury to the animal or the operators. Tree-kangaroos are generally darted with sedative (see below) and caught in netting stretched below the tree by several people. This system of catching must only be carried out by experienced personnel.

Tammar wallabies on Kangaroo Island are hand netted at night from a slow moving vehicle. The hand net consists of an aluminium pole with a metal hoop of 0.5- to 0.75-metre diameter and netting of approximately 20 mm mesh. This is a very efficient and safe method for the animals. They are held in temporary enclosures until transported.

A variety of methods can be used for the capture of the large kangaroos in the field. Drawstring traps can be set to capture kangaroos where they habitually move through fences. The trap consists of a tunnel of netting suspended from a steel mesh frame, with drawstring closures at each end. The operator closes the trap by hand from a nearby hide and then subdues the kangaroo in the trap. Pole syringes (or jab sticks) can be used to capture wild but habituated kangaroos in situations such as nature reserves or golf courses (see King et al. 2011). A range of light-weight extendible poles up to 4 metres long can be used to inject a syringe containing Zoletil® (1:1 ratio of zolezapam and tiletamine) into the haunch of the kangaroo at a dose rate of 5mg/kg estimated body weight. Syringe darts, propelled by blowpipe, compressed gas, powder charge or bow string, can be used to capture wallabies and kangaroos (Roberts et al. 2009). The dart injects a specific dose of anaesthetic, such as Zoletil®, into the haunch of the macropodids. This technique can be used only where macropodids occur in open areas, allowing the operator to fire from a relatively close range (less than 40 metres), and must be performed by a licensed and experienced operator.

There are two main methods of transporting macropodids. Smaller wallabies can be transported by:

- placing them in hessian sacks that are hung so that the animal is standing upright on its feet or resting on the bottom of the sack with plenty of room around their heads at the top
- placing them into rigid plastic standard small animal transport packs (for example, a 'Pet Pak') that are covered so the animals cannot see out, or dog crates in which the bag containing the animal is hung.

In both cases, if animals are being transported over long distances, cages should be insulated or placed in an air-conditioned vehicle. Animals can also be transported in these cages by air. Animals should be contained in boxes or bags singly. Animals may be sedated for transport using diazepam (1mg/kg in the hind leg muscle); however, it is recommended that sedation is avoided to allow the animal to adjust its behaviour in response to stresses.

Medium-to-small animals can also be carried in their sacks hanging from weldmesh racks in specially adapted caravans. For transport of large kangaroos when sedated, animals should not be hung in sacks, but laid on their side. If more than one animal is to be transported the animals' backs should be facing each other to avoid kicking injuries, and bags must have sufficient air vents (holes).

Kangaroos and wallabies are generally marked in the wild and captivity using ear tags and/or microchips. Ear tags may be of the small, metal, numbered fingerling variety, useful for individual identification of large numbers of animals. Ear tags can also be large, coloured plastic tags, often with a reflective area, several of which can be used in either ear at the base of the ear and with various colour combinations for individual identification at a distance during the day or night. The latter method is only suitable for the larger macropodids, and all tags are liable to loss, particularly in males.

Microchips (for example, passive integrated transponder tags; 'PIT' tags) can be used in all species of macropodids and are an excellent method of long-term animal identification, but they are expensive if large numbers of animals are to be marked. Microchips also make it necessary for the animal to be caught for identification. Microchips should be inserted subcutaneously between the scapulae. Because a microchip may migrate over time, all parts of an animal's body should be scanned with the reader when checking for a microchip. Microchips are sometimes lost just after implantation because they can be pushed out through the injection site. This may be avoided by sealing the entry wound with tissue glue.

Captive macropodids may be caught for routine sample collection or health checks by being netted. This capture method is associated with a risk of injury to the animal. Expert advice should therefore be sought.

Animals should not be captured at temperatures above about 30 °C, because they can easily become heat stressed. Animals are shoed into a corner of the yard and as they run out down the fence line they are caught using nets of the type described above. The diameter of the metal hoop and depth of the net need to be appropriate for the size of the macropodid being caught, with hoops of 60–70 cm in diameter and 60–80 cm deep being useful for animals up to 10 kg in weight. Animals larger than this may require a hoop 1 metre in diameter and a net approximately 1 metre deep. If the animals are being held in very large enclosures with a lot of cover, it may be more appropriate to catch them by darting or trapping.

Once the animal is caught, the catcher should remove the animal from the net by the base of the tail with legs toward the catcher. A second person then 'swoops' a hessian sack over the animal and the catcher releases the tail as the animal is enclosed by the open sack. The sacks should then be tightly tied at the opening to prevent the escape of animals. At all times, animals in sacks should be kept out of the sun, and on hot days catching should be discontinued. Only the part of the body to be examined is exposed. Unless handlers are experienced, two people are generally required to handle the smaller macropodids with one person holding the animal and the other conducting the examination. For large kangaroos, more people may be required to restrain the animals during brief examinations without anaesthesia or sedation.

For pouch checking, a smaller macropodid may be held on the lap of one person, with the animal's head against the person's abdomen and the tail passed between the animal's legs. The person holding the animal should hold the legs steady to prevent the animal from kicking, while the examiner has their face close to the wallaby's body from the side to access the pouch. For the larger wallabies and kangaroos, the animal should be laid on one side on the ground with one person lightly leaning across the animal's ribs and holding the feet steady while the other person examines the animal. Some macropodids such as tammar wallabies and swamp wallabies are amenable to handling in this way with no anaesthesia, the former because they are small and resilient and the latter because they are very calm once their heads are covered. Other macropodids should be anaesthetised before handling because of the high risk of stress-induced muscular myopathy or shock. Gaseous anaesthesia with isoflurane is suitable for captive animals. Particularly nervous animals may be sedated at a dose rate of 1mg/kg diazepam, IM in the haunch. This dose will keep the animal quiet for approximately four hours.

After capture and examination animals must be released carefully to avoid injury. The bag is placed on the ground and the opening is untied. Direct the opening of the bag into a clear, flat space with no obstacles for the animal to run into because animals will often leave the bags heedless of what they are heading towards. Captive animals that have been anaesthetised or sedated should only be released once the effects of the anaesthetic or sedative have worn off. Recovery and release of larger wild kangaroos is better if they are left undisturbed in a protected position or an open sack.

Some macropodid females have the tendency to drop their pouch-young if they are stressed during capture and handling, especially if the pouch-young is quite large. Smaller pouch-young need to be re-attached to the nipple in one of two ways:

- Anaesthetise the female and gently grasp the nipple in a pair of small forceps and guide the nipple into the mouth of the pouch-young and hold it there until the young re-attaches.
- Push the teat of an un-anaesthetised female back into the mouth of the young using a moistened and chewed matchstick end.

The pouch-young are more likely to remain in the pouch if their positioning is assisted by gravity. Both methods described above should only be used by experienced personnel to avoid trauma to the teat and the pouch-young's mouth. Older pouch-young that are no longer permanently attached to the teat can just be placed back in the pouch. For both smaller and older pouch-young, the pouch may then be taped shut using masking tape prior to release. The female will remove the sticky tape as she grooms.

Captive husbandry

The responses of wallabies to captivity are species specific, and experts should be consulted before planning close-quarter housing. For long-term housing, macropodids should be kept in large, grassed, outdoor enclosures. Perimeter fences should be at least 2 metres in height, with an electric wire run just above the top of the mesh. To prevent access by foxes, a barbed-wire overhang of approximately 50–60 cm that faces outwards from the enclosure at a 45° angle should run along the top of the fence. Electric fencing at the top of the fence but below the overhang to prevent the entry of foxes is essential. The bottom of the fence should be dug at least 0.5 metres into the ground, with an apron extending from the bottom of the fence outwards for at least 0.5 metres. This is to prevent animals such as wombats, foxes or rabbits breaching the perimeter by digging holes at the base of the fence.

Rock-wallabies, musky rat-kangaroos, tree kangaroos and quokkas (as well as foxes) are agile climbers. Hence there should not be any trees, branches or rocks within 2–3 meters of the perimeter fence. Ideally, the corners of every enclosure should be covered into smooth curves with extra wire netting and plastic sheeting. Fence posts must be placed on the outside of the wire netting to minimise injury to animals if they run into the fence.

Shelter must be provided in captive enclosures. Trees, shrubs, fallen logs, supplemented by artificial A-frame shelters are ideal. Rock-wallabies should also be provided with rock piles (or hay bales), ledges and caves which face north or northwest. Rock piles not only provide shelter but also allow rock-wallabies to exercise their agility. Musky rat-kangaroos and tree-kangaroos should be provided with angled and/or horizontal branches to climb. Two to three shelter spots should be provided per animal. North and north-west aspects provide places for all macropodids to sun themselves in the mornings.

In the event that wallabies need to be housed indoors on artificial surfaces for research purposes then it is important to have dry clean floors. Consider sponging the floors dry after hosing to prevent foot problems resulting from excess water. Bedding of wood shavings should be provided but do not use sawdust, as the dust creates a respiratory issue. Pens should be cleaned regularly, and when necessary, floors can be cleaned with hospital or veterinary grade disinfectant.

If wallabies are being housed in small numbers and require being caught on a regular basis (such as weekly), then marking the tails, for example with non-toxic tape, can identify individuals.

Close-quarter housing for kangaroos is necessary for some experimental work such as metabolism studies (see Munn & Dawson 2001; Munn & Dawson 2003; Munn, Banks & Hume 2006; Munn et al. 2009; Munn, Dawson & McLeod 2010). Use of floor pens and metabolism cages, require animals to be acclimated and maintained in indoor pens, and with close association and interaction with people. This is essential for habituating animals to daily routines that they are likely to experience during experimental procedures, such as weighing, feeding, measuring of food and water use, and faecal collections.

The metabolic cages should be 1 metre wide × 1.3 metres deep and have wall heights of 1.7 metres. This will provide animals with an area of 1.3 m². This area does not restrict kangaroo movements and they are able to lie down, stand, turn around and stretch upright. The cages need to be custom built to avoid injuries, with no sharp corners or edges internally. The length of the cage is a key factor for the minimisation of the risk of injury from impacts associated with animal movement. The cage length should be designed to allow animals only two steps, as the kangaroo form of locomotion has the most important power stroke at step three, when they begin to hop. Appropriate cage length therefore prevents animals from building sufficient speed or power in their step that might otherwise cause significant injury and distress for animals and could jeopardise the health and safety of personnel.

During training and acclimation to metabolic cages, the animals should be offered a 'resting board' of smooth, thick plywood. This cage has been designed for and used successfully on kangaroos ≤ 30 kg body mass. Feed or water hoppers should be placed externally to avoid food or water spillage inside the cage. The floor of the cage should consist of an open mesh with wire gauge and mesh aperture as determined by the species (Barboza & Hume 1992a,b; Barboza 1993). This caging is essential to allow urine and faeces to pass from the cage for collection via a sliding tray and funnel system that separates urine. Urine is funnelled to measuring cylinders containing a small amount of paraffin to prevent urine evaporation.

Diets for captive macropodoids

Fresh water must be available ad libitum in each enclosure. Ideally, animals should be held in enclosures with sufficient grasses and browse for them to eat on a daily basis. The grazing and browsing macropodids need supplementary feed, with compressed lucerne cubes and steamed and rolled oats or macropodid pellets provided ad libitum with a weekly supply of food items such as green, leafy vegetables and/or carrots and apples. These can often be obtained free of charge as old stock from vegetable markets. Do not feed bread in any form as it clogs the teeth and predisposes animals to lumpy jaw. Do not feed fresh oats, as the seeds are sharp and can penetrate the oral epithelium and allow invasion of lumpy jaw pathogens. Musky rat-kangaroos and potoroos require fruit as well as supplements of protein-rich foods such as insects, dog food, cheese and eggs. Full details of appropriate diets for all species can be found in Jackson (2003).

Routine sample/data collection

Small tissue samples for DNA are commonly collected from the ear in an area without large blood vessels (for adults and pouch-young where the ears are free of the head) or tail tips (small pouch-young). Samples are best stored in 70% ethanol for DNA studies, or snap frozen. Blood is generally taken from the lateral tail vein in heparinised syringes or vacutainer blood collection tubes, but the latter is not suitable for the smaller macropodids as the blood is drawn up into the vacutainer under high pressure and can cause bruising. Pouch-young are often collected whole for genetic studies or from excess animals during cross-fostering procedures. Faecal samples are easily collected for dietary analysis or for faecal DNA identification of individuals.

Standard morphometric measurements include body weight, head length, forearm length, forearm diameter (in males) and hind foot length. Head and hind foot length are the usual measurements taken to construct growth curves of pouch-young for the purposes of age estimation. Pouches are usually checked for an indication of reproductive status. Juvenile females who have never bred have extremely shallow (thimble-sized) and clean pouches with inverted teats. Females who have bred previously but are not about to give birth have dirty pouches. Females who are soon to give birth have clean, pink pouches and the pouch-young will be found in the pouch after birth. If the young is living outside of the pouch but is still sucking, or if the pouch-young has been recently lost, the pouch will contain one elongated teat with an enlarged mammary gland. If the pouch has been empty for some time, the teat and mammary gland will both be regressing.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Very nervous animals or animals being transported for long distances may sometimes be sedated with 1 mg/kg diazepam in the hind leg muscle. See also 'Capture, handling, marking for identification, transport' There have been no pharmacokinetic studies on any analgesics for this species, thus if pain relief is required you must seek veterinary advice (e.g. zoo or wildlife veterinarian).

Macropodids can be anaesthetised using Zoletil® (3 mg/kg, IM) to sedate them, followed by isoflurane delivered via mask by a veterinary anaesthetic machine operated according to the manufacturer's instructions. (Further information on anaesthesia of macropodids is provided in Vogelnest and Portas 2008). Depth of anaesthesia is regularly monitored by palpebral and pedal reflex and regularity and depth of breathing. To minimise the ataxia associated with recovery particularly after using Zoletil®, place the animal in a large wool sack (kangaroo) or hessian sack (small wallabies) placed in a covered and protected area. One side of the wool sack should be open so that once the kangaroo has recovered sufficiently, the animal will be able to stand and escape through the opening.

Hypothermia can be used as an anaesthetic agent in neonatal and early postnatal marsupial young (Renfree 2002). The young is gently removed from the teat, and after weighing and measuring the animal is placed on a clean saline-moistened swab. Once cleaned with sterile saline, the pouch-young is placed on another saline-moistened swab that lies on a bed of ice. The young should be turned during cooling and as soon as movements cease, placed on a stainless steel plate that rests on a dish of crushed ice. The heart rate slows to about 25 beats per minute or less, and breathing may cease. Apnoea is not serious in very small pouch-young as cutaneous gas exchange is apparently sufficient, so long as the young is kept moist. After the surgery is complete, the young is best slowly warmed in the hand. The movements gradually return, and the first large breath is frequently accompanied by a noticeable change of colour. The young tolerate surgery remarkably well and show no signs of pain or distress. Successful reattachment to the teat is essential for their continued growth. Although pouch-young of less than a week can re-attach themselves, smaller pouch-young are assisted by gently pushing the teat into the mouth with the softened end of a match (Mellor 2002). They show no effects of the hypothermia and healing is rapid with no sign of the incision within two days.

In older young of over 50–60 days old, gaseous anaesthesia should be administered using the barrel of a small syringe as a mask, or a small animal rubber mask, using various sizes to suit the size of the young. Isoflurane (2–4 % in oxygen) with a flow rate of 0.5–1.0 L/minute provides a good level of anaesthesia and recovery is rapid. Again, the young are replaced on the teat when fully active. Replacement on the teat is often the most hazardous and difficult part of the procedure and should only be carried out by handlers trained in the method.

Humane killing

Macropodoids can be humanely killed using an overdose of sodium pentobarbitone (150 mg/kg) injected IV or IP. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution. Alternatively, if required because of specific experimental protocols where anaesthesia may impact on results, cervical dislocation by a sharp blow to the base of the skull can be used. However cervical dislocation should only be carried out by experienced people with sufficient strength to kill the animal with one blow. It is recommended that the use of gunshot or captive bolt firearms be considered for the euthanasia of large animals, especially in the field. Small, early stage pouch-young (for most medium to large species this means animals with up to 2 cm head length and 8 g body weight) can be anaesthetised by cooling then decapitated using a sharp scalpel blade. Young over 40 days old can be killed by an overdose of sodium pentobarbitone, made up to 60 mg/mL in sterile saline (150 mg/kg, IP). Euthanasia of pre-furred pouch-young can be accomplished by an overdose of sodium pentobarbitone (150 mg/kg, IP).

Health issues, disease control and zoonoses

Macropodoids can suffer from toxoplasmosis, hydatidosis, coccidiosis, necrobacillosis, capture myopathy (as described in 'Capture, handling, marking for identification, transport'), shock, hyperthermia and hypothermia. Orbivirus can cause blindness and death but is not well documented. Further information is available in Appendix IV, Ladds 2009 and Vogelnest & Portas 2008.

Animals being brought into captivity from the wild should be 'backstriped' with ivermectin or moxidectin (applied to the skin on the dorsal surface) to rid them of endo- and ectoparasites and this should be repeated annually. If there is a parasite problem effective anthelmintics should be applied using an appropriate and effective regimen that may require 3–4 treatments on a weekly basis. Animals should be vaccinated annually against tetanus using, for example, Tet-Tox™, at a dose of approximately 0.1 mL/5 kg bodyweight, IM. Previously, annual SC vaccination with Footvax® assisted with prevention of lumpy jaw (Blanden, Lewis & Ferrier 1987, Blyde 1994); however, this vaccine is no longer available. Tylan® (tylosin) is a macrolide antibiotic that has some efficacy in treatment after the lumpy jaw lesion is cleaned.

Specific breeding requirements

Some of the macropodids (such as swamp wallabies) may stop breeding due to stress if they are handled too often. Swamp wallabies also need to be caught carefully, as they are susceptible to stress of capture even after long periods of captivity. Musky rat-kangaroos are the only macropodids which routinely give birth to twins.

Institutions and individuals with specialised expertise

Institutions

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Bibliography for macropodoids

- Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.
- Barboza, PS 1993, 'Digestive strategies of the wombats: feed intake, fiber digestion, and digesta passage in two grazing marsupials with hindgut fermentation', *Physiological Zoology*, vol. 66, pp. 983–99.
- Barboza, PS & Hume, ID 1992a, 'Digestive tract morphology and digestion in the wombats (Marsupialia: Vombatidae)', *Journal of Comparative Physiology*, vol. 162, pp. 552–60.
- Barboza, PS & Hume, ID 1992b, 'Hindgut fermentation in the wombats: two marsupial grazers', *Journal of Comparative Physiology*, vol. 162, pp. 561–66.
- Blanden DR, Lewis PR & Ferrier GR 1987, 'Vaccination against lumpy jaw and measurement of antibody response in wallabies (*Macropus eugenii*)', *Veterinary Record*, vol. 121, pp. 60–2.
- Blyde, D 1994, 'Management and diseases of macropods', in Proceedings 233. *Wildlife: the T. Hungerford refresher course for veterinarians, Post Graduate Committee in Veterinary Science, The University of Sydney*. pp. 247–51.
- Coulson, G 1996, 'A safe and selective draw-string trap to capture kangaroos moving under fences', *Wildlife Research*, vol. 23, pp. 621–27.
- Coulson, G, Poiani, A & Nave, CD 2003 'A modified draw-string trap for capture of kangaroos at fences', *Australian Mammalogy*, vol. 25, pp. 177–82.
- Dawson, TJ 2012, *Kangaroos*, 2nd edn, CSIRO Publishing, Melbourne.
- Di Stefano, J, Moyle, R & Coulson, G 2005, 'A soft-walled double-layered trap for capture of swamp wallabies, *Wallabia bicolor*', *Australian Mammalogy*, vol. 27, pp. 235–38.
- Hickford, D, Frankenberg, S & Renfree, MB 2010, 'The tammar wallaby, *Macropus eugenii*: a model kangaroo for the study of developmental and reproductive biology', in *Emerging Model Organisms*, Volume 2, Cold Spring Harbor Laboratory Press, Cold Spring Harbor, pp. 449–94.
- Higginbottom, K 1989, 'Macropod studies at Wallaby Creek.VII. Capture of wild red-necked wallabies by "blow-darting"', *Australian Wildlife Research*, vol. 16, pp. 173–78.
- Hume, I 1999, *Marsupial Nutrition*, Cambridge University Press, Cambridge.
- Jackson, S 2003, 'Macropods', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 245–96.
- King, W, Wilson, M, Allen, T, Festa-Bianchet, M & Coulson, G 2011, 'A capture technique for free-ranging eastern grey kangaroos (*Macropus giganteus*) habituated to humans', *Australian Mammalogy*, vol. 33, pp. 47–51.
- Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne, pp. 511–512.
- Munn, AJ, Banks, PB & Hume, ID 2006, 'Digestive plasticity of the small intestine and fermentative hindgut of a marsupial herbivore, the Tammar Wallaby (*Macropus eugenii*)', *Australian Journal of Zoology*, vol. 54, pp. 287–91.
- Munn, AJ, Clissold, F, Tarzisz, E, Kimpton, K, Dickman, CR & Hume, ID 2009, 'Hindgut plasticity in wallabies fed unchopped or chopped and pelleted lucerne hay: fibre is not be the only factor', *Physiological and Biochemical Zoology*, vol. 82, pp. 270–79.
- Munn, AJ & Dawson, TJ 2003, 'How important is milk for near-weaned red kangaroos (*Macropus rufus*) fed different forages?' *Journal of Comparative Physiology B*, vol. 173, pp. 141–48.
- Munn, AJ & Dawson, TJ 2001, 'Thermoregulation in juvenile red kangaroos (*Macropus rufus*) after pouch exit: higher metabolisms and evaporative water requirements', *Physiological and Biochemical Zoology* vol. 74, pp. 917–27.

