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Charles Darwin University Animal Ethics Committee

Standard Operating Procedure:

GSOP 01.2020 Standard Operating Procedure: Addressing Animal Welfare in Fauna Surveys

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CHARLES DARWIN UNIVESITY ANIMAL ETHICS COMMITTEE

GSOP 01.2020: STANDARD OPERATING PROCEDURE: ADDRESSING ANIMAL WELFARE IN FAUNA SURVEYS

This Standard Operating Procedure (SOP) describes methods to maximise animal welfare when conducting fauna surveys using techniques commonly used in the Northern Territory. It covers the collection of specimens, the holding of animals in the field and the use of live capture mammal traps, pitfall traps and nets for trapping bats. The welfare of the animals should be the primary consideration at all times during a fauna survey.

Logistics

The main logistical consideration relating to animal welfare is the need to clear traps