- Munn, AJ, Dawson, TJ & McLeod, S 2010, 'Feeding biology of two functionally different foregut-fermenting mammals, the marsupial red kangaroo (*Macropus rufus*) and the ruminant sheep (*Ovis aries*): how physiological ecology can inform land management', *Journal of Zoology*, vol. 282, pp. 226–37.
- Pollock, DC & Montague TL 1991, 'A new trap trigger mechanism for the capture of swamp wallabies, *Wallabia bicolor* (Marsupialia: Macropodidae)', *Wildlife Research*, vol. 18, pp. 459–61.
- Renfree, MB & Tyndale-Biscoe, CH 1978, 'Manipulation of marsupial embryos and pouch young', in JS Daniel Jnr (ed.), *Methods in mammalian reproduction*, Academic Press, New York, pp. 308–31.
- Renfree MB 2002, 'Hypothermic anaesthesia of early postnatal marsupial pouch young', in M Fisher, J Marbrook & G Sutherland (eds), *Learning, animals and the environment: changing the face of the future*, Royal Society of New Zealand, Wellington, pp. 125–7.
- Roberts, MW, Herbert, CA, Neaves, L & Claassens, R 2009, 'Darting eastern grey kangaroos (*Macropus giganteus*): A protocol for free ranging populations', in G Coulson & MDB Eldridge (eds), *Macropods. The biology of kangaroos, wallabies and rat-kangaroos*, CSIRO Publishing, Melbourne, 328–40.
- Robertson, G & Gepp, B 1982, 'The capture of kangaroos by stunning', *Australian Wildlife Research*, vol. 9, pp. 393–6.
- Tyndale-Biscoe, H & Renfree, M 1987, *Reproductive physiology of marsupials*, Cambridge University Press, Cambridge, pp. 476.
- Van Dyck, S & Strahan R (eds) 2008, *The mammals of Australia*, 3rd edn. Reed New Holland, Sydney.
- Vogelnest, L & Portas, T 2008, 'Macropods', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp 133–226.
- Williams, A & Williams, R 1999, *Caring for kangaroos and wallabies*, Kangaroo Press, Sydney.

Medium and larger possums

The medium- to large-sized possums weigh from 700 g to 5 kg and include the brushtail possums (three species) and cuscuses (two species), the scaly-tailed possum, the ringtail possums (seven species) and the greater glider. All are forest or woodland dependent, being arboreal or semi-arboreal and herbivorous. All are folivorous or partially folivorous. Some of these possums are rainforest specialists, while others occur in a range of habitat types. Most of them are nocturnal.

While this section is an overview of medium and larger possums, more specific information is available in the two sections, 'Brushtail possums (*Trichosurus* spp.) and cuscuses' and 'Ringtail possums and the greater glider'. Detailed information on the various species, including their basic biology, is provided in Van Dyck & Strahan (2008).

Medium and larger possums – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Common brushtail possum, <i>Trichosurus vulpecular</i> .	Not listed.	Widespread in eastern Australia, from Cape York in Qld to Vic., as well as south-eastern SA, Tas., south-western WA, northern WA and the north of NT. Limited in Central Australia.	Generalist folivore/herbivore, feeding on foliage, grasses, fruit, blossom, occasional small vertebrates and invertebrates.
Mountain brushtail possum and short-eared brushtail possum, <i>Trichosurus cunninghami</i> and <i>T. caninus</i> .	Not listed.	Moister forests of eastern Australia, from south-eastern Qld into the eastern half of Vic.	<i>Acacia dealbata</i> and <i>A. mearnsii</i> foliage and fungi, are very important dietary items. In addition, they feed on foliage of some other tree species such as eucalypts and fruit.
Common spotted cuscus, <i>Spilocuscus maculatus</i> .	Not listed.	Forests of the Cape York Peninsular, north of Coen, Qld.	Herbivorous, feeding on fruits, flowers, and foliage. Possibly partially carnivorous.
Southern common cuscus, <i>Phalanger mimicus</i> .	Not listed.	Limited distribution in forests on east of Cape York Peninsular, from Iron Range to Mcllwraith Range, Qld.	Herbivorous, feeding on fruits and foliage.
Scaly-tailed possum, <i>Wyulda squamicaudata</i> .	Not listed.	Kimberley in north-western WA, with distribution in coastal and adjacent areas.	Herbivorous, feeding on foliage, flowers, herbs and seeds.
Common ringtail possum, <i>Pseudocheirus peregrinus</i> .	Not listed.	Widespread in forests along the eastern part of Australia, from Cape York to Vic., into south-eastern SA and throughout Tas.	Specialist folivore, primarily feeding on foliage including <i>Eucalyptus leptospermum</i> and other native trees, plus fruits and flowers.
Western ringtail possum, <i>Pseudocheirus occidentalis</i> .	Vulnerable.	Confined to woodlands in south-western WA. Distribution has declined substantially.	Specialist folivore, feeding primarily on foliage of Myrtacea.
Lemuroid ringtail possum, <i>Hemibelideus lemuroides</i> .	Not listed.	Very restricted distribution, occurring in rainforest above 300 metres in north-eastern Qld.	Specialist rainforest folivore.
Rock ringtail possum, <i>Petropseudes dahlia</i> .	Not listed.	Occurs in rocky outcrops in northern Australia, including the Kimberley, NT and just across the border into north-western Qld.	Feeding on foliage, fruit and flowers from a range of local native trees.

Medium and larger possums – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Green ringtail possum, <i>Pseudochirops archeri</i> .	Not listed.	Very restricted distribution, occurring in rainforest above 450 metres in north-eastern Qld.	Specialist rainforest folivore, feeding on fruit occasionally.
Daintree River ringtail possum, <i>Pseudochirulus cinereus</i> .	Not listed.	Very restricted distribution, occurring in rainforest above 420 metres in north-east Qld.	Specialist rainforest folivore, feeding on fruit occasionally.
Herbert River ringtail possum, <i>Pseudochirulus herbertensis</i> .	Not listed.	Very restricted distribution, occurring in rainforest above 350 metres in north-eastern Qld.	Feeding on foliage, fruit and flowers.
Greater glider, <i>Petauroides volans</i> .	Not listed.	Widespread in eucalypt dominated forests along the Great Dividing Range in eastern Australia, occurring from north-eastern Qld through into the eastern half of Vic.	Specialist folivore, feeding on foliage of a limited range of <i>Eucalyptus</i> spp.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999 - List of Threatened Fauna*

A number of techniques for capture, handling, sampling and sedation are applicable to most of the species of medium and larger possums. These are outlined below, with more specific information in the following sections on the 'Brush-tail possums (*Trichosurus* spp.) and cuscuses' and the 'Ringtail possums and the greater glider'. Extensive information on captive management of the various species is also provided in Jackson (2003), but researchers are reminded that it is very important to consult experts in dealing with individual species and the appropriate literature. Information on some of the possum species is very well known, while for others, there is only limited information.

Capture, handling, marking for identification, transport

Many of the medium and larger possums, such as brush-tail possums and some of the ringtail possums can be captured in cage traps (see also separate sections on different groups of possums). At least half of the trap (roof, sides and rear end) should be covered, for example, with heavy plastic or light aluminium sheet to provide protection from adverse weather and to provide some visual protection from predators.

Possums are nearly all nocturnal; therefore traps should be set around dusk and cleared commencing, or before, dawn. In hotter weather, traps should be positioned to ensure they will not be exposed to hot sun. The number of traps set should be limited to ensure that they can be cleared before animals become overheated as the daytime environmental temperature increases. In extremely cold weather, especially when heavy rain or snow is forecast, traps should be closed. For handling and/or transport, animals should be transferred directly from traps into bags that should be large enough to fit over the end of the trap. Possums can also be removed from traps by firmly grasping the **base** of the tail. If this approach is used, gloves are recommended but be aware that gloves reduce dexterity. Possums should not be carried in traps as they will often become alarmed and charge into the sides of the traps, which may cause damage to their nose or face. One issue with trapping for some possum species is the possibility of repeated recapture of some individuals on consecutive nights ('trap-habituated' animals).

Individual animals should only be subjected once to procedures such as full handling and measurement during any particular field trip unless specifically required under the project objectives. To avoid such 'reuse', animals should be marked so that any individuals that are recaptured during the trip can be identified immediately and released without further handling; however, for short-term projects, marking can be achieved non-invasively, for example, using waterproof marker pens on ears, or trimming some fur from the top of the head. (See below for information about methods for marking.) If individuals are captured on two consecutive nights, it is recommended that traps in the vicinity be closed for a night to allow the animals time to forage. This

is especially important during periods when females are under high lactational load such as when they are carrying large pouch-young or small back-young, or during periods of extreme weather conditions such as very hot, very dry or very wet and cold conditions. Most of the medium- to large-sized possums can also be captured by darting, using small purpose-designed tranquiliser darts. This should not be attempted by anyone until they have been trained by an expert and have developed substantial expertise, particularly for smaller species. Seek advice from zoo veterinarians for how to pursue training.

In the field, animals being held during the day for recovery and/or awaiting release should be held in their bags inside a suitable secure, cool, quiet and dark area. In the event of very hot weather, the animals should be transferred from their bags into cage traps or suitable crates to prevent them from overheating, and they should be checked regularly for signs of heat stress such as panting and salivating onto their wrists. If these signs appear, cool the wrists and face with cold water. For field studies, it is strongly recommended that, whenever possible, possums should be released at dusk, because they are nocturnal, and if released during the day they often sit in an exposed position as they appear unable to find their way back to a den site. Possums should be released at the point of capture.

Medium and larger possums can be marked by tattooing numbers into their ears with black ink, using a tattoo gun or tattoo number punch kit (smaller species) such as those used by veterinarians. Tattooing should occur after swabbing the ear with 70% ethanol, preferably while animals are sedated (see 'Anaesthesia, analgesia and sedation'). Microchips (for example, passive integrated transponder tags; 'PIT' tags) are also suitable, and should be inserted under the skin between the shoulder blades. Ear tags are not recommended, especially for wild animals, as these invariably tear out, which means the animal can no longer be identified. In addition, after tag loss, large tears often develop in the ears, which may impair the possum's ability to detect predators.

For relatively short transport distances, the animals should be transported in individual cloth or hessian bags (except for dependent young who should be placed in the bag with their mother), firmly tied with string. The bags should be placed so that the animals are not cramped. If a large number of animals are being transported, place the bags in open boxes (two or three per box) to prevent animals from rolling around or being smothered.

In warmer weather, it is very important to transport animals with the air conditioner running in the vehicle to prevent the animals from becoming overheated. Check animals to determine whether they are overheated by looking for signs such as panting and salivation onto wrists. If the animals are overheated, cool them immediately by applying cold water over the wrists and the sides of face and then transfer the animals into a suitable, well-aired crate. Also, ensure that animals do not become chilled if they are wet and the weather is cold by transferring them into dry bags. If being transported for long distances or many hours, animals should be placed in appropriate animal crates (seek advice from zoo experts), as they may chew their way out of bags. Further information on transport for individual species is provided in Jackson (2003).

Captive husbandry

Specific information for the different groups (brushtail and ringtail possums) is available in the following two sections. In addition, extensive information on the captive husbandry requirements for the medium and larger possums is provided for individual species in Jackson (2003).

It should be noted that the diet of these species is quite variable, and some species that are folivorous will require considerable effort to keep in captivity. Expert advice should be sought on how to maintain folivores, and researchers should be aware that food-tree preferences can vary considerably across geographic location, thus local knowledge will also be important. In their enclosures, virtually all possum species require suitable denning sites such as nest boxes, hollow branches and tree limbs for climbing. When brought in from the wild, possums may carry parasites such as mites and ticks. Nests should therefore be checked monthly. Bedding should be changed and animals treated if necessary. Expert veterinarian advice on captive husbandry for medium and large possums should be sought.

Routine sample/data collection

Body mass can be measured while the animals are in a bag. However, collection of other standard morphometric data that is best conducted while animals are sedated includes measurement of head length, head width, hind foot length and wear patterns on the teeth in the upper jaw (for example, to indicate age; see Jackson 2003) as well as checking and measurement of pouch-young and fitting radio transmitters, (see 'Anaesthesia, analgesia and sedation').

Tissue samples for DNA can be collected from near the margins of ears, for example using a 2 mm ear punch or biopsy punch after swabbing the site with 70% ethanol. Use a torch behind the ear to avoid small blood vessels.

Faecal pellets can often be collected fresh from the holding bag or beneath the trap in which the animal was caught.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. For restraint for handling, tattooing, minor procedures such as collection of small tissue samples for DNA, blood sampling, fitting radio transmitters correctly, sedate with diazepam (0.5–1.0 mg/kg, IM), Zoletil® (tiletamine/zolazepam, at a dose rate of 5–7 mg/kg) or Alfaxan® (5 mg/kg body weight) injected IM into a hind leg.

If anaesthetised or sedated in the field, possums should be held in a quiet location in a bag inside a crate for recovery, and they must be held until they are fully awake and competent to climb. Take extra care to monitor animals regularly during recovery in hot weather to make sure they are not becoming overheated. In the laboratory and for surgical anaesthesia, use isoflurane in oxygen (5% for induction, 1–2% for maintenance), with an oxygen flow rate of 1 L/minute administered via a mask. Maintenance is generally with a face mask, because the animals' narrow mouth gape makes endotracheal intubation difficult. Further information on anaesthesia of possums is provided in Vogelnest & Woods (2008). There have been no pharmacokinetic studies on any analgesics for these species, thus if pain relief is required you must seek expert advice from a zoo or wildlife veterinarian.

Humane killing

Medium and larger possums can be humanely killed using an overdose with sodium pentobarbitone (150 mg/kg) injected IP, IC or IV. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Due to the large number of different species, it is advisable to seek expert veterinary advice on the health and diseases of possums (e.g. veterinarians in major zoos). Information is also provided in Vogelnest & Woods (2008) and Jackson (2003). All traps and bags should be thoroughly washed and disinfected when being moved from one field site to another, to avoid inadvertent movement of disease agents or parasites among populations or species.

Specific breeding requirements

Possums vary considerably in their breeding biology. Some are relatively easy to breed in captivity. Expert advice should be sought (e.g. from major zoos with captive colonies). Information on breeding requirements of possums is also provided in Jackson (2003).

Institutions and individuals with specialised expertise

Institutions

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Bibliography for medium and larger possums

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Goldingay, RL & Jackson, SM (eds) 2004, *The biology of possums and gliders*, Surrey Beatty and Sons, Sydney.

Jackson, S 2003, 'Possums and gliders' in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Johnson, R & Hemsley, S 2008, 'Gliders and possums', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 395–437.

Kerle, A 2003, *Possums: the brushtails, ringtails and greater glider*, UNSW Press, Sydney.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Van Dyck, S & Strahan R (eds) 2008, *The mammals of Australia*, 3rd edn. Reed New Holland, Sydney.

Brushtail possums (*Trichosurus* spp.) and cuscuses

This section provides more specific detail for the brushtail possums and cuscuses. Read in conjunction with the previous section, 'Medium and larger possums'.

For many research, management and husbandry techniques, the common brushtail possum (*T. vulpecula*), the mountain brushtail possum or bobuck (*T. cunninghami*) and the short-eared possum (*T. caninus*) can all be treated in a similar fashion (see 'Specific details for capture, handling, marking for identification, transport' below). While handling and management requirements of cuscuses are quite similar to those of brushtail possums, expert advice should be sought (e.g. from a zoological institution that has held these species).

Specific details for capture, handling, marking for identification, transport

Wild brushtails can be caught in wire-mesh cage traps (approximately 30 cm × 31 cm × 63 cm) set on the ground and baited with half an apple or pear smeared with peanut paste, or bait balls of rolled oats, peanut paste and golden syrup. For northern populations, traps must have Coopex® (or similar) applied near traps to prevent ant problems. At sites where field work is conducted frequently, some individual brushtail possums may become habituated to entering traps. If individuals are captured on two consecutive nights it is recommended that traps in the vicinity of these be closed on the following night to allow the animals time to forage, especially during periods when females are under high lactational load such as when they are carrying large pouch-young or small back-young or during periods of extreme weather conditions such as very hot, very dry or very wet and cold conditions. Animals should be transferred directly from traps into large hessian or cloth sacks (these should be large enough to fit over the end of the trap) for handling and/or transport.

Wild brushtails are best handled in a bag (see 'Medium and larger possums'). If not sedated, animals can be picked up by grasping them firmly with one hand around the shoulders and the other holding the base of the tail, but be aware that most wild brushtails will struggle vigorously and attempt to bite and scratch, thus wearing heavy gloves is recommended. However, this method of handling appears to be more distressing for the animals, thus if it is not essential to handle them in this manner it is preferable to conduct all handling inside a bag. Tame captive animals can be picked up briefly by the base of the tail for example, for transfer into a handling bag. Additional information is provided in Jackson (2003). For minor procedures such as collecting tissue for DNA analysis, collecting blood samples, fitting radio-collars, measuring pouch-young and tattooing, the animals can be sedated (see 'Anaesthesia, analgesia and sedation' in the 'Medium and larger possums' section).

Information about transport of possums is provided in 'Capture, handling, marking for identification, transport' in the 'Medium and larger possums' section, and in Jackson (2003). Expert advice should also be sought.

Captive husbandry

Brushtails (and cuscuses) habituate well to captivity and are relatively easy to house and maintain in good health. Suitable nest boxes, branches for climbing and an appropriate diet are key elements of good husbandry and maintenance. The degree of sociality varies between, and within, the various species, from pairing and den sharing to solitary. Solitary species will tolerate group housing provided there is sufficient space and surplus den sites. For research purposes, animals can be housed individually (especially adult males) in wire-mesh cages, such as standard bird aviaries (approximately 1.5 metres wide × 0.75 metres deep × 1.9 metres high) with a solid roof, rear and side walls, and a wire-mesh front. It is essential to protect smaller cages such as this from the sun in hot weather, as animals can rapidly become heat stressed. However, wherever possible, use larger cages to allow inclusion of tree trunks and other structures for climbing and general environmental enrichment. If animals are being held long term, natural substrate with ground cover should be provided, as brushtails will browse ground cover. Water must be provided at all times. Captive brushtails usually habituate well. In very hot weather they will happily accept frozen grapes and chilled fruit juice, which can assist with cooling them. Multiple animals should be housed in larger wire-mesh cages and allow approximately 1.5–2 m² floor area per animal.

If animals are housed as a colony it is essential that there are several more nest boxes available than the number of animals to ensure that subordinates always have easy access to a den site. Such cages should also have some shade cloth over the roof to prevent nest boxes from being directly exposed to the hot sun.

During long periods of extremely hot weather it is essential to monitor animals several times each day for heat stress. Captive animals will readily accept cold fruit juice, provided directly to individuals in their nest boxes, and animals can be cooled with water using a small handheld spray bottle. Also fit cages with a water mister system that can be turned on to help keep animals cool.

Extensive information on the captive husbandry requirements for the brushtail possums and cuscuses is provided for individual species in Jackson (2003). Advice can also be sought from a researcher or experts who have held these species in captivity for research purposes. If intensive research involving the holding of animals in very small cages in an animal facility is intended, seek advice from researchers or experts who have been involved with such projects. Longer-term holding of wild species such as possums in very small cages should not be considered unless required to meet the specific aims of the project, and only if these can be fully justified to the satisfaction of the animal ethics committee.

Diet for captive brushtail possums (*Trichosurus* spp.) and cuscuses

Captive diets should include fresh food such as carrot, sweet potato, apple, other fruit and vegetables, suitable kibble, and browse. Provide leafy green material and, if possible, at least some native browse that is known to be preferred by the individual possum species, for example, *Acacia dealbata* or *A. mearnsii* foliage for mountain brushtails or short-eared possums.

Routine sample/data collection

Routine data should include body mass, head length, head width, hind foot length, sex, and age (using wear patterns on the teeth in the upper jaw if possible, see Winter 1980, Jackson 2003). If possible, data about reproductive status should also be collected including pouch condition, testis length, width and depth, and the head length and sex of pouch-young.

Blood samples (2–3 mL) can be collected from the central tail vein (a few centimetres distal to the bare area on the underside) or from lateral tail or femoral veins using a 23- to 25-gauge needle. Researchers who have not previously performed this procedure should seek advice from experts with experience before attempting to collect blood samples from possums.

For information about collecting samples for DNA analysis, see 'Routine sample/data collection' in the 'Medium and larger possums' sections.

Anaesthesia, analgesia and sedation

For information, see 'Anaesthesia, analgesia and sedation' in the 'Medium and larger possums' section.

Humane killing

For information, see 'Humane killing' in the 'Medium and larger possums' section.

Health issues, disease control and zoonoses

For information, see 'Health issues, disease control and zoonoses' in the 'Medium and larger possums' section.

Mycobacterium ulcerans, a potentially zoonotic organism, has been recorded in brushtail possums from Victoria.

Bibliography for brushtail possums (*Trichosurus* spp.) and cuscuses

Jackson, S 2003, 'Possums and gliders' in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Winter J. W. (1980) Tooth wear as an age index in a population of the brush-tailed possum, *Trichosurus vulpecula* (Kerr). *Australian Wildlife Research* 7, 359–63.

Ringtail possums and the greater glider

This section provides more specific detail for the ringtail possums and the greater glider. Read in conjunction with the section, 'Medium and larger possums'.

Specific capture, handling, marking for identification, transport

Some of the species of wild ringtail possums (and the greater glider) can be caught in wire-mesh cage traps set in trees (see the 'Medium and larger possums' section). Traps must be covered with plastic to provide protection from rain and this also serves to provide the animals with a retreat area in the event of predators visiting the trap. Traps can be baited with a bait ball of rolled oats, peanut butter and golden syrup, or a third of an apple or pear smeared with peanut butter. Traps are set 1–2 hours before dark and should be cleared commencing at dawn on the following morning.

In hotter weather, care must be taken to ensure that traps are not placed in positions where they will receive full sunlight, as this could result in animals becoming overheated. The trap-line should be closed in extreme weather conditions such as very cold and wet weather, severe storms or very high temperatures. Such conditions may prevent you from working safely in the forest, thus could cause delays in clearing traps that could impact on trapped animals.

Ringtail possums can also be caught by hand, during the day if they are in low nests or at night from trees. Such hand captures are generally conducted when animals are less than 3 metres above the ground. This involves shaking the tree and catching the animal as it falls using a hand net, or if the animal is very low, stalking and grasping the animal by the **base** of the tail. **Never** hold or grab animals by the tip of the tail as it can be de-gloved. Ringtail possums can also be caught using a specifically designed noosing pole, with the noose designed to go around the neck, but it must also be placed across the chest and under one armpit, so that strangulation is avoided. Substantial expertise is required for this method. People with inexperience in the capture of possums must seek expert advice.

Greater gliders are caught using a variety of techniques, depending on forest type. Consult experts with relevant experience. One approach is to hook a stout pole onto the greater glider's branch and shake the branch vigorously while holding a spotlight on the animal. The greater glider will usually then launch into a glide and safely reach the ground.

After capture, animals should be transferred immediately into soft cotton bags for handling and transport. Both ringtail possums and greater gliders can be captured by darting (see the 'Medium and larger possums' section), but because of their relatively small size this should only ever be attempted by a very experienced operator.

Wild ringtail possums and greater gliders are best handled in a soft cotton bag such as a pillow case or calico bag. For minor procedures such as collecting tissue for DNA analysis, collecting blood samples, fitting radio-collars, measuring pouch-young and tattooing, the animals can be sedated (see 'Anaesthesia, analgesia and sedation' in the 'Medium and larger possums' section). If not sedated, animals can be picked up by grasping them firmly with one hand around the shoulders and the other holding the base of the tail. Tame captive animals can be picked up briefly by gently placing the hands beneath the animal while it is in a sitting position, and cradling it against the handler's chest, or by picking up briefly by the base of the tail, for example, for transfer into a handling bag. As for any species being held or transported in a bag, animals must be carefully monitored for heat stress in warmer conditions.

Information about transport is provided in 'Capture, handling, marking for identification, transport' in the 'Medium and larger possums' section, and in Jackson (2003). Expert advice should also be sought.

The key issue is to avoid animals from overheating in hot weather (see 'Capture, handling, marking for identification, transport' in the 'Medium and larger possums' section) or from becoming chilled if they are wet and the weather is cold.

Ringtail possums can be marked by tattooing their ears using a small number punch tattoo system, but greater gliders tend to have dark pigment in their ears, making this less effective. All can be marked with microchips (for example, passive integrated transponder tags; 'PIT' tags; see 'Capture, handling, marking for identification, transport' in the 'Medium and larger possums' section). Ear tags should be avoided, as they tend to pull out and leave tears in the ears.

Captive husbandry

Ringtail possums and greater gliders habituate quite well to captivity. While some species are relatively easy to house and maintain in good health, the strictly folivorous species require extra care and attention with diet.

Virtually all ringtail species, and the greater glider, are highly arboreal. Networks of branches should therefore be provided in cages. Suitable nest boxes and an appropriate diet are key elements of good husbandry and maintenance. The degree of sociality varies between the species, from pairing and den sharing to solitary. Solitary species will tolerate group housing provided there is sufficient space and den sites, however check tolerance with an expert for individual species. For research, pairs of animals (for example, common ringtail possums) can be housed in wire-mesh cages, such as standard bird aviaries. These should be approximately 1.5 metres wide × 0.75 metres deep × 1.9 metres high, with a solid roof, rear and side walls, and a wire-mesh front. It is essential to protect the cages from the sun in hot weather, as animals will rapidly become heat stressed. Alternatively, multiple animals can be housed in larger wire-mesh cages, allowing approximately 1.5–2 m² of floor area per animal.

If animals are housed as a colony it is essential that there are several more nest boxes available than the number of animals to ensure that subordinates always have easy access to a den site. Such cages should also have shadecloth over at least part of the roof to prevent nest boxes from being directly exposed to the hot sun. During long periods of extremely hot weather, it is essential to monitor animals several times each day for heat stress. Animals can be cooled with water using a small handheld spray bottle, and cages should also be fitted with water misters that can be turned on during very hot weather.

Information on captive husbandry requirements is provided in Jackson (2003). Advice can also be sought from researchers or experts who have held these species in captivity for research purposes.

Diets for captive ringtail possums and the greater glider

Captive diets for common ringtail possums should include fresh fruit (but not citrus) and vegetables, suitable kibble and browse. Seek expert advice for each different species. The greater glider and the ringtail species that are strictly folivorous must be fed **fresh** browse of locally-preferred tree species, which involves considerable time and effort for handlers. Because of the animals' specialist dietary needs and as food tree preferences may vary regionally, it is important to consult an expert (preferably one with local experience) or a zoo with experience with that particular species before bringing animals into captivity, particularly when choosing browse plant species. Information on captive husbandry requirements is provided in Jackson (2003).

Routine sample/data collection

Routine data should include body mass, head length, head width, hind foot length, sex, and age using wear patterns on the teeth. Information on routine data to be collected for ringtail possums is provided in Pahl (1987) and Jackson (2003). If possible, data about reproductive status should be collected including pouch condition, testis length, width and depth and details of any pouch-young (number of young, head length and sex).

Blood samples of 0.5–1.5 mL can be relatively easily collected from the central tail vein or femoral veins. Expert advice should be sought if you have not previously performed this procedure.

For information about collecting samples for DNA analysis, see 'Routine sample/data collection' in the 'Medium and larger possums' sections.

Anaesthesia, analgesia and sedation

For information, see 'Anaesthesia, analgesia and sedation' in the 'Medium and larger possums' section.

Humane killing

For information, see 'Humane killing' in the 'Medium and larger possums' section.

Health issues, disease control and zoonoses

For information, see 'Health issues, disease control and zoonoses' in the 'Medium and larger possums' section.

Mycobacterium ulcerans is a potential zoonotic organism that has been recorded in common ringtail possums from Victoria.

Bibliography for ringtail possums and the greater glider

Jackson, S 2003, 'Possums and gliders' in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Pahl, L. (1987) Survival, Age-Determination and Population Age Structure of the Common Ringtail Possum, *Pseudocheirus-Peregrinus*, in a Eucalyptus Woodland and a Leptospermum Thicket in Southern Victoria. *Australian Journal of Zoology* 35(6) 625–639.

Smaller possums and gliders

In Australia, there are a number of small-sized possums (6–500 g) and gliders (10–700 g). The smaller possums and gliders include the pygmy-possums (five species), petaurid possums (six species), feathertail glider (one species) and the honey possum (one species). These species occur in locations ranging from forested habitats to the semi-arid zone, and there is considerable ecological variation between species. Some species are rainforest specialists, others occur in a range of habitat types. They are mostly nocturnal and many of the pygmy possums, which are quite small, enter torpor. Detailed information on the various species, including their basic biology, is provided in Van Dyck & Strahan (2008).

While this section is an overview of smaller possums and gliders, more specific information is available in the four following sections: ‘Petaurid possums: gliders, Leadbeater’s possum and the striped possum’, ‘Pygmy-possums’, ‘Feathertail glider’ and ‘Honey possum’.

Smaller possums and gliders – overview			
Name	Status under EPBC Act ^{1,2,3}	Distribution	Natural diet
Sugar glider, <i>Petaurus breviceps</i> .	Not listed.	Widespread from Kimberley, WA, through northern NT, northern Qld and in eastern Australia from Qld to NSW and Vic., south-eastern SA, and throughout Tas.	Nectar, pollen, exudates and arthropods.
Squirrel glider, <i>Petaurus norfolcensis</i> .	Not listed.	Eastern Australia from north Qld through NSW and into north-eastern Vic.	Nectar, pollen, exudates and arthropods.
Yellow-bellied glider, <i>Petaurus australis</i> .	Widespread, but vulnerable in the population of the Qld Wet Tropics.	Limited distribution in north-eastern Qld, with more extensive distribution in forests from south-eastern Qld through eastern NSW and Vic., with limited distribution in south-western Vic.	Nectar, pollen, exudates and arthropods.
Mahogany glider, <i>Petaurus gracilis</i> .	Endangered.	Very limited forest distribution near Cardwell north-eastern Qld.	Nectar, pollen, exudates and arthropods.
Leadbeater’s possum, <i>Gymnobelideus leadbeateri</i> .	Endangered.	Very limited distribution in forests of the central highlands of Vic.	Arthropods and exudates.
Striped possum, <i>Dactylopsila trivirgata</i> .	Not listed.	Rainforests and adjacent woodlands of Far North Queensland and Cape York.	Specialist feeder on wood-boring beetle larvae, plus other arthropods, nectar and sap.
Feathertail glider, <i>Acrobates pygmaeus</i> .	Not listed.	Widespread in forests of eastern Australia, from Cape York in Qld to Vic. and south-eastern SA.	Nectar, pollen and arthropods.
Honey possum, <i>Tarsipes rostratus</i> .	Not listed.	South-western WA.	Specialist feeder on nectar and pollen.
Eastern pygmy- possum, <i>Cercartetus nanus</i> .	Not listed. Listed in NSW ² .	South-eastern Qld, eastern NSW, Vic., south-eastern SA and Tas., in a range of forest types.	Nectar, pollen and arthropods.
Western pygmy-possum, <i>Cercartetus concinnus</i> .	Not listed.	South-western WA, southern SA, into north-western Vic. and limited distribution in south-western NSW, temperate semi-arid and arid regions.	Nectar, pollen and arthropods.
Little pygmy-possum, <i>Cercartetus lepidus</i> .	Not listed.	North-western Vic. and south-eastern SA, including Kangaroo Island, and widespread in Tas.	Nectar, pollen and arthropods.

Smaller possums and gliders – overview			
Name	Status under EPBC Act ^{1,2,3}	Distribution	Natural diet
Long-tailed pygmy-possum, <i>Cercartetus caudatus</i> .	Not listed.	Limited distribution in rainforests of north-eastern Qld.	Arthropods, nectar and pollen.
Mountain pygmy-possum, <i>Burramys parvus</i> .	Endangered.	Very limited distribution in alpine areas of NSW and Vic.	Bogong moths, arthropods, seeds and fruits of shrubs.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² *Threatened Species Conservation Act 1995*, NSW Department of Primary Industries

³ Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

A number of techniques for capture, handling, sampling and sedation are applicable to most of the species of smaller possums and gliders. These are outlined below, with more specific information in the following sections on the ‘Petaurid possums: gliders, Leadbeater’s possum and the striped possum’, ‘Pygmy-possums’, ‘Feathertail glider’ and ‘Honey possum’.

Extensive information on captive management of the various species is also provided in Jackson (2003), however researchers are reminded that it is very important to seek expert advice and consult relevant literature. Some of the possum species are very well known, while for others, there is only limited information.

Capture, handling, marking for identification, transport

Capture techniques for small possums and gliders vary considerably for different species and also across different habitat types. Expert advice from an experienced field biologist should be sought.

The most commonly-used methods are wire-mesh cage traps, small aluminium box traps (for example, Elliott traps), pitfall traps and nest boxes (for further information, see separate sections for ‘Petaurid possums: gliders, Leadbeater’s possum and the striped possum’, ‘Pygmy-possums’, ‘Feathertail glider’ and ‘Honey possum’).

When setting any type of trap, thermal extremes in the environment must be taken into consideration. In hotter weather, care must be taken to ensure that traps are not placed in positions where they will receive full sunlight, as this could result in animals becoming overheated. The number of traps set should always be limited to ensure that they can be cleared in a timely manner so that animals do not remain in traps for extended periods and become hyperthermic or hypothermic. Trap lines should be closed during extremely hot or extremely cold, wet weather conditions, as small animals can succumb rapidly. In addition, it is best to close traps if severe storms are forecast, as these may prevent you from working safely in the forest, and thus could delay trap clearing which could impact on the animals. It is usually important to cover traps to protect against sun or rain and to provide bedding material, unless local environmental temperatures are relatively close to the thermo-neutral conditions for the species. For nocturnal species, traps should be set around dusk and cleared commencing at dawn. If trapping is conducted during the day, the traps often need to be checked more regularly. If individuals are captured on two consecutive nights, it is recommended that traps be closed in that area for a night to allow the animals time to forage, especially during periods when females are under high lactational load such as when they are carrying large pouch-young or have nestlings or during periods of extreme weather conditions including very hot, very dry or very wet and cold conditions.

Small possums and gliders should be transferred directly from traps into cotton or calico bags for handling and transport. The very small species of possums can be held in the hand for data collections such as measurement and checking, but some of the petaurids bite fiercely and are best handled in a bag, which is probably also less distressing for the animals. For field studies, small possums can be released into thick cover at the site of capture, if this is available, or replaced in nest boxes. Alternatively, consider releasing animals at the point of capture at dusk. It is important to consider diurnal predators when releasing small possums during daylight. If animals remain in the open they are susceptible, especially to predatory birds such as ravens, currawongs and kookaburras. If animals are being retained during the day awaiting release, they should be held in their bags inside a suitable secure, quiet and dark area. In very hot weather, they should be checked regularly for signs of hyperthermia and, if necessary, transferred into appropriate well-aerated holding boxes to prevent them from overheating. If it is cold, do not leave animals in wet or damp bags.

Depending on the characteristics of these species, such as size and degree of ear pigmentation, small possums can be marked with numbers in their ears, using a small tattoo number punch, after swabbing the ear with 70% ethanol. Tattoos are not easily seen on pigmented ears and so, in these cases, microchips can be more suitable. Microchips (for example, passive integrated transponder tags; 'PIT' tags) are also suitable for animals over 10 g and should be inserted under the skin between the shoulder blades. However, some migration of PIT tags has been recorded in the smaller species, and as this may result in complications, it is best to mark the smaller species using ear-notching (for further information, see separate sections for 'Petaurid possums: gliders, Leadbeater's possum and the striped possum', 'Pygmy-possums', 'Feathertail glider' and 'Honey possum'). Ear tags are not recommended for small possums, as these invariably tear out, and this often results in large tears developing in the ears later on, which may compromise the possum's ability to detect predators.

For relatively short distances, animals can be transported in individual cloth bags, firmly tied with string, and placed so that the animals are not cramped or at risk of being smothered. In warmer weather, it is very important to transport animals in air-conditioned vehicles to prevent them from becoming overheated. Check to determine if animals are becoming overheated by looking for signs such as excessive panting and obvious distress. If the animals are overheated, they must be cooled immediately, with cold water over the wrists and sides of face, and transferred into a suitable well-aerated holding box or cage trap. If being transported for long distances or many hours, animals should be placed in their bags inside a suitable well-aerated transport box, as they may chew their way out of bags. Seek expert advice on transporting small possums and gliders for substantial time periods.

Captive husbandry

Most of the possum species are not particularly difficult to maintain in captivity. Before bringing possums into captivity, seek expert advice from experienced researchers and zoos that hold captive colonies of the species being investigated. There is also extensive information available on captive management of various possum species, including details and quantities for diets, provided in Jackson (2003). Suitable nest boxes, cage furniture, including branches for climbing, and an appropriate diet are key elements of good husbandry and maintenance. The degree of sociality varies among the species. Some form social groups, while others are essentially solitary, and this should be considered when housing animals. Captive animals should always be provided with fresh water, *ad libitum*.

Routine sample/data collection

Standard morphometric data, including body weight, head length, hind foot length, sex, pouch condition, testis size (length, width and depth) and checking and measurement of pouch-young can generally be conducted with animals handled in a bag.

Tissue samples for DNA can be collected from near the margins of ears using a small ear punch designed for laboratory mice or a 1 mm biopsy punch, after swabbing the site with 70% ethanol. If possible, use a torch behind the ear to avoid small blood vessels.

Faecal pellets can often be collected fresh from the holding bag or beneath the trap in which the animal was caught.

Blood sampling very small animals is difficult and requires training and experience. Hence, if blood collection is essential for the project, expert advice must be sought. Blood collection from the orbital sinus is not acceptable because of the adverse impacts of this procedure on animal wellbeing. Any proposal to collect blood from the orbital sinus would require specific justification to the animal ethics committee.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Gaseous anaesthesia of captive small possums can be achieved using isoflurane in oxygen (4–5% for induction, 1–2% for maintenance), with an oxygen flow rate of 1 L/minute. Some information is provided in Johnson & Hemsley (2008). Seek expert advice for your target species if anaesthetics are required in the field for wild possums or gliders. If possums are anaesthetised or sedated in the field they must be held until they are fully awake and competent to climb. Wherever possible, they should be released at dusk. There have been no pharmacokinetic studies on any analgesics for these species, thus if pain relief is required you must seek expert advice (e.g. zoo or wildlife veterinarian).

Humane killing

Small possums and gliders can be humanely killed using an overdose of sodium pentobarbitone (150 mg/kg) injected IP. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Due to the large number of different species, it is advisable to seek expert veterinary advice on health and diseases of small possums, in particular from veterinarians in major zoos. Information is also provided in Johnson & Hemsley (2008) and Jackson (2003). All traps and bags should be washed and disinfected when being moved from one field site to another, to avoid inadvertent movement of disease agents or parasites between populations or species.

Specific breeding requirements

Small possums vary considerably in their breeding biology. Some are relatively easy to breed in captivity. Expert advice should be sought (e.g. from major zoos with captive colonies of the target species). Information on breeding requirements is also provided in Jackson (2003).

Honey possums

Honey possums are a special case as these are one of the few truly nectarivorous mammals in the world with a very restricted distribution (Wooller et al. 1984; Renfree 1995). See separate section on 'Honey possums' for details.

Institutions and individuals with specialised expertise

Institutions

Lone Pine Koala Sanctuary Jesmond Road, Fig Tree Pocket Brisbane, Queensland Phone: (07) 3378 1366 Email: jacqui@koala.net Internet: http://www.koala.net	Perth Zoo Perth, Western Australia Phone: (08) 9474 0444 Email: email@perthzoo.wa.gov.au Internet: http://www.perthzoo.com.au
Taronga Zoo Sydney, New South Wales Phone: (02) 9969 2777 Email: tz@zoo.nsw.gov.au Internet: http://taronga.org.au/taronga-zoo	Zoos Victoria Veterinary Departments: Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville Werribee Open Range Zoo Phone: (03) 9731 9600 Email: worz@zoo.org.au Internet: http://www.zoo.org.au/werribee

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Bibliography for small possums and gliders

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Goldingay, RL & Jackson, SM (eds) 2004, *The biology of possums and gliders*, Surrey Beatty and Sons, Sydney.

Jackson, S 2003, 'Possums and gliders', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Johnson, R & Hemsley, S 2008, 'Gliders and possums' in L Vogelnest & R Woods, R (eds) *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 395–437.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Lindenmayer, D 2002, *Gliders of Australia: a natural history*, UNSW Press, Sydney.

Renfree, M. B. 1995 Honey possum *Tarsipes rostratus*. In: "The Mammals of Australia". Strahan, R. (Ed.) New rev. Ed. Reed Books Australia, Chatswood, pp. 257-260.

Van Dyck, S & Strahan R (eds) 2008, *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Wooller, RD, Russell, EM & Renfree, MB 1984, 'Honey possums and their food plants', in AP Smith & ID Hume (eds), *Possums and gliders*, Australian Mammal Society, Sydney, pp. 439–43.

Petaurid possums: gliders, Leadbeater's possum and the striped possum

This section provides more specific detail for the petaurid possums. Read in conjunction with the previous section, 'Smaller possums and gliders'.

Capture, handling, marking for identification, transport

The petaurids are highly arboreal. They can be caught in wire-mesh cage traps (approximately 20 cm × 17 cm × 50 cm) baited with a mix of peanut butter, rolled oats and golden syrup. These are set in trees and a dilute mix of golden syrup and water can be sprayed onto the trunk to attract the animals. To provide protection from adverse weather and to provide some visual protection from predators, approximately one-half or more of the roof and sides of cage traps should be covered, for example with heavy plastic or light aluminium sheet. Petaurids are nocturnal, and therefore traps should be set around dusk and cleared commencing at dawn. In warmer weather, traps should be positioned to ensure they will not be exposed to the hot sun. The number of traps set should be limited to ensure that they can be cleared before animals become overheated as the daytime environmental temperature increases.

Petaurids can also be caught using nest boxes attached to trees. Animals can simply be retrieved from these during the day. For striped possums, hand capture (by grasping the base of the tail close to the rump) and tranquilliser darting are the only techniques available to date. Darting requires a **very** high level of expertise as this species weighs 500 g or less, thus darting should not be considered without expert training and considerable experience. Because most petaurids will bite and scratch when handled, gloves should be worn when transferring them into a holding or handling bag. For collection of data and samples, petaurids are best handled while in a bag.

Petaurids can be marked using microchips (for example, passive integrated transponder tags; 'PIT' tags) injected subcutaneously between the scapulae. Because these species have darkly pigmented ears, tattoos on ears are difficult to discern.

Captive husbandry

Petaurid possums generally habituate well to captivity and are relatively easy to house and maintain in good health. Suitable nest boxes, branches for climbing and an appropriate diet are key elements of good husbandry and maintenance.

Petaurids can be housed alone, however most are social, living in family groups and often sharing dens, and can be housed as male–female pairs. It is essential to provide at least two nest boxes per pair. Limited information on striped possums suggests that they are the least social of all petaurids and do not commonly share dens, and thus it is advisable to house them separately. For research purposes, wire-mesh cages are suitable with dimensions of approximately 2 metres × 2 metres × 2 metres. If cages are outside, a solid roof, and at least two solid walls are required in cooler climates. Cages should be protected from direct sun in hot weather, as animals can become heat stressed. During long periods of extremely hot weather, it is essential to monitor animals several times each day for heat stress and to use water misters over the cage to help keep animals cool.

Extensive information on the captive husbandry requirements, including diet, for the petaurid possums is provided in Jackson (2003) or consult researchers or experts who have held these species in captivity for research purposes.

Diets for captive petaurid possums: gliders, Leadbeater's possum and the striped possum

Captive diets for petaurids should include nectar mix, pollen, fruit, invertebrates and high quality kibble. Seek expert advice from major zoos with captive colonies of the target species. Wherever possible, also provide flowers of native plants such as banksias and eucalypts. Striped possums are much more insectivorous than other petaurids and should be fed crickets, mealworms, insectivorous bird mix and fruit, with nectar mix provided occasionally.

Routine sample/data collection

For information, see 'Routine sample/data collection' in the 'Smaller possums and gliders' section.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. For information about gaseous anaesthesia, see 'Anaesthesia, analgesia and sedation' in the 'Smaller possums and gliders' section. While Zoletil® (tiletamine/zolazepam at 5 mg/kg, IM) has been used successfully for striped possums, this sedative has been implicated in the mortality of squirrel gliders and so should not be used without consultation with a zoo veterinarian with relevant experience with petaurids.

There have been no pharmacokinetic studies on any analgesics for these species so if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

For information, see 'Humane killing' in the 'Smaller possums and gliders' section.

Health issues, disease control and zoonoses

For information, see 'Health issues, disease control and zoonoses' in the 'Smaller possums and gliders' section, as well as Johnson & Hemsley (2008).

Specific breeding requirements

Sugar gliders, squirrel gliders and Leadbeater's possum generally breed quite well in captivity, if housed as compatible male–female pairs, but consult with major zoos that have experience with your species. Requirements for breeding striped possums in captivity remain unknown.

Bibliography for petaurid possums: gliders, Leadbeater's possum and the striped possum

Jackson, S 2003, 'Possums and gliders' in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Pygmy-possums

This section provides more specific detail for the pygmy-possums. Read in conjunction with the earlier section, 'Smaller possums and gliders'.

Capture, handling, marking for identification, transport

Capture techniques for pygmy-possums vary for different species and also across different habitat types. Expert advice should be sought from an experienced field biologist. For example, in *Banksia* habitats, pygmy-possums can be captured in small Elliott traps (33 cm × 10 cm × 10cm), baited with a peanut butter, rolled oats and golden syrup mix, and set on the branches or ground. Trapping is also used for the endangered mountain pygmy-possum in the alpine area, with the highest success achieved in late summer.

In colder conditions or climates, traps should be covered with a plastic bag to provide protection from rain, and furnished with a generous handful of fufu filling, to keep the animals warm. Pygmy-possums can also be caught using pitfall traps, which is one of the most successful methods for western pygmy-possums. Pitfall traps contain some sand or soil, leaf litter and small sticks or bark for shelter, with holes drilled in the base so that traps will drain if it rains. In extreme weather (and for pitfall trapping, if heavy rain is forecast), trap lines should be closed. Nest boxes have also been used to capture eastern pygmy-possums.

Most pygmy-possums are nocturnal, therefore traps should be set around dusk and cleared commencing at dawn. In warmer weather, traps should be positioned to ensure they will not be exposed to the hot sun, and the number of traps set should be limited to ensure that they can be cleared before animals become overheated as the daytime environmental temperature increases. If individuals are captured on two consecutive nights, it is recommended that traps be closed for a night to allow the animals time to forage, especially during periods when females are under high lactational load such as when they are carrying large pouch-young or during periods of extreme weather conditions such as very hot, very dry or very wet and cold conditions.

Be aware that pygmy possums may enter torpor in traps in cooler weather. They can be rewarmed gently in the hand or by placing them in a soft cotton bag and then placing this against your skin under clothing, but avoid over handling. The biology of long-tailed pygmy-possums is very poorly known. They do not appear to enter traps, but have very occasionally been caught by hand when encountered occupying low vegetation.

Because of their small size, pygmy-possums are easily handled. Animals should be transferred directly from traps into soft cotton bags for handling and or transport. For field studies, small possums can be released into thick cover at the site of capture. It is important to consider diurnal predators when releasing small possums during daylight. If animals remain in the open, they are susceptible to predators such as ravens, currawongs and kookaburras. In the field, if animals are being retained during the day awaiting release, they should be held in their bags inside a suitable secure quiet and dark area. In the event of very hot weather, they should be checked regularly for signs of hyperthermia and, if necessary, transferred into appropriate well-aerated holding boxes to prevent them from overheating. If it is cold, do not leave animals in wet or damp bags.

Pygmy-possums can be marked using microchips (for example, passive integrated transponder tags; 'PIT' tags) injected subcutaneously between the scapulae. However, because of their small size and the risk of tag migration, which may have negative impacts on the animals, ear notching is recommended as an alternative. If tissue for DNA analysis is being collected (see 'Routine sample/data collection' in the 'Smaller possums and gliders' section) the position on the ear from which the sample is taken can also be used for identification.

Captive husbandry

Pygmy-possums generally habituate well to captivity and are relatively easy to house and maintain in good health. They can be housed in relatively small wooden cages or glass aquaria with at least one wall of wire mesh. A mesh size of 0.5 cm is recommended to prevent escape by very small individuals. Pygmy-possums are generally regarded as solitary in the wild, but they are relatively tolerant in captivity and can be housed in groups in larger cages. Suitable nest boxes, branches for climbing and an appropriate diet are key elements of good husbandry and maintenance.

Extensive information on the captive husbandry requirements, including diet, for the pygmy-possums is provided in Jackson (2003) or consult researchers or experts who have held these species in captivity for research purposes.

Diets for captive pygmy-possums

Captive diets for pygmy-possums should include nectar mix, pollen, fruit and invertebrates. Seek expert advice (e.g. major zoos, researchers with experience with the species under investigation). Wherever possible, also provide flowers of native plants such as banksias and eucalypts. Mountain pygmy-possums should be fed fine seed mix, mealworms, fruit, vegetables and sunflower seed, supplemented with high quality kibble, almonds, crickets and moths.

Be aware that pygmy-possums often enter torpor and, as a result, they may gain weight quite rapidly and become obese in captivity. To deal with this problem, pygmy-possums should be monitored for weight gain regularly and their diet restricted if necessary.

Routine sample/data collection

For information, see 'Routine sample/data collection' in the 'Smaller possums and gliders' section.

Anaesthesia, analgesia and sedation

For information, see 'Anaesthesia, analgesia and sedation' in the 'Smaller possums and gliders' section.

Humane killing

For information, see 'Humane killing' in the 'Smaller possums and gliders' section.

Health issues, disease control and zoonoses

For information, see 'Health issues, disease control and zoonoses' in the 'Smaller possums and gliders' section, as well as Johnson & Hemsley (2008).

Specific breeding requirements

There are few records of successful breeding of *Cercartetus* pygmy-possums in captivity. Seek expert advice from major zoos with breeding colonies and expertise in the target species. For example, Healesville Sanctuary has a breeding program for mountain pygmy-possums. Information is also provided in Jackson (2003).

Bibliography for pygmy-possums

Jackson, S 2003, 'Possums and gliders', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Feathertail glider

This section provides more specific detail for the feathertail glider. Read in conjunction with the earlier section, 'Smaller possums and gliders'.

Capture, handling, marking for identification, transport

Feathertail gliders are very small, nocturnal and highly arboreal. They can be caught using suitably designed nest boxes attached to trees and animals can simply be retrieved from these during the day. Because of their small size, feathertail gliders are easily handled. Animals should be transferred directly from nest boxes into soft cotton bags for handling or transport. For field studies, they should be replaced in the nest box after procedures such as handling and data collection.

It is always important to consider diurnal predators when releasing feathertail gliders during daylight. If animals remain in the open they are susceptible, especially to predatory birds such as ravens, currawongs and kookaburras. In the field, if animals are being retained during the day pending release, they should be held in their bags inside a suitable secure quiet dark area. In very hot weather, they should be checked regularly for signs of hyperthermia and, if necessary, transferred into appropriate well-aerated holding boxes to prevent them from overheating. If it is cold, do not leave animals in wet or damp bags.

Because of their small size, feathertail gliders should not be marked using microchips (for example, passive integrated transponder tags; 'PIT' tags). It is recommended that small ear notches, on the edges of the ears, be used instead. The tissue from these notches can be collected and stored for DNA analysis.

Captive husbandry

Feathertail gliders live and nest in groups. They can be housed indoors in relatively small wooden, glass or stainless steel cages with at least one wall of wire mesh. A mesh size of 0.5 cm is recommended to prevent escape by very small individuals. Cages should contain a network of small branches, surplus nest boxes and a substrate of sand and leaf litter.

Feathertails are agile and good at escaping. Hence, it is useful to lock them into their nest box immediately after opening the cage for procedures such as cleaning and feeding.

Diets for captive feathertail gliders

Captive diets for feathertails should include nectar mix, pollen, fruit and invertebrates such as moths, crickets and mealworms. Wherever possible, also provide flowers of native plants such as eucalypts, melaleucas and banksias.

Be aware that feathertails often enter torpor, and as a result they may gain weight quite rapidly and become obese in captivity. To deal with this problem, the animals should be monitored for weight gain regularly and their diet restricted if necessary.

Expert advice on dietary and health management for captive feathertail gliders should be sought from researchers or zoos with experience with the species under investigation. Information is also provided in Jackson (2003).

Routine sample/data collection

For information, see 'Routine sample/data collection' in the 'Smaller possums and gliders' section.

Anaesthesia, analgesia and sedation

For information, see 'Anaesthesia, analgesia and sedation' in the 'Smaller possums and gliders' section.

Humane killing

For information, see 'Humane killing' in the 'Smaller possums and gliders' section.

Health issues, disease control and zoonoses

For information, see 'Health issues, disease control and zoonoses' in the 'Smaller possums and gliders' section above, as well as Johnson & Hemsley (2008).

Specific breeding requirements

In south-eastern Australia, there is a breeding season with births occurring between June and January. Females typically produce two litters each year, with the second litter going through a period of embryonic diapause. Successful breeding of feathertail gliders is limited, but they have been bred at Taronga Zoo when housed in larger enclosures with multiple animals, rather than as male–female pairs. Expert advice on breeding requirements should be sought from Taronga Zoo. Information is also provided in Jackson (2003).

Bibliography for feathertail glider

Jackson, S 2003, 'Possums and gliders', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 205–44.

Honey possum

This section provides more specific detail for the honey possums. Read in conjunction with the earlier section, 'Smaller possums and gliders'.

Capture, handling, marking for identification, transport

Honey possums need to be handled with great care due to their stress-prone nature. Honey possums possess adrenal glands that are approximately 100 times larger per kilogram of body mass than those of other marsupials. Rough handling of honey possums can result in cardiac arrest within a matter of seconds, and they are particularly sensitive to high frequency noise.

Honey possums can be caught using pitfall traps made from polyvinylchloride (PVC) piping approximately 20 cm in diameter and 40–50 cm deep. These should be sunk into the soil flush with the surface, with leaf litter and small sticks or bark added for shelter. No drainage is required due to the porous nature of the sandy soils in habitats occupied by these animals, however if very heavy rain is forecast, trap lines should be closed. Honey possums experience no distress when confined on rainy nights in pitfall traps with sandy bottoms (Bradshaw et al. 2007). They are highly mobile, with recent radio tracking showing that 9 g males move up to 0.8 km in a night, and this should be considered when installing traplines (Bradshaw & Bradshaw 2002).

The honey possum is nocturnal, except in cooler weather. Traps should therefore be opened at dusk, checked at dawn, and then closed during the day to prevent bi-catch. Be aware that if food is scarce, or the weather is cool, honey possums often become torpid. If individuals are captured on two consecutive nights it is recommended that traps be closed for a night to allow the animals time to forage, especially during periods when females are under high lactational load such as when they are carrying large pouch-young or during periods of extreme weather conditions such as prolonged drought.

Because they are small, honey possums are easily handled. Animals should be transferred directly from traps into soft cotton or calico bags for handling and or transport. For field studies, they can be released into thick cover at the site of capture. Always consider diurnal predators when releasing small possums during daylight. If animals remain in the open they are susceptible, especially to predatory birds such as ravens, currawongs and kookaburras. In the field, if animals are being retained during the day pending release, they should be held in their bags inside a suitable secure, quiet and dark area. In the event of very hot weather, they should be checked regularly for signs of hyperthermia and if necessary transferred into appropriate well-aerated holding boxes to prevent them from overheating. If it is cold, do not leave animals in wet or damp bags.

Because of their very small size (mostly less than 10 g), honey possums should not be marked using microchips (for example, passive integrated transponder tags; 'PIT' tags). It is recommended that small ear notches, on the edges of the ears, be used instead. The tissue from the notches can be collected and stored for DNA analysis.

Captive husbandry

Honey possums can be housed indoors, in medium-to-large wooden or stainless-steel cages with at least one wall of wire mesh. A mesh size of 0.5 cm is recommended to prevent escape by very small individuals. Not all individuals adapt to confinement in small cages and death due to inanition is common. Rather, the need for large-sized cages with the provision of adequate nutrition and access to fresh blossoms should be emphasised, as confinement in small cages with controlled light and temperature regimes leads inevitably to a cessation of oestrous cycling in females (Oates et al. 2007). Cages should contain a network of small branches, surplus nest boxes and a substrate of sand and leaf litter.

Although the social system of honey possums in the wild is not known, in captivity females are dominant over males (Russell & Renfree 1989). However, animals huddle together to conserve body heat, and multiple animals can be housed successfully together in larger cages.

Diets for captive honey possums

Honey possums are highly-specialised feeders and the diet in captivity consists of nectar or honey and pollen (Weins, Renfree & Wooller 1979). Measurements in free-ranging honey possums have shown that a 9 g adult consumes, on average, 7 mL of nectar and 1 g of pollen per day (Bradshaw & Bradshaw 1999) and that this provides the animals with approximately 10 times their daily minimal nitrogen requirement (MNR) to maintain nitrogen balance (Bradshaw & Bradshaw 2001). The maintenance of a long-term colony over a number of years, with successful breeding and recruitment of young, was achieved after careful control of the composition and volume of diet offered to both males and females (Bradshaw, Everett & Bradshaw 2000). Supplying 'high levels of nitrogen' above a daily intake of 30 mg per day was found to be associated with gross kidney disease and renal failure (Bradshaw & Bradshaw 2012).

Be aware that honey possums may enter torpor and, as a result, may gain weight quite rapidly and become obese in captivity. To deal with this problem, the animals should be monitored for weight gain regularly and their diet restricted if necessary.

Expert advice should be sought on diets for captive honey possums (e.g. major zoos, researchers with experience with the species under investigation).

Routine sample/data collection

For information, see 'Routine sample/data collection' in the 'Smaller possums and gliders' section. To date, the only successful method for blood collection that has been used with large numbers of honey possums is collection from the orbital sinus. However, this technique is no longer acceptable because of the adverse impacts of this procedure on animal wellbeing. Any proposal to collect blood from the orbital sinus would require specific justification to the animal ethics committee.

Anaesthesia, analgesia and sedation

For information, see 'Anaesthesia, analgesia and sedation' in the 'Smaller possums and gliders' section.

Humane killing

For information, see 'Humane killing' in the 'Smaller possums and gliders' section.

Health issues, disease control and zoonoses

For information, see 'Health issues, disease control and zoonoses' in the 'Smaller possums and gliders' section above, as well as Johnson & Hemsley (2008).

Specific breeding requirements

In the wild, honey possums have no obvious breeding season, and females with pouch-young can be found throughout the year, but predominantly in early autumn, winter and spring when pollen and nectar are most abundant (Renfree, Russell and Wooller 1984). Females may breed in their first year and, in the wild, some females breed at least twice a year. Honey possums have only been bred successfully in captivity in large outdoor enclosures containing multiple animals, vegetation such as grasses and shrubs and a diet manipulated to supply high levels of nitrogen.

Bibliography for honey possum

- Bradshaw, FJ & Bradshaw, SD 2001, 'Maintenance nitrogen requirement of an obligate nectarivore, the Honey possum, *Tarsipes rostratus*', *Journal of Comparative Physiology B*, vol. 171, pp. 59–67.
- Bradshaw, SD & Bradshaw, FJ 1999, 'Field energetics and the estimation of pollen and nectar intake in the marsupial honey possum, *Tarsipes rostratus*, in heathland habitats of south-western Australia', *Journal of Comparative Physiology B*, vol. 169, pp. 569–80.
- Bradshaw, SD & Bradshaw, FJ 2002, 'Short-term movements and habitat utilisation of the marsupial Honey possum, *Tarsipes rostratus*', *Journal of Zoology (Lond)*, vol. 258, pp. 343–48.
- Bradshaw, SD & Bradshaw, FJ 2012, 'The physiology of the honey possum, *Tarsipes rostratus*, a small marsupial with a suite of highly specialised characters: a review', *Journal of Comparative Physiology B*, vol. 182, pp. 469–89.
- Bradshaw, FJ, Everett, L & Bradshaw, SD 2000, 'On the rearing of Honey possums', *Western Australian Naturalist*, vol. 22, pp. 281–88.
- Bradshaw, SD, Phillips, R, Tomlinson, S, Holley, R, Jennings, S & Bradshaw FJ 2007, 'Ecology of the Honey possum, *Tarsipes rostratus*, in Scott National Park, Western Australia', *Australian Mammalogy*, vol. 29, pp. 25–38.
- Oates, J, Bradshaw, FJ, Bradshaw, SD, Stead-Richardson, EJ & Philippe D 2007, 'Reproduction and embryonic diapause in a marsupial: Insights from captive female Honey possums, *Tarsipes rostratus*, (Tarsipedidae)', *General and Comparative Endocrinology*, vol. 150, pp. 445–61.
- Renfree, MB, Russell, EM & Wooller, RD 1984, 'Reproduction and life history of the honey possum, *Tarsipes rostratus*', in AP Smith & ID Hume (eds), *Possums and gliders*, Australian Mammal Society, Sydney, pp. 427–37.
- Renfree, M. B. 1995 Honey possum *Tarsipes rostratus*. In: "The Mammals of Australia". Strahan, R. (Ed.) New rev. Ed. Reed Books Australia, Chatswood, pp. 257-260.
- Renfree, MB 2008, 'Honey possum *Tarsipes rostratus*', in S Van Dyck & R Strahan (eds), *The mammals of Australia*, Reed New Holland, Sydney, pp. 258–60.
- Russell, EM & Renfree MB 1989, 'Tarsipedidae', in DW Walton & BJ Richardson (eds), *Fauna of Australia: Mammalia*, Australian Government Publishing Service, Canberra, pp. 769–82.
- Weins, D, Renfree, MB & Wooller, RD 1979, 'Pollen loads of honey possums (*Tarsipes spenserae*) and nonflying mammal pollination of southwestern Australia', *Annals of the Missouri Botanical Garden*, vol. 66, pp. 830–39.
- Wooller, RD, Russell, EM & Renfree, MB 1984, 'Honey possums and their food plants', in AP Smith & ID Hume (eds), *Possums and gliders*, Australian Mammal Society, Sydney, pp. 439–43.
- Wooller, R & Wooller, S 2013, *Sugar and sand: The world of the honey possum*, Swanbrae Press.

Subclass Theria: Eutherians

Bats

The order Chiroptera contains two suborders: Megachiroptera that consists of flying foxes, fruit bats and relatives, and Microchiroptera that consists of the microbats. Megachiropterans weigh up to one kilogram and have wing spans of up to 1.6 metres. They feed on fruit and nectar. Microchiropterans are a diverse group of bats (many are insectivores) that weigh up to 170 g and have wingspans up to 30 cm, but there are also species as small as 2.6 g in weight such as the northern pipistrelle.

Megabats

Megabats (Megachiropterans) such as the flying foxes and fruit bats tend to be distributed more in coastal regions of northern and eastern Australia and offshore islands. Many species are highly mobile, migrating long distances. Information on the general biology and distribution of individual species is provided in Van Dyck & Strahan (2008).

Megabats – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Grey-headed flying fox, <i>Pteropus Poliocephalus</i> .	Vulnerable.	East coast of Australia from Western Victoria to Townsville in Queensland.	A wide variety of fruits and flowers.
Black flying fox, <i>Pteropus alecto</i> .	Not listed.	Throughout coastal northern Australia from near Carnarvon to Grafton.	Blossoms of flowering trees.
Spectacled flying fox, <i>Pteropus conspicillatus</i> .	Vulnerable.	Restricted to northeast Queensland rainforests.	Primarily frugivorous.
Little red flying fox, <i>Pteropus scapulatus</i> .	Not listed.	Widely distributed especially in northern Australia.	Predominantly nectar and pollen from trees.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Flying foxes are usually captured using mist nets set on very high poles of 13 metres. Catching is conducted in the early morning when bats are returning to their roost sites and nets must be constantly monitored. The animals can also be trapped using very large harp nets suspended on yacht masts with a catch bag at the base that the animals slide into and with the poles set in concrete. These traps do not need to be constantly monitored but they are not manoeuvrable because of their size. Animals must be removed from traps carefully as they generally wriggle, flap their wings and legs wildly and try to bite. One hand must be used to hold the wings against the body and the other hand must securely hold the head by grasping around the neck. Alternatively, if animals are being removed from tall mist nets, one person wearing gauntlets can hold the bat while the other extracts it from the net.

For inspection, sample collection or marking, the animals are wrapped firmly in a folded towel and held through the towel as a second handler carries out the necessary procedures. Allowing the animal to grasp a fold of towel or the handler's clothing with their feet will help to keep it calm.

As Australian bats can carry Australian Bat Lyssavirus (ABL), handlers must be vaccinated against rabies and must wear gloves when handling bats.

If females are caught from October to December, teats, which are located in the armpits, should be inspected. If lactating, females must be released immediately because they are likely to have a young back at the colony.

Animals are transported in carry cages that can contain up to two adults or, if travelling only a short distance, the animals can be placed in a pillowcase that is hung from a pole. Cages need to contain dowelling or a natural branch and be tall enough to accommodate a hanging bat. Provided the cages are covered, most animals remain calm. Bats should be transported in air-conditioned vehicles, as they are prone to heat stress.

All bat banding in Australia is regulated by the Australian Bird and Bat Banding Scheme so banding can only be carried out by experienced, licensed operators. Other methods of individual identification include microchips (for example, passive integrated transponder tags; 'PIT' tags) inserted under the skin between the scapulae. These generally require the animal to be caught for identification but tag readers can be set up at roosting sites. Because PIT tags may migrate over time, tag readers should be scanned all over an animal's body. The tags are sometimes lost just after implantation by being pushed out through the injection site. This is avoided by sealing the entry wound with tissue glue. Animals may also be ear notched for individual identification. Nail polish can be used if bats are caught regularly and the polish reapplied.

Captive husbandry

Environmental enrichment can be provided in the form of small toys, such as mango seeds, hessian or polar fleece baffles, or strong ropes to allow swinging and climbing. Additional information on captive husbandry is provided in Jackson (2003).

Animals should be housed in large cages at least 3 metres high × 3 metres wide × 18 metres long. The cages should not be much taller than 3 metres otherwise retrieving bats hanging on the roof will be problematic. Up to 50 bats can be kept in an enclosure of this size. Bats should not be housed alone as they are social animals. They will readily accept any newcomers. If it is necessary to house them singly or in small groups, a mirror can be used to give the impression of larger numbers.

Part of the cage should be covered to provide protection from the elements. The floor of the cages should be concrete or similar. Some handlers and researchers recommend placing sawdust on the floor but flying foxes are very messy thus it is generally easier if they are housed above a concrete floor that can be easily hosed. Because flying foxes tend to defecate as they feed, it is best to set up a feeding area so that their waste is concentrated in one site.

Diets for captive megabats

Captive flying foxes should be fed a mixture of chopped fruits that do not include any citrus. Vegetables should not be given, other than cooked corn and leaf vegetables such as bok choy or celery. A protein supplement such as Wombaroo High Protein Supplement should also be fed at a rate of 10 g/bat per day. Animals eat a very large amount of food, approximately one-third to one-half of their body weight per night.

It is best to provide food and water in buckets suspended at various heights to encourage movement and minimise fouling. Whole fruit can also be hung on skewers throughout the enclosure. Native fruits, flowers and foliage can be offered when available. Artificial nectar (Wombaroo) is offered in rat drinkers. Approximately half a teaspoon of salt should be added per 800 ml of fresh water. Fresh, unsalted water should also be available.

Routine sample/data collection

While the animal is under anaesthetic, tissue for DNA analysis can be collected from the wing with a 3 mm diameter biopsy punch, or the tissue derived from marking by ear notching can be used.

Blood samples can be collected from a wing vein.

If age determination is essential for the project, the pre-molar, which is non-functional and shallow-rooted, may be extracted. This must be done under anaesthesia. Lignocaine should be applied to the gum once the tooth is removed. The animal's age can then be subsequently determined by characteristics of the sectioned tooth.

Faecal material can often be collected from bags during handling.

Morphometric measurements include forearm length, ear length, thumb length, testes measurements and body weight.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. The method of choice is gaseous anaesthesia using 5% isoflurane delivered in oxygen at a flow rate of 1 L/minute. Maintenance generally requires 2% isoflurane. Animals can also be anaesthetised with 5 mg/kg Alfaxan® injected into the main wing vein. If intravenous injection is not possible, 2 mg/kg xylazine combined with 10 mg/kg ketamine or 50 µg/kg medetomidine combined with 5 mg/kg ketamine can be given intramuscularly.

Analgesics that have been used in flying foxes include butorphanol (0.4 mg/kg), buprenorphine (0.03 mg/kg) and meloxicam (0.2 mg/kg).

Humane killing

If the animal is already anaesthetised, it is acceptable to administer an overdose of sodium pentobarbitone (150 mg/kg), injected IV, IC or IP. Because of the risk of infection with Australian Bat Lyssavirus and other zoonoses all bats should be anaesthetised prior to euthanasia. It is then acceptable to administer an overdose of sodium pentobarbitone (150 mg/kg), injected IV, IC or IP.

Health issues, disease control and zoonoses

Information on health and disease issues for flying foxes is provided in Olsson and Woods (2008). Captive flying foxes tend to have few health issues as long as they are kept clean and well fed. They can suffer from fungal skin conditions if they are housed inappropriately in dark, poorly-ventilated cages without access to direct sunlight. Wing and foot trauma can also occur if inappropriate cage materials are used.

Surveys of wild flying foxes have found up to 40% with antibodies to Hendra virus. This virus causes no clinical signs in bats but will cause fatal disease in horses. People exposed to infected horses have also developed fatal disease. To date there have been no recorded cases of people being infected directly from flying foxes. The virus is believed to be transmitted in flying fox urine and birthing fluids.

Flying foxes can also carry ABL, which is closely related to rabies. Handlers must therefore be vaccinated against rabies and must wear gloves when handling flying foxes. The prevalence of ABL is less than 1% in healthy wild bats but increases up to 20% in sick and injured bats. Affected bats may show a variety of symptoms including aggression, abnormal vocalisation, paralysis, inability to fly or sitting on the ground and not attempting to escape handling. Because the virus is found in the saliva, infection generally occurs through a bite or scratch. Anyone bitten by a bat should wash the wound with soap and water and seek medical advice immediately (Animal Health Australia 2009).

All traps and bags should be washed and disinfected when being moved from one field site to another to avoid inadvertent movement of disease agents between populations or species.

Flying foxes do not carry fleas, lice or any other obvious ectoparasites that can be passed to humans. They may become infested with mites and nycteribiid flies. Nycteribiid flies are small wingless flies that feed off blood in the same way as fleas, but are non-pathogenic. Young flying foxes may become infested with roundworms, which are treated with a cat wormer.

If injured or dead adult animals are found between September and December, they are likely to be carrying young that are dependent on their mothers for the first three weeks of life. If the mother dies then the young will be unable to survive. The teats are in the armpit and babies may not be seen without a thorough inspection of the bat. Juveniles can be given to a wildlife carer to hand raise.

Institutions and individuals with specialised expertise

Institutions

<p>Australasian Bat Society Email: president@ausbats.org.au Internet: http://ausbats.org.au</p>	<p>Lone Pine Koala Sanctuary Jesmond Road, Fig Tree Pocket Brisbane, Queensland Phone: (07) 3378 1366 Email: jacqui@koala.net Internet: http://www.koala.net</p>
<p>Perth Zoo Perth, Western Australia Phone: (08) 9474 0444 Email: email@perthzoo.wa.gov.au Internet: http://www.perthzoo.com.au</p>	<p>Taronga Zoo Sydney, New South Wales Phone: (02) 9969 2777 Email: tz@zoo.nsw.gov.au Internet: http://taronga.org.au/taronga-zoo</p>
<p>Zoos Victoria Veterinary Departments:</p> <p>Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au</p> <p>Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville</p>	

Individuals

<p>Dr Deborah Middleton (flying fox maintenance at PC3 and PC4 bio-containment) Veterinary Pathologist CSIRO Australian Animal Health Laboratory Geelong, Victoria Email: deborah.middleton@csiro.au</p>	<p>Dr Rodney van der Ree (flying foxes) Australian Research Centre for Urban Ecology Royal Botanic Gardens Melbourne Email: rvdr@unimelb.edu.au</p>
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Bibliography for megabats

- Animal Health Australia 2009, 'Disease strategy: Australian bat lyssavirus, Version 3.0'. *Australian Veterinary Emergency Plan (AUSVETPLAN)*, Edition 3, Primary Industries Ministerial Council, Canberra, ACT.
- Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.
- Churchill, S 2009, *Australian bats*, 2nd edn, Allen and Unwin, Sydney.
- Griffith, M 2004, *Husbandry manual for grey-headed flying fox*, [http://nswfmpa.org/Husbandry%20Manuals/Published%20Manuals/Mammalia/Grey%20Headed%20Flying%20Fox%20\(Griffith\).pdf](http://nswfmpa.org/Husbandry%20Manuals/Published%20Manuals/Mammalia/Grey%20Headed%20Flying%20Fox%20(Griffith).pdf)
- Hall, L & Richards, G 2000, *Flying foxes: fruit and blossom bats of Australia*, UNSW Press, Sydney.
- Jackson, S 2003, 'Bats', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 297–350.
- Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.
- Olsson, A & Woods, R 2008, 'Bats', in L Vogelneust & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 465–502.
- Van Dyck, S & Strahan, R (eds) 2008, *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Microbats

Microbats are a diverse and widespread group in Australia. Their small size and nocturnal aerial lifestyle mean that special expertise is required to conduct research on this group, especially in the field. Much of our knowledge about the care of captive microbats comes from wildlife shelters. Information on the various species and their general biology is provided in Van Dyck & Strahan (2008).

Microbats – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Eastern horseshoe bat, <i>Rhinolophus megaphyllus</i> .	Least concern.	East coast of Australia from East Gippsland in Vic. to Cape York Peninsula, Qld.	A variety of mainly flying insects, although animals have been observed taking spiders from the ground.
Lesser long-eared bat, <i>Nyctophilus geoffroyi</i> .	Least concern.	Throughout the majority of mainland Australia and Tas., but excluding offshore islands and the Qld coast.	A variety of both flying and flightless insects.
Greater long-eared bat, <i>Nyctophilus timorensis</i> .	Insufficient data.	Southern WA, western and south-eastern SA (but not north-eastern or around Adelaide), western Vic., western NSW and southern Qld.	Insectivorous, but details are unknown.
Southern forest bat, <i>Vespadelus regulus</i> .	Least concern.	Southern corner of WA, the south coast of SA from the Eyre Peninsula to the Vic. border, most of Victoria, coast of NSW and a small area of south-eastern Qld.	A variety of flying insects.
Ghost bat, <i>Macroderma gigas</i> .	Vulnerable.	A small patch in the Pilbara, WA and along the northern Australian coast in the Kimberly, Arnhem Land, NT, and most of northern Qld.	Ghost bats are carnivorous and eat other bats, small mammals, small birds and frogs, as well as larger insects.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

The preferred method of capture is harp traps, which are set overnight and checked in the morning. Bats fly into the fishing line of the harp trap and slide down into a protected holding area at the base of the trap. Once caught in the trap animals tend to be calm and even go to sleep. There are reports of microbats being killed by ants while in the trap. If this is a possibility traps need to be checked more frequently or steps taken to deny ants access to the trap.

Bats can also be caught in mist nets set up in their flight paths. Mist nets must be constantly monitored and animals removed immediately to avoid injury. Bats may also be caught by placing a bag or net over the exit of a roost and catching them as they fly out at dusk. This method is more likely to cause injury to animals and should only be considered if harp trapping is not possible. In addition, some species can be caught directly from nest boxes set on trees.

Microbats are extremely delicate and must be removed from traps with the utmost care. It is best to hold them in a cupped hand with the wings in the folded position, applying only enough pressure to restrain the animal if it tries to escape. After removal from traps bats should be placed into small soft cotton or calico bags. Up to 10 individuals can be stored in a single bag (as microbats are colonial animals, they happily huddle together). As the bats easily become entangled and may be badly injured, it is important that the bag has no loose threads and bags are best if made with no seams on the inside. Tie the bag very tightly and ensure there are no gaps in the fabric as the bats will readily escape through very small openings. Bags should be hung in a secure place off the ground.

Gloves must be worn during handling because of the risk of infection with Australian Bat Lyssavirus (ABL). However, handlers need to be aware that wearing gloves reduces dexterity and sensitivity and may cause injury to animals. To minimise the risk of contracting ABL, all microbat researchers must be vaccinated against rabies.

Canvas bags containing bats can be transported in vehicles over long distance hung from something like a cargo net attached to the roof, or bags can be placed into boxes so that animals are not crushed during transport. If transporting animals in boxes, prevent overheating by minimising the number per box, and run the air conditioner in the vehicle during hot weather.

All bat banding in Australia is regulated by the Australian Bird and Bat Banding Scheme. Banding can only be carried out by experienced, licensed operators. Other methods of individual identification include microchips (for example, passive integrated transponder tags; 'PIT' tags) inserted beneath the skin between the scapulae, but this method can only be used on individuals that are anaesthetised and weigh more than 10 g. These generally require the animal to be caught for identification but readers can be set up at nest boxes or other roosting sites. Ear notching is not recommended as it may interfere with echolocation.

Captive husbandry

Injured animals should be kept on a heat pad in winter. Injured animals need to be examined by a veterinarian and should be kept on a heat pad, wrapped in a towel or cloth to prevent burns.

Unlike other microbats, horseshoe and leaf-nosed bats are prone to handling and capture stress, which can cause death in these two taxa.

Microbats can be housed in flexariums with towels, polar fleeces, cotton pouches or other materials hanging from the roof, to provide security and roosting sites. In the wild, different species usually do not roost together and should therefore be housed separately in captivity. In the wild, females tend to be communal and males solitary. However, in captivity, the sexes are often housed separately. For example, males and females of yellow-bellied sheath-tail bats should be housed separately, whereas other species may be more communal. It is essential to obtain expert advice on species-specific requirements.

Bats should be provided with flight enclosures to allow them to exercise. The enclosures should be constructed of flywire mesh, and should be at least 3.5 metres wide × 3.5 metres long × 2 metres high. Commercially available picnic mosquito shrouds are good for housing bats such as the lesser long-eared, the wattle or Gould's bats, but are not suitable for species with longer, narrower wings that are adapted for speed but not manoeuvrability. Such species may need enclosures approximately 10 metres long.

Diets for captive microbats

It is easiest to feed captive insectivorous species on mealworms, but as these are low in several essential minerals and vitamins, supplements must be included in the mealworm diet. It is best to grow the mealworms on Wombaroo Insectivore (Wombaroo Products) and small carnivore mix, with the addition of a multivitamin supplement such as Penta-vite®. Full details can be found in Jackson (2003).

Because wild microbats capture flying prey, they need to be taught how to feed in captivity. Initially they need to be hand fed by removing the head from a mealworm and smearing it around their mouth until they learn to take food from the handler's hand. After they have mastered this, they need to be trained to eat out of a dish by putting a mealworm into a bat's mouth and holding the bat over the dish until the bat learns to grab worms from the dish. Species of gleaning bats, such as lesser long-eared bats, learn how to feed in captivity readily, whereas species that are strict aerial hunters, such as freetail bats, need to be hand fed for their entire life in captivity.

Water should be provided in a shallow container, no more than 1 cm high.

The frequency of feeding required is seasonal, because bats exhibit seasonal behaviour patterns. In winter wild bats enter torpor. It is best to keep animals in an unheated room so that they are exposed to natural temperature patterns. When housed like this, animals should be fed every 4–5 days in winter and every 1–2 days in summer. Do not leave mealworms in with bats in torpor, unless they have been decapitated, as they can attack and eat the bat. In the wild, animals gain weight in autumn to help them cope with the low winter temperatures. In captivity, despite the reduced frequency of feeding during winter, animals still gain weight in autumn and shed weight in spring despite any increased feeding. Bats consume a large amount of food, and in the wild will eat one-half to three-quarters of their body weight in a single night. As an indicator, a bat of 10 g should eat about 10–20 mealworms per feeding and a 5 g bat should eat about 5–10 mealworms, but seek expert advice on individual species (e.g. major zoos with colonies).

Unlike other Australian microbats, Ghost bats are carnivorous and cannot be fed a diet consistently solely of mealworms. Their diet must be supplemented with meat such as mice or day old chicks. Advice should be sought from an expert with this species.

Routine sample/data collection

Tissue for DNA analysis can be collected from the wing with a 3 mm diameter biopsy punch while the animal is under anaesthetic.

Faecal material can often be collected from holding bags.

Morphometric measurements include forearm length, ear length, testes measurements and body weight.

Blood is generally collected from the vein running along the forearm in the wing in larger species and from a tail membrane vein in smaller species.

Swab samples, typically from the mouth, nose and urogenital opening, can be taken to determine the presence of pathogens. Droppings can also be collected for analysis of endo-parasites.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Microbats are usually anaesthetised by administering isoflurane (5% for induction; 2–2.5% for maintenance) and oxygen at 1 L/minute using a small face mask. Sedation is not needed for routine handling.

Analgesics that have been used in microbats include butorphanol (2 mg/kg), buprenorphine (0.05 mg/kg) and meloxicam (1–2 mg/kg).

Humane killing

Microbats can be humanely killed using cervical dislocation or an overdose of sodium pentobarbitone (150 mg/kg). If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethobarb®), prior anaesthesia or sedation is essential to prevent pain or irritation from the sodium pentobarbitone solution. Hypothermia is not to be used.

Health issues, disease control and zoonoses

Information on health and disease issues for microbats is provided in Olsson and Woods (2008). ABL antibodies have been reported from many microbat species; however the only species identified carrying the virus to date is the yellow-bellied sheath-tail bat. While only about 5% of sick and injured microchiroptera have tested antibody positive, up to 62.5% of yellow-bellied sheath-tail bats tested were positive. One human fatality has resulted following a bite from a yellow-bellied sheath-tail bat. The same precautions used for handling megachiroptera should also be adopted when handling microchiroptera.

All traps and bags should be washed and disinfected when being moved from one field site to another to avoid inadvertent movement of disease agents between populations or species.

It is rare to see sick or injured microbats in the wild, although chocolate wattled-bats are prone to a mite infection around the eyes. In extreme cases, this infection can cause the skin to grow over the eyes (mainly over the right eye). The mites can be treated with a single drop (0.05 ml) of selamectin (Revolution®, kitten dose; diluted 1:10) on the back of the neck.

Specific breeding requirements

Although microbats are easy to keep in captivity they are quite difficult to breed. For successful breeding the microclimate of enclosures needs to mirror that of the wild. Females should be housed in a single enclosure separated from males. Males should only be introduced into the enclosure one at a time during the natural breeding season of the particular species.

Institutions with specialised expertise

Australasian Bat Society Email: president@ausbats.org.au Internet: http://ausbats.org.au	Perth Zoo Perth, Western Australia Phone: (08) 9474 0444 Email: email@perthzoo.wa.gov.au Internet: http://www.perthzoo.com.au
Taronga Zoo Sydney, New South Wales Phone: (02) 9969 2777 Email: tz@zoo.nsw.gov.au Internet: http://taronga.org.au/taronga-zoo	Zoos Victoria Veterinary Departments: Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville

Bibliography for microbats

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Churchill, S 2009, *Australian bats*, 2nd edn, Allen and Unwin, Sydney.

Jackson, S 2003, 'Bats', S Jackson (ed.), in *Australian mammals:biology and captive management*, CSIRO Publishing, Melbourne, pp. 297–350.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Lollar, A & Schmidt-French, B 2002, *Captive care and medical reference for the rehabilitation of insectivorous bats*, Bat World Publication, Mineral Wells.

Olsson, A & Woods, R 2008, 'Bats', in L Vogelneust & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 465–502.

Van Dyck, S & Strahan, R (eds) 2008, *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Dingo

The dingo is a sub-species of wolf that originated in Asia about 15,000 years ago and transported to Australia approximately 5,000 years ago. Dingoes were bred by humans and did not exist in nature previously. General information on the biology of this species' biology is provided in Corbett (2001).

During research on wild populations, purebred (*Canis lupus dingo*), crossbred and wild, domestic (*Canis lupus familiaris*) dogs may be encountered.

Dingo – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Dingo, <i>Canis lupus dingo</i> .	Common.	Widespread across Australian mainland, but absent from Tas.	Primarily mammals, but birds, reptiles and invertebrates are taken occasionally.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999 - List of Threatened Fauna*

Capture, handling, marking for identification, transport

Tractable dingoes can be handled in the same way as domestic dogs but should be muzzled because of their unpredictable nature. Dingoes that will not tolerate handling can be confined to a small pen and caught in a net. To avoid the handler being bitten, the head of the dingo must be restrained. The dingo can then be injected with an anaesthetic agent if necessary.

Wild dingoes can be caught with padded leg-hold traps. Once caught, the head should be restrained using a dog-catching noose and the animal pinned to the ground on its side. The dingoes might then be strapped to a restraining board to permit procedures such as blood collection, and radio collaring without sedation. Dingoes can also be captured using tranquilliser darts (see Corbett 2001; Hulst 2008).

Captive husbandry

Dingoes require a pen with shelter. Fences need to be constructed to prevent animals digging, climbing or jumping their way out. The suggested minimum area for a compatible pair of dingoes is 220 m², with an additional 6 m² for each additional animal. Solid fencing may be required between enclosures if there is a high level of aggression between animals.

Routine sample/data collection

Standard data including sex, weight and head length may be collected. Blood can be collected, as in a domestic dog, from either the cervical vein or jugular vein.

Anaesthesia, analgesia and sedation

Sedation and anaesthesia should only be performed by trained personnel. Food and water should be withheld for 12 hours prior to general anaesthesia. Any sedatives and anaesthetic agents used for domestic dogs can be used in dingoes at similar dose rates. (See Appendix III: Anaesthesia and sedation)

Analgesics are also used at similar dose rates used for domestic dogs including butorphanol (0.1 mg/kg), buprenorphine (0.01 mg/kg) and meloxicam (0.2 mg/kg).

Humane killing

Dingoes can be humanely killed using a firearm, or an overdose of sodium pentobarbitone (150 mg/kg) injected IV.

Health issues, disease control and zoonoses

Dingoes can be afflicted by the same range of diseases and parasites as domestic dogs and are vaccinated and wormed accordingly. The main zoonotic disease that affects dingoes is hydatidosis, caused by the *Echinococcus granulosus* tapeworm. Not feeding offal and regular treatment with anthelmintics will minimise the risk of infection.

Specific breeding requirements

Dingoes breed readily in captivity. Female dingoes usually become sexually mature at two years of age. They produce one litter of pups per year, containing between 1–10 individuals, with an average of 5 pups. Mating season is from April to May. Gestation period is 63 days. Pups are weaned by 8 weeks and are fully independent at 3–4 months.

Institutions and individuals with specialised expertise

Institutions

Perth Zoo Perth, Western Australia Phone: (08) 9474 0444 Email: email@perthzoo.wa.gov.au Internet: http://www.perthzoo.com.au	Zoos Victoria Veterinary Departments: Melbourne Zoo Phone: (03) 9285 9300 Email: mz@zoo.org.au Internet: http://www.zoo.org.au Healesville Sanctuary Phone: (03) 5957 2800 Email: hs@zoo.org.au Internet: http://www.zoo.org.au/healesville
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Individuals

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Bibliography for dingo

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Corbett, L 2001, *The dingo in Australia and Asia*, 2nd edn, J.B. Books, Marleston.

Hulst, F 2008, 'Dingoes', in L Vogelneust & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 527–40.

Jackson, S 2003, 'Dingoes', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 381–407.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Marine mammals

Marine mammals in Australian waters are represented by the orders Cetacea (whales and dolphins), Carnivora (seals and sea lions), and Sirenia (dugongs). While the seals are the group most commonly maintained in captivity, a range of seal species are also the subject of extensive wildlife research, especially in Antarctic waters.

While dolphins are kept in captivity, the dugong and whales are rarely kept. These three groups are not dealt with in these guidelines. If you wish to conduct research on dolphins, dugongs or cetaceans, it is imperative that you contact people with specialist expertise in dealing with your chosen species.

Seals

This group includes the fur seals and sea lions (Family Otariidae) and the elephant and leopard seals (Family Phocidae). Seals breed on land and forage at sea, with different species occurring from southern Australia to the Antarctic. Information on the general biology of the various species occurring in Australian waters is provided in Van Dyck & Strahan (2008).

Seals – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Australian fur seal, <i>Arctocephalus pusillus doriferus</i> .	Common.	NSW, Vic., Tas. and SA.	Fish, squid.
New Zealand fur seal, <i>Arctocephalus forsteri</i> .	Common.	NSW, Vic., Tas., SA, WA.	Cephalopods, fish and seabirds.
Antarctic fur seal, <i>Arctocephalus gazella</i> .	Common.	Antarctic and sub-Antarctic waters and islands.	Fish.
Sub-Antarctic fur seal, <i>Arctocephalus tropicalis</i> .	Vulnerable.	Sub-Antarctic waters and islands.	Fish, cephalopods.
Australian sea lion, <i>Neophoca cinerea</i> .	Vulnerable.	SA, WA.	Fish, cephalopods and crustaceans.
Southern elephant seal, <i>Mirounga leonine</i> .	Vulnerable.	Antarctic and sub-Antarctic waters.	Squid.
Leopard seal, <i>Hydrurga leptonyx</i> .	Common.	Antarctic and sub-Antarctic, occasional visitor to NSW, Vic., Tas., SA and WA.	Penguins, seals, fish, krill.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

Physical restraint is suitable for minor procedures such as injections, and inspection of minor injuries, however, this should only be undertaken by experienced personnel. Herding boards may be used or squeeze cages of appropriate size for the species.

Otariids are the most agile of the seals on land and the safest capture method for both handler and animal is a conical net of appropriate size for the species and individual, with a circular opening at the end large enough to allow the snout to protrude. The body of the net should have a small mesh size (less than 10 mm) with

the apex constructed from a canvas or a fine weave netting to protect the eyes and prevent the teeth from snagging on the fabric. The seal or sea lion fleeing from the catcher will run into the net and the snout will protrude through the hole. Once in this position, other catchers can immobilize the animal by physical restraint holding the pectoral flippers against the body and off the ground. Then either chemical or gaseous sedation or anaesthesia can be safely administered. This method may be used for otariids up to 100 kg. Chemical restraint administered by dart gun or pole syringe should be used on larger animals; however a high level of expertise is required for this procedure. Expert advice should therefore be sought (e.g. wildlife and veterinary experts).

Similar capture methods can be used for phocids, however, an alternative net design may be more appropriate. Because phocids tend to have a more prone posture on land, they can be physically restrained using a head bag. Once the head bag is in position, the animal can be manually restrained with one person straddling its shoulders, and another, its hindquarters. A third handler can then perform minor procedures, such as blood collection and flipper tagging.

Each animal should have a means of permanent individual identification such as a plastic flipper tag that is positioned on the trailing edge of the flipper, a tattoo or a microchip.

Smaller seals (less than 50 kg) may be transported by road or air in plastic or wooden carry cages or crates designed for dogs, provided the animal has sufficient space to turn around. Care should also be taken to ensure that there is good ventilation and that the crate or cage is not in direct sunlight. Larger seals will require custom built crates designed to allow the animal room to move and thermoregulate. The length of the crate should be 1.5 times the body length of the animal, the height should be 1.5 times the sitting, head height, and the width should be twice the width of the animal at the shoulders.

For animals in good body condition, hyperthermia will be a greater concern in transportation than hypothermia. Crated seals should be kept out of direct sun and wetted down periodically with water spray if ambient temperatures are above the norm for the species concerned. Ice blocks on top of the crate can also help by cooling the air and allowing cold water to drip onto the animal. Good ventilation is essential and in some circumstances, air-conditioned vehicles may be required. While seals do not often drink water, it should be offered every four hours during a long journey either as fresh water or as ice blocks.

Captive husbandry

Seek expert advice from zoos with colonies. In captivity, seals require both water and a dry haul-out area. Large seawater pens and oceanarium pools are ideal but allow limited viewing and handling of the animals. Circular or curved pools are best because they allow better water circulation, ease of cleaning and cause fewer abrasions to animals. A variety of building materials may be used but the pools need to be watertight, easily cleaned, and not readily punctured by teeth. Similarly the decking material of the haul-out area should be non-abrasive. Floors should be hard and durable, impervious to water, easily sanitised and resistant to chemical disinfectants. The haul-out areas should have good drainage and contaminated water should not flow back into the pool. Drains should have traps to collect excreta and be of suitable diameter (greater than 10 cm). Walls and ceilings should also be impervious and easily cleaned and not offer shelter for vermin. Electrical fittings should be grounded, easy to clean, and be corrosion resistant.

Cages are sometimes used for short-term holding of seals. They should be constructed of non-toxic, corrosion-resistant materials. The gap between wires or bars should prevent a full-mouth grasp to prevent gnawing or mouth injuries and to prevent handlers being bitten through the mesh. There should be good drainage to facilitate cleaning.

Minimum space requirements are difficult to determine. However, a general rule is that animals should have a pool that is 2 or more times the adult body length and a depth of not less than one-half to two-thirds the adult length. Generally, smaller species are maintained in pools considerably bigger than this. More active fur seals and sea lions need more space than the more sedentary phocids.

Clean water is important. All species require natural or artificial sea water and access to fresh water. A safe natural salinity range is 2.5–3.5‰ with pH of 7.5–8.2.

The filtration system should remove all animal wastes, prevent growth of microbes, provide an environment relatively free of toxic chemicals and maintain water clarity. Chemicals such as chlorine, bromide and ozone may also be used to assist in the maintenance of a clean environment. All chemicals should be used judiciously and water analysed daily to check for disease potential. Seek expert opinion from zoos with colonies.

The light in the enclosure should duplicate, as closely as possible, the photoperiod, photospectrum and photo intensity of light in the wild environment. Moulting and reproduction are likely influenced by light and it is unlikely that a 1:1 day:night ratio using incandescent or fluorescent lights will provide the proper stimulus for normal physiological function. For animals maintained outdoors, it may be necessary to provide shade areas in the enclosure to allow animals to escape intense light. This may be achieved by covering the pools with shade cloth or sails.

The natural environmental temperature of the species in the wild should be reproduced as closely as possible. Excess heat is as threatening as excess cold. Seals and sea lions exposed to direct summer sun can face heat prostration in less than 1 hour. Temperatures of 26 °C to 28 °C are probably the thermal maximum for most well-blubbered seals. Higher temperatures can be alleviated by providing shade, cold-water sprays or ice blocks. What is most critical is not the absolute temperature but the rate of change. Indoor facilities should be ventilated to allow noxious gases to escape.

Diets for captive marine mammals

Captive diets should include fish, squid and octopus, supplemented with vitamins B1 and E. Expert advice should be sought (e.g. from zoos with colonies). Information on diets for captive marine mammals is also provided in Barnes, Higgins & Gray (2008).

Routine sample/data collection

Captive seals can be trained to present for non-invasive sampling such as dental examination, venipuncture, or oral and rectal swabs. In these cases, diazepam is contraindicated as it will inhibit the control of the training and increase the risk of the handlers being bitten. However, more invasive sampling will require sedation, or general anaesthesia.

The most commonly used veins for blood sampling are the caudal gluteal veins and the hind flipper inter-digital veins, followed by the brachial vein and jugular vein. Because the intra-vertebral extra-dural venous sinus is not easily accessible in otariids, venipuncture from this site is not recommended.

The gluteal veins run parallel to the spine from the tarsus to the pelvis and deep to the superficial gluteal muscle. With the animal in sternal recumbency and the hind flippers spread laterally, a groove can be palpated lateral to the sacral vertebrae. The needle can be directed perpendicularly into the groove, approximately one-third of the distance caudal to the hip joint. The needle can be 'walked' into the vein if venipuncture is not achieved at the first attempt. For smaller seals (less than 25 kg) a 20- to 22-gauge, 25 mm needle is sufficient. Larger animals may require an 18-gauge, 38 mm needle.

The inter-digital veins are accessed on the dorsal webbing of the hind flipper, with the phalanges held spread by an assistant. The slightly larger veins traversing the tarsus may be more accessible than the small inter-digital veins in the webbing. They are located on the dorsal aspect of the tarsus at the junction of the haired and non-haired skin. Because these veins are small, venipuncture may be facilitated by warming the flipper to cause vasodilation or by using a tourniquet. A heparinised butterfly catheter should be used to prevent clotting.

The brachial vein drains the fore flipper and runs towards the body in the anti-brachial web between the elbow and thoracic wall. It is accessed with the seal in dorsal recumbency. Jugular venipuncture is achieved by palpating the jugular groove in the mid-cervical region and restricting return flow with one hand while placing the needle with the other.

The preferred venipuncture site on phocids is the intra-vertebral extra-dural venous sinus in the lumbar region. The animal must be restrained in sternal recumbency and the needle (20-gauge, 25–38 mm for smaller seals of less than 30 kg; or 18-gauge, 90 mm spinal needle in larger animals) directed perpendicularly in the midline between two dorsal spinal processes into the intervertebral space. Large volumes of blood can be collected rapidly from this site. More difficult are the inter-digital veins on the plantar aspect of the hind flipper. As with otariids, warming the flipper and using a heparinised 20-gauge needle attached to a butterfly catheter can assist if this approach is used.

Anaesthesia, analgesia and sedation

Sedation and anaesthesia should only be performed by trained personnel. Before chemical restraint of unrestrained seals is attempted, care should be taken to ensure that darted animals cannot flee into water or into a situation where they become inaccessible on cliffs or rocky coast. Darts are generally fired into the thinner skin of the lumbar or gluteal region. Where physical restraint is possible, inhalation agents may be used to induce anaesthesia. Parenteral induction is also routinely used in phocids by using the intra-vertebral extra-dural vein, or by IM injection into the gluteal muscles using a needle appropriate to the size of the animal. For the latter, a 1.2 metre extension pole may be used. Seek expert advice before considering capture by darting.

Analgesics that have been used in pinnipeds include butorphanol (0.1 mg/kg), flunixin (1 mg/kg), ketoprofen (1 mg/kg) and carprofen (2–4 mg/kg).

Humane killing

If the animal is fully alert or where there may be risk to handlers, sedation should first be achieved using one of the methods outlined in the previous section. The dosage of sodium pentobarbitone is 150 mg/kg, IV. Alternative sites for moribund, comatose or anaesthetized animals are IP, IH or IC. If humane killing is conducted in the field, the carcass should be disposed of appropriately to prevent scavengers such as sea birds having access to the barbiturate-contaminated tissues. In field situations, the use of firearms to kill moribund seals is the method of choice as it is less stressful because restraint is not required. It is also safer for an experienced marksman, and rapid and humane when the animal is shot through the brain.

Health issues, disease control and zoonoses

Extensive information on health and disease issues in seals is provided in Barnes, Higgins & Gray (2008). Many of the health problems observed in captive seals are related to poor husbandry or dietary deficiencies, such as thiamine deficiency, vitamin E deficiency or hyponatraemia, that are often resolved by changing management practices.

A range of bacteria and viruses have been isolated from seals including *Salmonella* spp., *Mycobacterium pinnipedii*, *Brucella* spp., *Campylobacter* spp., *Burkholderia pseudomallei*, influenza A, and morbillivirus. The aforementioned bacteria can cause zoonotic diseases.

Respiratory mites, lice and nematodes have all been reported from Australian seals. Similar anthelmintics may be used on seals at equivalent dose rates as for terrestrial carnivores (see Appendix IV).

Specific breeding requirements

Australian seals are monoestrous with synchronous breeding and embryonic diapause. The Australian sea-lion is an exception among seals in that it exhibits synchrony within breeding colonies but marked asynchrony between colonies over its range and an extended breeding cycle of approximately 18 months, rather than annually. Pseudopregnancy, defined as elevated serum progesterone in a non-pregnant animal, has been documented in some otariids and phocids. Placentation is zonal and endotheliochorial. In captivity, pregnant females should be provided with ample haul-out space and seclusion. Presentation may be cephalic or posterior and both dystocia and twinning are infrequent. Management of dystocia is similar to that in domestic carnivores. Parturition may be induced by prostaglandin F₂ (Cloprostenol 500 mg, IM), while oxytocin (20 IU, IM) may be used to stimulate milk let down. Contraception may be achieved by separation of males and females during the breeding season, castration, or use of gonadotropin-releasing hormone agonists such as Deslorelin (2 × 4.7 mg implants, SC) or a porcine zona pellucida vaccine.

Institutions and individuals with specialised expertise

Institutions

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Bibliography for marine mammals

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Barnes, J, Higgins, D & Gray, R 2008, 'Pinnipeds (SEALS)', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 541–89.

Kirkwood, R & Goldsworthy, S 2013, *Fur seals and sea lions*, CSIRO Publishing, Melbourne.

Van Dyck, S & R Strahan (eds) 2008, *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney.

Native rodents

There are approximately 60 species of native rodents in Australia, comprising approximately 25% of all Australian mammal species. (Descriptions of the various species is provided in Van Dyck & Strahan 2008.) While some species have been studied extensively, others are only poorly known, partly because of rarity and due to limited and/or remote distributions. All Australian rodent species are in the subfamily Murinae, a very successful group that once occupied nearly every habitat type in Australia. Many species have suffered great reductions in distribution, with at least 11 species becoming extinct since European settlement (Van Dyck & Strahan 2008). The rodents are a relatively diverse group with animals ranging in size from only a few grams (for example, the delicate mouse) to just over 1 kilogram (for example, the water rat).

The table below shows the species most commonly used for research.

Native rodents – overview			
Name	Status under EPBC Act ^{1,2}	Distribution	Natural diet
Desert mouse, <i>Pseudomys desertor</i> .	Least concern.	Wide spread in Australia's central arid zone.	Herbivorous, feeding on grasses, shoots, rhizomes, seeds and flowers.
Spinifex hopping mouse, <i>Notomys alexis</i> .	Least concern.	Wide spread throughout Australia's central and western arid zone.	Omnivorous, eating seeds, root shoots and invertebrates in response to availability.
Long-haired rat, <i>Rattus villosissimus</i> .	Least concern.	Occurs in Australia's central and northern arid and semi-arid tropics.	Omnivorous.
Bush rat, <i>Rattus fuscipes</i> .	Least concern.	Coastal and sub-coastal forests of south-eastern Australia.	Omnivorous, feeding mainly on grasses and sedges supplemented by fruits, seeds and arthropods.
Swamp rat, <i>Rattus lutreolus</i> .	Least concern.	Coastal and sub-coastal forests of south-eastern Australia.	Mainly grasses and sedges and occasionally fruits, seeds and arthropods.
Sandy inland mouse, <i>Pseudomys hermannsburgensis</i> .	Least concern.	Occurs throughout Australia's central and western arid zone.	Omnivorous, feeding on roots, shoots, seeds, small tubers and invertebrates.
Ash-grey mouse, <i>Pseudomys albocinereus</i> .	Least concern.	Low heath and shrub land of south-western WA.	Predominantly herbivorous but will include arthropods in winter.
Silky mouse, <i>Pseudomys apodemoides</i> .	Least concern.	Restricted to dry mallee heath lands of north-western Vic., and eastern SA.	Omnivorous including Banksia nectar, seeds and arthropods.
Water rat, <i>Hydromys chrysogaster</i> .	Least concern.	Large coastal and inland distribution in permanent water bodies.	Opportunistic predator of large aquatic insects, fishes, crustaceans, mussels, frogs, lizards, small mammals and water birds. Fresh carrion may be taken.
Plains mouse, <i>Pseudomys australis</i> .	Vulnerable.	Restricted to the north-eastern corner of SA in the Lake Eyre Basin.	Predominantly a seed-eater supplementing the diet with vegetable matter and insects.
Chestnut mouse, <i>Pseudomys gracilicaudatus</i> .	Least Concern.	Coastal forest of Qld and NSW from Cooktown to Sydney.	A seed-eater supplementing the diet with vegetable matter, fungi and insects.

¹ EPBC Act = *Environment Protection and Biodiversity Conservation Act 1999*

² Status may be amended from time to time. For current status, refer to *Environment Protection and Biodiversity Conservation Act 1999* - List of Threatened Fauna

Capture, handling, marking for identification, transport

When trapping small mammals, such as rodents, there are a number of basic rules to follow. The number of traps set should always be limited to ensure that they can be cleared in a timely manner so that animals do not remain in traps for extended periods and become hyperthermic or hypothermic. Trap lines should be closed during extremely hot or extremely cold and wet weather conditions, as small animals can succumb rapidly. For nocturnal trapping, traps should be set around dusk and cleared commencing at dawn. If trapping is conducted during the day, the traps often need to be checked more regularly.

The most common trapping method for small mammals in arid and open habitats is pitfall trapping. Pitfall traps are made of polyvinylchloride (PVC) pipe of approximately 20–30 cm diameter and up to 60 cm in length with wire mesh at the bottom, or 10 L plastic or tin buckets with holes drilled in the base so that traps will drain if it rains. These are sunk into the ground with the top of the trap flush with the surrounding ground.

Pitfall traps should contain some sand or soil, leaf litter and small sticks or bark for shelter and protection from predators. A fence line of fine mesh (approximately 30 cm high) is placed vertically across the opening of the trap to channel animals into the trap. Depending on habitat and target species, long drift fences (tens of metres) with numerous pitfalls along their length, or individual traps with approximately 2.5 metres of drift fence either side of the trap are used. No bait is required. Traps need to be deep otherwise animals will escape, especially if trying to catch hopping mice. Animals can be removed from pitfall traps either by reaching in with a bare hand and grasping the scruff of the neck and transferring the animal into a bag, or by covering the hand with a calico bag and grasping the scruff of the neck through the bag, removing the animal from the trap then inverting the bag to cover the animal.

Forest-dwelling rodents are usually captured in Elliott traps, which are collapsible sheet metal traps available in a number of sizes. The size most commonly used for rodents is 23 cm × 9 cm × 8 cm. In colder areas, bedding should be provided inside traps, preferably futon filling. For rats, particularly arboreal species, smaller wire mesh cage traps can be set in trees, similar to the capture of some of the small to medium-sized possums (see the 'Medium and larger possums' and 'Smaller possums and gliders' sections).

Different baits are used depending on the target species, but a mixture of peanut butter, oats and golden syrup can be used to successfully trap most forest rodents. Plastic covers should be placed over the outside of all traps in cold and especially wet weather to prevent hypothermia. In hotter weather, care must be taken to ensure that traps are not placed in positions where they will receive full sunlight, as this could result in animals becoming overheated. Animals can be removed from traps by placing a calico bag over one end of the trap and encouraging the animals to move into the bag, for example by blowing onto them. When using Elliott traps, the trap can be tipped to slide the animal into the bag. Larger species, such as the water rats can be trapped in cage traps approximately 40 cm high × 40 cm wide × 70 cm deep.

Animals trapped in areas with high average daily temperatures should not be handled during the hottest part of the day. In particular, if a large number of animals are trapped, processing of these will often progress into the hottest time of the day. In this situation, the bags containing the animals can be hung in a cool, shady spot until they are processed and released in the late afternoon. Individual animals should always be released at the exact point of capture.

For handling, animals need to be either grasped firmly by the scruff of the neck or by positioning the head between the index and middle fingers of one hand and restraining the bottom half of the body with the other hand. Rodents should not be held solely by the tail, as this can result in degloving, where the skin is stripped from the tail. However, for many measurements in the field, rodents are most easily handled in a soft cotton bag, and this appears to be considerably less distressing for the animal than direct physical restraint.

Animals can be transported over short distances for 2–3 hours in calico bags or pillow cases. However, it is advisable to place the bags inside a secure box or cage trap, as rodents will chew their way out of a bag. For longer periods of transport, animals should be placed in small cages, either plastic or metal. The cages should be furnished with substrate such as leaf litter, shredded paper or sand, or cardboard tubes. Animals can be housed in cages for several days before transport but must be provided with fresh food and water daily. During transport in hotter weather, air conditioning should be turned on in the vehicle.

The medium to large rodents can be marked using ear tags, although these are not ideal as they often tear out. Some of the smaller rodents are amenable to ear tattooing. Some people suggest microchips (for example, passive integrated transponder tags; 'PIT' tags), but there have been some issues with these migrating and causing issues in very small animals. Toe clipping is not permitted as it may cause excess trauma. Ear notching is also not recommended as ears damaged from fighting may resemble notches.

Captive husbandry

Many species can be successfully held in captivity, including water rats, bush and swamp rats, tree rats and hopping mice (see Jackson 2003). Animals can be held in cages or aquaria with an appropriate substrate such as sand, leaf litter, paper or sawdust, with the last two of these being the easiest to keep clean. Structures for the animals to nest or hide in, such as nest boxes or plastic or cardboard tubes should be provided.

Water rats must be provided with areas of water at least 100 cm × 100 cm × 50 cm and arboreal species should be given climbing structures such as vertical and horizontal branches. When spinifex hopping mice are housed in colonies, reintroducing them back into the colony or introducing new stock must be conducted cautiously, as colony members will show extreme aggressiveness to newcomers and even kill individuals.

Diets for captive native rodents

Australian rodents have a wide diversity of diets. Information on appropriate diets for native rodents is provided in Jackson (2003). Diet should generally include small amounts of standard rat or mouse pellets to ensure adequate nutrition. Animals should also be supplied with sticks and branches to chew.

Routine sample/data collection

Standard data collection should include sex, body mass, head length and hind foot length.

Ear biopsies very close to the outer margin of the ear can be collected for DNA analysis and are stored in 70% ethanol. Blood up to a maximum volume of 10% of the body weight can be collected from the lateral tail vein, saphenous vein or femoral vein.

Anaesthesia, analgesia and sedation

Anaesthesia should only be performed by trained personnel. Animals are commonly anaesthetised by using isoflurane (4–5% for induction; 1.5–3% for maintenance) and oxygen 1.5 L/minute either using an anaesthetic box or a small face mask. There have been no pharmacokinetic studies on any analgesics for this group, thus if pain relief is required you must seek expert veterinary advice (e.g. zoo or wildlife veterinarian).

Humane killing

Native rodents can be humanely killed using an overdose of sodium pentobarbitone (50 mg/kg) injected IP or IC. If using a concentrated solution of sodium pentobarbitone (e.g. >300mg/ml, Lethabarb®), prior anaesthesia or sedation (e.g. using Zoletil®, 10 mg/kg IM) is essential to prevent pain or irritation from the sodium pentobarbitone solution.

Health issues, disease control and zoonoses

Extensive information on health and disease issues in rodents is provided in Breed & Eden (2008) and Jackson (2003). Spinifex hopping mice are susceptible to death from Tyzzer's Disease. This disease is a bacterial infection that affects the liver. Rodents should be checked regularly for mites and treated if infested. Seek expert veterinary advice (e.g. experienced wildlife veterinarian).

Specific breeding requirements

Information on breeding rodents in captivity is provided in Jackson (2003). For successful breeding in captivity, male and female spinifex hopping mice are initially housed individually in the same cage with a clear perspex divider containing holes to allow the mice to see and smell one another. This divider can be left in place for successively decreasing time periods. This approach has been used to successfully reduce aggression when introducing males and females of this species.

Female spinifex hopping mice that are pregnant or in early lactation are extremely aggressive and will attack and injure males or other females. During this period they should be housed individually. Females will also behave aggressively towards males until the males acquire the group odour.

Institutions and individuals with specialised expertise

Institutions

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Bibliography for native rodents

Australian Government, Department of Environment 2009, *EPBC Act List of Threatened Fauna*, <http://www.environment.gov.au/cgi-bin/sprat/public/publicthreatenedlist.pl>.

Breed, B & Ford, F 2007, *Native mice and rats*, CSIRO Publishing, Melbourne.

Breed, A & Eden, P 2008, 'Rodents', in L Vogelnest & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 503–26.

Jackson, S 2003, 'Rodents', in S Jackson (ed.), *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne, pp. 351–80.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Old, J, Connelly, L, Francis J & Gogler, J 2007, 'Haematology and serum biochemistry of the Carpentarian rock-rat (*Zyomys palatalis*)', *Comparative Clinical Pathology*, vol. 16, pp. 249–52.

Old, J, Connelly, L, Francis, J, Branch, K, Fry, G & Deane E 2005, 'Haematology and serum biochemistry of three Australian desert murids, the plains rat (*Pseudomys australis*), the spinifex hopping-mouse (*Notomys alexis*) and the central rock-rat, (*Zyomys pedunculatus*)', *Comparative Clinical Pathology*, vol. 14, pp. 130–37.

Van Dyck, S & Strahan R (eds) 2008, *The mammals of Australia*, 3rd edn. Reed New Holland, Sydney.

Appendices

Appendix I: Taxonomic key for Australian native mammals (Class: Mammalia)

Subclass Prototheria

Order Monotremata

Family Tachyglossidae (echidna)

Family Ornithorhynchidae (platypus)

Subclass Theria

Infraclass Marsupialia

Order Dasyuromorphia (carnivorous marsupials)

Family Dasyuridae (such as antechinuses, quolls, Tasmanian devil)

Family Thylacinidae (Tasmanian tiger)

Family Myrmecobiidae (numbat)

Order Peramelemorphia

Family Peramelidae (bandicoots)

Family Thylacomyidae (bilbies)

Order Notoryctemorphia

Family Notoryctidae (marsupial moles)

Order Diprotodontia

Suborder Macropodiformes

Family Macropodidae (kangaroos, wallabies)

Family Potoroidae (potoroos, bettongs)

Family Hypsiprymnodontidae (musky rat-kangaroo)

Suborder Vombatiformes

Family Vombatidae (wombats)

Family Phascolarctidae (koala)

Suborder Phalangeriformes

Family Phalangeridae (brushtail possums, cuscuses)

Family Pseudocheiridae (ringtail possums, greater glider)

Family Petauridae (gliders, Leadbeater's possum, striped possum)

Family Burramyidae (pygmy-possums)

Family Acrobatidae (feathertail glider)

Family Tarsipedidae (honey possum)

Infraclass Eutheria

Order Rodentia

Family Muridae (native murid rats and mice)

Order Carnivora

Family Canidae (dingo)

Family Otariidae (eared seals such as fur seals, sea lions)

Family Phocidae (earless seals such as elephant seal, leopard seal)

Order Chiroptera bats

Suborder Megachiroptera megabats

Family Pteropodidae (flying foxes, fruit bats)

Suborder Microchiroptera (microbats)

Family Emballonuridae (sheath-tail bats)

Family Megadermatidae (ghost bat)

Family Rhinolophidae (horseshoe bats)

Family Hipposideridae (leaf-nosed bats)

Family Vespertilionidae (vespertilionid bats)

Family Molossidae (freetail bats)

Appendix II: General blood sampling guidelines

There are general rules that should be followed when blood sampling animals. It is regarded as safe to take up to 8–10% of an animal's blood volume in a one-off sampling event, with the total volume of blood calculated as approximately 10% of body mass. However, if repeated blood sampling of individual animals is required over relatively short time periods, samples should be smaller to allow animals to recover. Alternatively, red blood cells can be stored in a preservative solution and replaced each 24 hours if longer-term sampling is necessary. In addition, blood sample volumes should always be the minimum required to achieve adequate sampling for the project. Follow the general guidelines as provided in the *Guidelines to promote the wellbeing of animals used for scientific purposes: The assessment and alleviation of pain and distress in research animals 2008*.

Blood samples from animals in the wild: special consideration must be given when blood sampling from animals caught from the wild, as these will be of unknown health, nutritional and hydration status. Factors such as prolonged drought may mean that sampling regimes should be far more conservative than those for captive animals that have access to food and water ad libitum. In addition, repeated sampling of blood from wild animals generally requires repeated venipuncture because the use of catheters is usually not possible. This may not be sustainable beyond a certain number of samples or may require minimum time periods between resampling. Always seek expert advice.

Appendix III: Anaesthesia and sedation

Anaesthesia and sedation should **only** be performed by trained and competent personnel because of significant risk to the animal if not performed well. Monitoring is essential and will vary with the species.

Most mammals can be sedated with 1–2 mg/kg diazepam, IM. Midazolam (0.25–0.50 mg/kg, IM) can be used to sedate pinnipeds. Medetomidine (0.03–0.08 mg/kg, IM) is useful for sedating dingoes and flying foxes. Medetomidine can be reversed with atipamezole given at five times the medetomidine dose.

For animals that can be physically restrained, inhalation anaesthesia using 5% isoflurane for induction is the method of choice. This is delivered using a non-rebreathing circuit with an oxygen flow rate of 200 mL/kg/minute with a minimum of 1 L/minute for mammals weighing less than 10 kg. Induction is via a mask placed over the face. Maintenance generally requires 2% isoflurane, but this varies between species and individuals.

If gaseous anaesthesia is not possible, injectable agents can be used. Tiletamine/zolazepam (Zoletil®) is the most commonly used agent and is given IM. Dose rates are in Table 1. For tractable dingoes, general anaesthesia can be induced with propofol (5 mg/kg, IV), alfaxalone (2 mg/kg, IV) or thiopentone (10 mg/kg, IV). Propofol (8–10 mg/kg, IV) can also be used to anaesthetise flying foxes.

Heart rate, respiratory rate and body temperature of selected monotremes and marsupials are shown in Table 2.

Table 1: Dose rates of tiletamine/zolazepam for immobilisation of selected mammals (see Holz, 2007)^{1,2}

Species	Tiletamine/zolazepam dose ¹
Echidnas	5–7 mg/kg
Dasyurids	10 mg/kg
Koalas	4–10 mg/kg
Wombats	3–8 mg/kg
Possums and Gliders	5–10 mg/kg
Macropodids	10 mg/kg
Otariids	1.2–2.0 mg/kg
Phocids	1.0–1.4 mg/kg
Dingoes	8–10 mg/kg
Flying Foxes	10–40 mg/kg

¹ Holz, P 2007, 'Monotremes', in G West, D Heard & N Caulkett (eds), *Zoo animal and wildlife immobilization and anesthesia*, Blackwell Publishing, Ames, pp. 337–39.

² Holz, P 2007, 'Marsupials', in G West, D Heard & N Caulkett (eds), *Zoo animal and wildlife immobilization and anesthesia*, Blackwell Publishing, Ames, pp. 341–46.

Table 2: Heart rate, respiratory rate and body temperature of selected monotremes and marsupials (Holz, 2007)^{1,2}

Species	Heart rate (beats/minute, BPM)	Respiratory rate (breaths/minute)	Temperature (°C)
Platypus	60–150	25	32.5–34.5
Echidna	80–110	8–14	28–33
Tasmanian devil	85–120	20	31–38
Sugar glider	200–300	16–40	35–36
Koala	65–90	10–15	35.5–36.5
Common wombat	90–120	20	35.4–36.7
Macropodid	60–150	10–30	35–36.5

¹ Holz, P 2007, 'Monotremes', in G West, D Heard & N Caulkett (eds), *Zoo animal and wildlife immobilization and anesthesia*, Blackwell Publishing, Ames, pp. 337–39.

² Holz, P 2007, 'Marsupials', in G West, D Heard & N Caulkett (eds), *Zoo animal and wildlife immobilization and anesthesia*, Blackwell Publishing, Ames, pp. 341–46.

Appendix IV: Health issues, disease control and zoonoses

Some information on health issues, disease control and zoonoses has been provided in each of the sections for specific taxonomic groups, and substantial information is also available in Jackson (2003), Vogelnest and Woods (2008) and Ladds (2009). The following information provides additional details.

Australian Bat Lyssavirus (ABL) has been isolated from the black flying fox, spectacled flying fox, grey-headed flying fox, little red flying fox, and the yellow-bellied sheath-tail bat. Signs in affected bats vary and include an inability to fly, hindquarter paresis, general weakness, and occasionally aggression. Transmission is via saliva (bites). Affected bats should be killed humanely and brains submitted for evaluation. Due to its zoonotic potential anyone handling bats must be vaccinated against rabies.

Bertiella obesa is the small intestinal tapeworm of koalas. The worm causes debilitation in large numbers of koalas and uses an arthropod intermediate host. Treat with praziquantel (5 mg/kg).

Caecal stasis primarily occurs in ringtail possums when treated with antibiotics or when maintained on an inappropriate diet high in sugar. Ring-tailed possums are mainly folivores and, if fed excessive quantities of fruit, can develop caecal stasis. Affected animals appear weak and bloated with decreased faecal output. Treatment involves increasing the fibre content of the diet by providing more browse.

Capture myopathy occurs as a result of prolonged sustained stress and exertion, which causes heat and lactic acid build up. Heat-stressed animals will lick their forelegs. Clinical signs are variable. Peracute death may occur within hours of capture. Acute capture myopathy leads to death in 3–4 hours due to hyperthermia and shock. Individuals show increased heart and respiratory rates, reluctance to move and muscle rigidity. Subacute capture myopathy occurs several hours to days post-capture. Affected animals are ataxic and produce dark-coloured urine due to extensive muscle and kidney necrosis. Body temperature is usually normal. Muscles can rupture up to 4 weeks post-capture.

Treatment is usually unsuccessful, but dexamethasone has been effective in swamp wallabies (5 mg, IM). Preventative techniques include rapid but quiet capture, acclimating animals to handling and confinement, not capturing on hot days, ensuring adequate ventilation, abandoning capture if the animal becomes unduly excited or stressed, use of long-acting tranquilisers and sedatives such as diazepam, and ensuring animals are on a good plane of nutrition, particularly with respect to vitamin E and selenium. Low levels of these nutrients have been shown to predispose to capture myopathy.

Chlamydophilosis affects koalas. There are two species. *Chlamydophila pneumoniae* has been isolated from ocular lesions as well as the trachea, penis and kidney and *Chlamydophila pecorum* has been found in urogenital, rectal and ocular lesions. There are no recorded cases of koala chlamydophila infecting humans. Clinical signs are variable. The ocular form results in a mucopurulent conjunctivitis that may eventually lead to blindness. Urogenital disease manifests as soiling and staining of the fur around the rump as a result of cystitis. This may ascend to involve the kidneys. Reproductive tract infection includes cysts in the vicinity of the ovary, pyometra and vaginitis.

Diagnosis is by swabbing the conjunctiva, urogenital sinus or penile urethra for polymerase chain reaction (PCR) testing. The disease can be treated with a range of antibiotics in captive animals, but chronically affected, emaciated koalas have a poor prognosis for recovery. Glutaraldehyde (2%) applied for 1–10 minutes is the disinfectant of choice for chlamydophila. Chloramine (2%) is also effective.

Coccidiosis occurs most commonly in hand-raised eastern grey kangaroos and common wombats. Animals may die with no previous signs or develop severe dysentery prior to death. By the time clinical signs occur, treatment is generally unsuccessful but can be attempted using toltrazuril (25 mg/kg) orally for three days combined with sulphonamides (30 mg/kg, IM) and intravenous fluids to combat fluid loss.

Cryptococcosis is a disease of koalas caused by the fungus *Cryptococcus gattii*. The fungus is associated with eucalypts of the Murray Darling River system. Transmission is via aerosol and inhalation of desiccated discharges. Animal-to-animal or animal-to-human transmission has not been reported. Clinical signs are variable and include depression, coughing, sneezing, nasal discharge, anorexia, maxillary swelling, neurological signs, subcutaneous cervical abscessation and death.

Diagnosis is by smears of discharges or biopsies of affected areas, which display yeast-like organisms surrounded by a clear capsule. Culture or a blood test can also be used. Treatment options include ketoconazole (10 mg/kg, daily), fluconazole (3 mg/kg, daily) or itraconazole (20–40 mg/kg, daily) for up to 90 days.

Diarrhoea commonly occurs in hand-reared orphan marsupials. The causes are many and varied. Marsupials cannot digest lactose and cow's milk should never be fed. Di-vetalact, Biolac and Wombaroo (appropriate for the specific taxonomic group) are all satisfactory milks. However, feeding more than 10% body weight per day may cause diarrhoea. Some animals scour when starting on solid foods, particularly lush grass. This can be overcome by feeding a more coarse diet such as good quality lucerne hay. Infections may cause diarrhoea.

To identify an infectious agent a sample of faeces must be collected. A faecal float can be performed to check for worm eggs. A faecal smear can be stained with Diff-Quik to look for *Candida*, which appears as small oval, blue budding bodies. Treatment is with nystatin (100 000 IU/kg, PO) 3 times a day for 5 days. *Salmonella* and *Campylobacter* infections can be diagnosed by faecal culture and treated with appropriate antibiotics.

The cause of the diarrhoea may not be determined, in which case symptomatic treatment is necessary. Milk is withdrawn for a day and replaced with oral electrolyte solution. If the diarrhoea stops the milk can be gradually reintroduced. Any stresses should be minimised. Some joeys scour but continue to remain bright and gain weight. As long as this is happening, the diarrhoea may not be a cause for concern but should be monitored.

Hydatid disease is caused by the tapeworm, *Echinococcus granulosus*. Dogs and dingoes are the definitive host. They are not affected clinically but shed eggs into the environment. When consumed by macropods and wombats, *Echinococcus granulosus* forms cysts in the liver and lungs. Affected animals may develop difficulty breathing, exercise intolerance and emaciation. Prevention focuses on not feeding offal to carnivores and regular anthelmintic treatment. The eggs can infect humans who develop similar cysts. This is a reportable disease.

Hyponatraemia of seals occurs when plasma sodium levels decrease. Affected seals become extremely weak and uncoordinated. The condition is most common in seals maintained in fresh water and can be treated by administering 100–200 mg NaCl/kg of body weight, IP or PO. A daily dose of 3 g NaCl/kg of food prevents the disorder in pinnipeds that are kept in fresh water.

Internal parasites Macropodids are host to a number of internal parasites. *Strongyloides* can cause a haemorrhagic gastritis with ulceration. *Globocephaloides* is a hookworm found in the duodenum of young macropodids that can cause anaemia and death.

Treatment options include a single dose of ivermectin (0.2 mg/kg, SC) or moxidectin (0.2 mg/kg, SC). Alternatively, fenbendazole (7.5 mg/kg, PO) or oxfendazole (5 mg/kg, PO) can be administered daily for five days. Mebendazole causes bone marrow suppression and death and should not be used.

Lumpy jaw is primarily a disease of captive macropodids, beginning as gingivitis and progressing to abscessation of the muscles over the mandibles and maxilla, followed by osteomyelitis with tooth loss, bone reabsorption and fractures. Affected animals may have a purulent nasal discharge, obvious facial swelling, increased salivation, halitosis, difficulty eating and weight loss.

Treatment involves flushing abscessed material, curetting affected bone, removing teeth and long-term systemic antibiotics such as Tylan® (tylosin). Lumpy jaw occurs most commonly on over-stocked paddocks with a high faecal load. Daily raking and faecal removal will decrease the bacterial load in the paddock. Placing feed in trays or hay racks will also decrease exposure. Bread is not recommended as it forms a starchy paste on the teeth and gums promoting bacterial colonisation and infection.

Mycobacteriosis occurs most commonly in dasyurids and macropodids. It causes weight loss, breathing difficulties, lameness, abscesses, skin ulcers and neurological signs. Abscesses are found in multiple organs. There is no treatment and affected animals must be killed humanely. Exposure to organisms is by inhalation or ingestion of faeces and discharges. Atypical mycobacteria can cause pneumonia in immunocompromised people. *Mycobacterium ulcerans* also causes ulcerative skin disease in possums, koalas and humans.

Nephrosis leading to chronic renal failure in koalas is not uncommon, often secondary to a period of dehydration. Affected koalas are depressed, anorexic, drink excessively and have elevated blood urea. By the time clinical signs are present, treatment is usually too late to be successful but can be attempted using intravenous fluids. Prevention centres on providing access to water and fresh eucalyptus leaves. Periods of dehydration should be treated aggressively with intravenous fluids.

Pneumonia is generally bacterial. Often there are no preceding clinical signs, with sudden death being the result. Other cases demonstrate more typical signs of exercise intolerance, sneezing, coughing, or a mucopurulent nasal discharge. Treatment can be attempted using broad-spectrum antibiotics.

Pouch infections have been reported in brushtail possums, macropodids and koalas. *Pseudomonas* has been isolated from some of these. Hairs around the infected pouch are moist. The pouch itself contains a greasy, foul-smelling liquid. Treatment involves twice daily cleaning of the pouch with an antiseptic such as chlorhexidine followed by drying with cotton swabs.

Ringworm is caused by a fungus. Clinical signs include alopecia, erythema and hyperkeratotic scales that lift off with the hair. Infections may or may not be pruritic. Diagnosis is by scraping affected areas or culture. Treatment is effective using topical antifungal agents. Many cases clear up without treatment. The condition is a zoonosis.

Salmonella is a bacterium. Many mammals culture positive for *Salmonella* spp. and other enteric pathogens. Infected animals are usually asymptomatic but these bacteria can cause gastrointestinal disease in people. Vigorous hand washing with soap is enough to kill *Salmonella*.

Sarcoptic mange is caused by a mite and is seen most commonly in wombats but has also been diagnosed in koalas, macropodids and possums. Affected animals may be emaciated, blind, intensely pruritic and develop hair loss and the skin develops a thick dry crust. This crust may become secondarily infected and fly blown. Diagnosis is by skin scraping. Severely affected animals are best killed humanely as these are usually highly compromised, often having developed accompanying internal infections. If diagnosed before secondary complications have occurred, treatment can be attempted using ivermectin (0.3 mg/kg), moxidectin (0.3 mg/kg) or selamectin (6 mg/kg).

Thiamine deficiency in marine mammals is caused by feeding with fish species that contain thiaminase such as herring, smelt, mackerel and anchovy. Clinical signs include anorexia, lethargy, difficulty breathing, tremors, convulsions and death. Signs can be reversed by giving 100 mg thiamine, IM, and supplementing the diet with thiamine (25 mg/kg of fish).

Toxoplasmosis is a zoonotic infection caused by the protozoan parasite *Toxoplasma gondii*. Marsupials are extremely susceptible to toxoplasmosis. Signs include sudden death, blindness, depression, breathing difficulties and neurological signs. Treatment is usually unsuccessful. Prevention focuses on denying animal exposure to cat faeces and not feeding meat that has not been frozen, even if meant for human consumption. *Toxoplasma* cysts can be found in the muscles of animals such as wallabies. If pregnant women are infected, the disease can cause abortion and birth defects.

Tyzzler's disease is caused by *Clostridium piliforme*. It has been documented in Mitchell's hopping mice, spinifex hopping mice, common rock-rats and ringtail possums. Clinical signs include diarrhoea, dehydration, lethargy and sudden death. Treatment involves antibiotics, but is often too late. Prevention requires good husbandry and hygiene.

Vitamin E deficiency in marine mammals is caused by eating improperly stored dark-fleshed fish such as mackerel and tuna. Signs include myopathy, steatitis (inflammation of fat) and anaemia. Prevention is by proper storage of fish and supplementation with 100 IU of vitamin E/kg fish, once weekly.

Further Information

The Australian Wildlife Health Network (<https://www.wildlifehealthaustralia.com.au/>) is an Australian Government initiative of Wildlife Exotic Disease Preparedness Program managed by the Department of Agriculture. Fact sheets about specific wildlife diseases can be found on the website (<https://www.wildlifehealthaustralia.com.au/FactSheets.aspx>).

The Australian Registry of Wildlife Health (<http://www.arwh.org>) is a national wildlife health resource based at Taronga Zoo in Sydney. The website has information on common wildlife diseases (<http://arwh.org/common-diseases>).

The Wildlife Disease Association – Australasian Section (<http://www.wildlivedisease.org/wda/SECTIONS/Australasian.aspx>) is a professional association of biologists and veterinarians with knowledge of health and diseases of wildlife in conjunction with their ecology, biology and conservation.

The Australian Veterinary Association maintains a wildlife special interest group called the Australian Association of Veterinary Conservation Biologists (<http://conservation.ava.com.au>).

References

Jackson, S (ed.) 2003, *Australian mammals: biology and captive management*, CSIRO Publishing, Melbourne.

Ladds, P 2009, *Pathology of Australian native wildlife*, CSIRO Publishing, Melbourne.

Vogelnest L & Woods R (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne.

Appendix V: Terms of reference

The Review of the Australian Native Mammals Guides Working Committee will provide expert advice on the development, revision and finalisation of *A Guide to the Care and Use of Australian Native Mammals in Research and Teaching*.

The working committee will:

- provide expert advice on the form and content of the revision and preparation of the documents prior to public consultation and finalisation
- provide regular progress reports to the NHMRC
- present guidelines and documents to the AWC and to Council for endorsement.

Appendix VI: Working Committee membership

Members involved in development of the Guidelines

<p>Dr Kathrine Handasyde (Chair) Senior Lecturer Department of Zoology The University of Melbourne</p>	<p>Dr Peter Holz Veterinarian Tidbinbilla Nature Reserve</p>
<p>Dr Deborah Kelly Animal Welfare Conservation and Land Management Branch SA Department of Environment, Water and Natural Resources</p>	<p>Professor Marilyn Renfree University Laureate Professor and Ian Potter Chair of Zoology Department of Zoology The University of Melbourne</p>

Member from 2011 to 2014

<p>Dr Carole Webb NHMRC Animal Welfare Committee Member (2006–2009 Triennium, 2009–2012 Triennium, 2012-2015 Triennium)</p>
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Members before 2011

<p>Dr Justyna Paplinska Department of Zoology The University of Melbourne</p>	<p>Associate Professor Christine Wright NHMRC Animal Welfare Committee Member (2006–2009 Triennium, 2009–2012 Triennium) Department of Pharmacology The University of Melbourne</p>
<p>NHMRC staff Ms Mary Bate Ms Catherine Chippendale Ms Renee Trentini Ms Vivienne A Moyle</p>	<p>Special consultant RLC Feach Moyle March 2010 – June 2010</p>

Appendix VII: Legislation (including endangered species)

In Australia, legislation to protect animal welfare is the responsibility of state and territory governments. Although there are some differences in specific legislation requirements, in each case the *Australian code for the care and use animals for scientific purposes 8th edition 2013* (and subsequent iterations) is the basis for describing practices and procedures to protect the welfare of the animals used for scientific purposes.

Commonwealth	
<p><i>Environment Protection and Biodiversity Conservation Act 1999</i></p> <p>Administered by the Department of the Environment</p> <p>GPO Box 787 Canberra, ACT 2601 Phone: (02) 6274 1111</p> <p>Internet: http://www.environment.gov.au/topics/about-us/legislation/environment-protection-and-biodiversity-conservation-act-1999/about-epbc</p>	<p><i>Environment Protection and Biodiversity Conservation Amendment (Wildlife Protection) Act 2001</i></p> <p>Administered by the Department of the Environment</p> <p>GPO Box 787 Canberra, ACT 2601 Phone: (02) 6274 1111</p> <p>Internet: http://www.comlaw.gov.au/Details/C2004A0084</p>
Australian Capital Territory	
<p><i>Animal Welfare Act 1992</i></p> <p>Administered by the Department of Territory and Municipal Services</p> <p>GPO Box 158 Canberra, ACT 2601 Phone: (02) 6207 5111</p> <p>Internet: http://www.legislation.act.gov.au/a/1992-45/default.asp</p>	<p><i>Nature Conservation Act 1980</i></p> <p>Administered by the Environment and Sustainable Development Directorate</p> <p>GPO Box 158 Canberra, ACT 2601 Phone: 13 22 81</p> <p>Internet: http://www.legislation.act.gov.au/a/1980-20/default.asp</p>
New South Wales	
<p><i>Animal Research Act 1985</i></p> <p>Administered by the Department of Primary Industries</p> <p>Locked Bag 21 Orange, NSW 2800 Phone: (02) 6391 3100</p> <p>Internet: http://www.austlii.edu.au/au/legis/nsw/consol_act/ara1985134/</p>	<p><i>National Parks and Wildlife Act 1974</i></p> <p>Administered by the Department of Environment and Heritage</p> <p>PO Box A290 Sydney South, NSW 1232 Phone: (02) 9995 5000</p> <p>Internet: http://www.legislation.nsw.gov.au/xref/inforce/?xref=Type%3Dact%20AND%20Year%3D1974%20AND%20no%3D80&nohits=y</p>
<p><i>Threatened Species Conservation Act 1995</i></p> <p>Administered by the Department of Environment and Heritage</p> <p>PO Box A290 Sydney South, NSW 1232 Phone: (02) 9995 5000</p> <p>http://www.legislation.nsw.gov.au/xref/inforce/?xref=Type%3Dact%20AND%20Year%3D1995%20AND%20no%3D101&nohits=y</p>	

Northern Territory	
<p><i>Animal Welfare Act 1999</i></p> <p>Administered by the Department of Primary Industry and Fisheries Animal Welfare Unit</p> <p>GPO Box 3000 Darwin, NT 0801 Phone: 1300 720 386</p> <p>Internet: http://notes.nt.gov.au/dcm/legislat/legislat.nsf/linkreference/Animal%20Welfare%20Act</p>	<p><i>Territory Parks and Wildlife Conservation Act</i></p> <p>Administered by the Department of Land Resource Management</p> <p>PO Box 496 Palmerston, NT 0831 Phone: (08) 8999 5511</p> <p>Internet: http://lrm.nt.gov.au/plants-and-animals/information-and-publications</p>
Queensland	
<p><i>Animal Care and Protection Act 2001</i></p> <p>Administered by Animal Welfare and Ethics, Department of Agriculture, Fisheries and Forestry</p> <p>GPO Box 46 Brisbane, Qld 4001 Phone: 13 25 23</p> <p>Internet: http://www.legislation.qld.gov.au/Acts_SLs/Acts_SL_A.htm</p>	<p><i>Nature Conservation Act 1992</i></p> <p>Administered by the Department of Environment and Heritage Protection</p> <p>GPO Box 2454, Brisbane, Qld 4001 Phone: 13 74 68</p> <p>Internet: http://www.legislation.qld.gov.au/LEGISLTN/CURRENT/N/NatureConA92.pdf</p>
South Australia	
<p><i>Animal Welfare Act 1985</i></p> <p>Administered by the Department of Environment, Water and Natural Resources</p> <p>GPO Box 1047 Adelaide, SA 5001 Phone: (08) 8124 4800</p> <p>Internet: http://www.legislation.sa.gov.au/LZ/C/A/Animal%20Welfare%20Act%201985.aspx</p>	<p><i>National Parks and Wildlife Act 1972</i></p> <p>Administered by the Department of Environment, Water and Natural Resources</p> <p>GPO Box 1047 Adelaide, SA 5001 Phone: (08) 8124 4800</p> <p>Internet: http://www.legislation.sa.gov.au/LZ/C/A/NATIONAL%20PARKS%20AND%20WILDLIFE%20ACT%201972.aspx</p>
Tasmania	
<p><i>Animal Welfare Act 1993</i></p> <p>Administered by the Department of Primary Industries, Parks, Water and Environment</p> <p>PO Box 44 Hobart, Tas. 7001 Phone: 1300 368 550</p> <p>Internet: http://www.dpiw.tas.gov.au/inter.nsf/WebPages/EGIL-535VF?open#Thelaw</p>	<p><i>Nature Conservation Act 2002</i></p> <p>Administered by the Department of Primary Industries, Parks, Water and Environment</p> <p>PO Box 44 Hobart, Tas. 7001 Phone: 1300 368 550</p> <p>Internet: http://www.thelaw.tas.gov.au/tocview/index.w3p;cond=;doc_id=63++2002+AT%40EN+20040816110000;histon=;prompt=;rec=;term</p>

Victoria	
<p><i>Prevention of Cruelty to Animals Act 1986 Part 3</i></p> <p>Administered by the Department of Environment and Primary Industries</p> <p>PO Box 500 Melbourne, Vic.8002 Phone: 136 186</p> <p>Internet: http://www.austlii.edu.au/au/legis/vic/consol_act/poctaa1986360/</p>	<p><i>Wildlife Act 1975</i></p> <p>Administered by the Department of Environment and Primary Industries</p> <p>PO Box 500 Melbourne, Vic. 8002 Phone: 136 186</p> <p>Internet: http://www.austlii.edu.au/au/legis/vic/consol_act/wa197593/</p>
Western Australia	
<p><i>Animal Welfare Act 2002</i></p> <p>Administered by the Department of Agriculture and Food</p> <p>Locked Bag 4 Perth, WA 6983 Phone: (08) 9368 3333</p> <p>Internet: http://www.slp.wa.gov.au/legislation/statutes.nsf/main_mrttitle_50_homepage.html</p>	<p><i>Wildlife Conservation Act 1950</i></p> <p>Administered by the Department of Parks and Wildlife</p> <p>Locked Bag 104 Bentley Delivery Centre, WA 6983 Phone: (08) 9219 9000</p> <p>Internet: http://www.slp.wa.gov.au/legislation/statutes.nsf/main_mrttitle_1080_homepage.html</p>

Appendix VIII: Guidelines and policies

As state and territory governments have the responsibility for the protection of animal welfare, the guidelines and regulations listed below are intended to assist the researcher and are not an exhaustive list. A full list of state and territory specific guidelines and regulations may be found by contacting the relevant government departments (see Appendix IX). Please also be aware that subsequent revisions may be available.

<p>National Health and Medical Research Council</p> <p><i>Australian code for the care and use of animals for scientific purposes 8th edition (2013)</i></p> <p>http://www.nhmrc.gov.au/guidelines/publications/ea28</p>	<p>National Health and Medical Research Council</p> <p><i>Guidelines to promote the wellbeing of animals used for scientific purposes: The assessment and alleviation of pain and distress in research animals (2008)</i></p> <p>http://www.nhmrc.gov.au/publications/synopses/ea18syn.htm</p>
<p>Marine mammals</p> <p><i>Guidelines for the treatment of marine mammals in field research</i></p> <p>Published by the Society for Marine Mammalogy</p> <p>http://marinemammalogy.org/index.php?option=com_content&view=article&id=156&Itemid=182</p>	<p>Wildlife research</p> <p><i>National principles and guidelines for the ethical conduct of research in protected and environmentally sensitive areas 1998</i></p> <p>Published by the Australian Science, Technology and Engineering Council</p> <p>http://trove.nla.gov.au/work/8426292</p>
<p>Wildlife research</p> <p><i>Guideline on captive wildlife (2003)</i></p> <p>Published by the NSW Animal Research Review Panel</p> <p>http://www.animaethics.org.au/policies-and-guidelines/wildlife-research/captive-wildlife</p>	<p>Wildlife research</p> <p><i>Guideline on wildlife surveys (2004)</i></p> <p>Published by the NSW Animal Research Review Panel</p> <p>http://www.animaethics.org.au/policies-and-guidelines/wildlife-research/wildlife-surveys</p>
<p>Wildlife research</p> <p><i>Guidelines for the use of native animals in teaching and research</i></p> <p>Published by the University of Queensland</p> <p>http://www.uq.edu.au/research/integrity-compliance/wildlife-information</p>	<p>Wildlife research</p> <p><i>CCAC guidelines on: the care and use of wildlife (2003)</i></p> <p>Published by the Canadian Council on Animal Care</p> <p>http://www.ccac.ca/Documents/Standards/Guidelines/Wildlife.pdf</p>
<p>Administration of substances</p> <p>'Refining procedures for the administration of substances'</p> <p>Report of the BVA/AVMA/FRAME/ RSPCA/UFAW Joint Working Group on Refinement, <i>Laboratory Animals</i>, 2001, vol. 35, pp. 1–41</p> <p>http://lan.sagepub.com/content/35/1/1.full.pdf</p>	<p>Blood collection</p> <p><i>Guidelines on blood collection (2003)</i></p> <p>Published by the NSW Animal Research Review Panel</p> <p>http://www.animaethics.org.au/policies-and-guidelines/research-procedures/blood-collection</p>
<p>Assessment of pain and distress</p> <p>'The assessment and control of severity of scientific procedures on laboratory animals'</p> <p>Report of the Laboratory Animal Science Association Working Party, <i>Laboratory Animals</i>, 1990, vol. 24, pp. 97–130.</p> <p>http://lan.sagepub.com/content/24/2/97.full.pdf</p>	<p>Humane killing</p> <p><i>CCAC guidelines on: choosing appropriate end-points in experiments using animals for research, teaching and testing (1998)</i></p> <p>Published by the Canadian Council on Animal Care</p> <p>http://www.ccac.ca/en/CCAC_Programs/Guidelines_Policies/GDLINES/Guidelis.htm</p>

<p>Humane killing</p> <p><i>Euthanasia of animals used for scientific purposes</i> 2001</p> <p>Published by the Australian and New Zealand Council for the Care of Animals in Research and Teaching</p> <p>http://www.adelaide.edu.au/ANZCCART/publications</p>	<p>Humane killing</p> <p>'Recommendations for euthanasia of experimental animals: part 2'</p> <p><i>Laboratory Animals</i>, 1997, vol. 31, pp. 1–32</p> <p>http://lan.sagepub.com/content/31/1/1.full.pdf</p>
<p>Humane killing</p> <p><i>AVMA Guidelines for euthanasia of animals</i> (2013)</p> <p>Published by the American Veterinary Medical Association</p> <p>http://www.avma.org/issues/animal_welfare/euthanasia.pdf</p>	<p>Handling and transport</p> <p><i>Guidelines of the American Society of Mammalogists for the use of wild mammals in research</i> 2011</p> <p>Published by the American Society of Mammalogists</p> <p>http://www.mammalsociety.org/articles/guidelines-american-society-mammalogists-use-wild-mammals-research-0</p>
<p>Handling and transport</p> <p>Global standard for the transportation of live animals by air</p> <p>The International Air Transport Association (IATA) live animals Regulations</p> <p>http://www.iata.org/publications/Pages/live-animals.aspx</p>	<p>Housing and husbandry</p> <p><i>Australian mammals: biology and captive management</i>, 2nd Edition 2007</p> <p>Stephen Jackson</p> <p>Published by CSIRO Publishing</p> <p>http://www.publish.csiro.au/nid/18/pid/3619.htm</p>
<p>Animals in teaching</p> <p><i>Animals in schools: animal welfare guidelines for teachers</i></p> <p>Schools Animal Care and Ethics Committee</p> <p>Jointly published by the NSW Department of Education and Training, Catholic Education Commission and the Association of Independent Schools of NSW</p> <p>http://www.schools.nsw.edu.au/animalsinschools/resources/guidelines/index.htm</p>	<p>Animals in teaching</p> <p><i>Use of animals in post-graduate surgical training</i> (2003)</p> <p>Published by the NSW Animal Research Review Panel</p> <p>http://www.animaethics.org.au/policies-and-guidelines/animals-in-teaching/surgical-training</p>

Appendix IX: Relevant government departments

Commonwealth	
<p>Health and Research Ethics Research Translation Group National Health and Medical Research Council</p> <p>16 Marcus Clarke Street Canberra, ACT 2601</p> <p>Internet: http://www.nhmrc.gov.au</p>	<p>Threatened Species Scientific Committee Department of the Environment</p> <p>John Gorton Building, King Edward Terrace Parkes, ACT 2600</p> <p>Internet: http://www.environment.gov.au/topics/biodiversity/threatened-species-ecological-communities/threatened-species-scientific</p>
<p>Australian Marine Mammal Centre Department of the Environment Australian Antarctic Division</p> <p>203 Channel Highway Kingston, Tas. 7050</p> <p>Phone: (03) 6232 3209 Internet: http://www.marinemammals.gov.au</p>	
Australian Capital Territory	
<p>Territory and Municipal Services</p> <p>GPO Box 158 Canberra, ACT 2601</p> <p>Phone: 13 22 81 Internet: http://www.tams.act.gov.au</p>	<p>Environment and Sustainable Development Directorate</p> <p>GPO Box 158 Canberra, ACT 2601</p> <p>Phone: 13 22 81 Internet: http://www.environment.act.gov.au</p>
New South Wales	
<p>Animal Research Review Panel Department of Primary Industries Animal Welfare Inspectorial Office</p> <p>PO Box 100 Beecroft, NSW 2119</p> <p>Internet: http://www.animaethics.org.au</p>	<p>Office of Environment and Heritage</p> <p>PO Box A290 Sydney South, NSW 1232</p> <p>Phone: (02) 9995 5000 Internet: http://www.environment.nsw.gov.au</p>
Northern Territory	
<p>Department of Primary Industry and Fisheries Animal Welfare Unit</p> <p>PO Box 3000, Darwin NT 0801</p> <p>Phone: 1300 720 386 Internet: http://www.nt.gov.au/d/Primary_Industry/</p>	<p>Department of Land Resource Management</p> <p>PO Box 496 Palmerston, NT 0831</p> <p>Phone: (08) 8999 5511 Internet: http://www.lrm.nt.gov.au/</p>

Queensland	
Animal Welfare Unit Department of Agriculture, Fisheries and Forestry GPO Box 46 Brisbane, QLD 4001 Phone: 13 25 23 Internet: http://www.daff.qld.gov.au/animal-industries/welfare-and-ethics/animal-welfare/about-the-unit	Department of Environment and Heritage Protection PO Box 2454 Brisbane, QLD 4001 Phone: 13 74 68 Internet: http://www.ehp.qld.gov.au/
South Australia	
Department of Environment, Water and Natural Resources PO Box 1047 Adelaide, SA 5001 Phone: (08) 8124 4800 Internet: http://www.environment.sa.gov.au	
Tasmania	
Department of Primary Industries, Parks, Water and Environment GPO Box 44 Hobart, Tasmania 7001 Phone: 1300 368 550 Internet: http://www.dpiw.tas.gov.au	
Victoria	
Department of Environment and Primary Industries PO Box 500 Melbourne, Vic. 8002 Phone: 136 186 Internet: http://www.depi.vic.gov.au/	
Western Australia	
Department of Agriculture and Food Locked Bag 4 Perth, WA 6983 Phone: (08) 9368 3333 Internet: http://www.agric.wa.gov.au/	Department of Parks and Wildlife Locked Bag 104 Bentley Delivery Centre, WA 6983 Phone: (08) 9219 9000 Internet: http://www.dpaw.wa.gov.au/

Appendix X: Useful organisations and institutions

Australian	
<p>Australian and New Zealand Council for the Care and Use of Animals in Research and Teaching (ANZCCART)</p> <p>c/o The University of Adelaide, SA 5005 Phone: (08) 8303 7587</p> <p>Internet: http://www.adelaide.edu.au/ANZCCART</p>	<p>Australian Mammal Society</p> <p>Internet: http://www.australianmammals.org.au</p>
<p>The Australasian Wildlife Management Society (AWMS)</p> <p>http://www.awms.org.au</p>	<p>RSCPA National Office</p> <p>PO Box 265 Deakin West, ACT 2600</p> <p>Phone: (02) 6282 8300 Internet: http://www.rspca.org.au</p>
<p>Zoo and Aquarium Association</p> <p>PO Box 20 Mosman, NSW 2088</p> <p>Phone: (02) 9978 4797 Email: admin@zooaquarium.org.au Internet: http://www.zooaquarium.org.au/</p>	
International	
<p>Association for Assessment and Accreditation of Laboratory Animal Care International</p> <p>Suite 203 5283 Corporate Drive Frederick, Maryland 21703 USA</p> <p>Email: accredit@aaalac.org Internet: http://www.aaalac.org</p>	<p>Animals in Science Committee</p> <p>3rd floor, Seacole SW Quarter 2 Marsham Street, London SW1P 2AW, UK</p> <p>Email: asc.secretariat@homeoffice.gsi.gov.uk Internet: https://www.gov.uk/government/organisations/animals-in-science-committee</p>
<p>Animal Welfare Information Centre (AWIC)</p> <p>US Department of Agriculture National Agriculture Library</p> <p>10301 Baltimore Avenue Beltsville, Maryland 20705 USA</p> <p>Internet: http://awic.nal.usda.gov/</p>	<p>Canadian Council on Animal Care (CCAC)</p> <p>130 Albert Street Ottawa Ontario, K1P 5G4 Canada</p> <p>Email: ccac@ccac.ca Internet: http://www.ccac.ca</p>

Appendix XI: Experts not listed under a particular species

<p>Dr Wayne Boardman School of Animal and Veterinary Science University of Adelaide Phone: (08) 8313 1246 Email: wayne.boardman@adelaide.edu.au</p>	<p>Dr Rosie Booth Principal Conservation Officer David Fleay Wildlife Park Phone: (07) 5576 2411 Email: fleays@derm.qld.gov.au</p>
<p>Dr Peter Holz Veterinarian Tidbinbilla Nature Reserve Email: holz@megalink.com.au</p>	<p>RLC Feach Moyle Principal Ecologist Kleinfelder Phone: 040 040 1918 Email: fmoyle@kleinfelder.com Website: http://www.kleinfelder.com/australia/</p>
<p>Dr Larry Vogelnest Senior Veterinarian Taronga Wildlife Hospital Taronga Conservation Society Australia Email: lvogelnest@zoo.nsw.gov.au</p>	

Appendix XII: Additional references

- Blyde, D & Vogelneust, L 2008, 'Cetaceans', in Vogelneust, L & R Woods (eds.), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 591–614.
- Collins, LR 1973, *Monotremes and marsupials. A reference for zoological institutions*, Smithsonian Institution Press, Washington.
- Corbett, L 1995, *The dingo in Australia and Asia*, UNSW Press, Sydney.
- Corbett, L 2008, 'Dingo', in S Van Dyck & R Strahan (eds), *The mammals of Australia*, 3rd edn, Reed New Holland, Sydney, pp. 737–39.
- Flannery, T 1995, *Mammals of New Guinea*, Australian Museum/Reed Books, Sydney.
- Gemmell, RT 1986, 'Sexual maturity in the female bandicoot *Isoodon macrourus* (Gould, 1842) in captivity', *Australian Journal of Zoology*, vol. 34, pp. 199–204.
- Gemmell, RT 1987, 'Sexual maturity in the captive male bandicoot *Isoodon macrourus*', *Australian Journal of Zoology*, vol. 35, pp. 433–41.
- Holz, P 2007, 'Marsupials', in G West, D Heard & N Caulkett (eds), *Zoo animal and wildlife immobilization and anesthesia*, Blackwell Publishing, Ames, pp. 341–46.
- Holz, P 2007, 'Monotremes', in G West, D Heard & N Caulkett (eds), *Zoo animal and wildlife immobilization and anesthesia*, Blackwell Publishing, Ames, pp. 337–39.
- Lyne, AG 1964, 'Observations on the breeding and growth of the marsupial *Perameles nasuta* with notes on other bandicoots', *Australian Journal of Zoology*, vol. 12, pp. 322–39.
- McCracken, HE 1986, 'Observations on the oestrus cycle and gestation period of the greater bilby, *Macrotis lagotis* (Reid) (Marsupialia: Thylacomyidae)', *Australian Mammalogy*, vol. 9, pp. 9–16.
- McCracken, HE 1990, 'Reproduction in the greater bilby, *Macrotis lagotis* (Reid) – a comparison with other perameloids', in JH Seebeck, PR Brown, RL Wallis & CM Kemper (eds), *Bandicoots and bilbies*, Surrey Beatty and Sons, Sydney, pp. 199–204.
- Menkhorst, PW (ed.) 1995, *Mammals of Victoria*, Oxford University Press, Melbourne.
- Sikes, RS, Gannon, WL and the Animal Care and Use Committee of the American Society of Mammalogists 2011, 'Guidelines of the American Society of Mammalogists for the use of wild mammals in research', *Journal of Mammalogy*, vol. 92, pp. 235–53.
- Southgate, RI 1990, 'Habitats and diet of the greater bilby *Macrotis lagotis* Reid (Marsupialia: Peramelidae)', in JH Seebeck, PR Brown, RL Wallis & CM Kemper (eds), *Bandicoots and bilbies*. Surrey Beatty and Sons, Sydney, pp. 303–9.
- Southgate, R & S Carthew 2006, 'Diet of the bilby (*Macrotis lagotis*) in relation to substrate, fire and rainfall characteristics in the Tanami Desert', *Wildlife Research*, vol. 33, pp. 507–19.
- Tyndale-Biscoe, CH 2005, *Life of marsupials*, CSIRO Publishing, Melbourne.
- Woods, R, Ladds, P & Blyde, D 2008, 'Dugongs', in L Vogelneust & R Woods (eds), *Medicine of Australian mammals*, CSIRO Publishing, Melbourne, pp. 615–27.

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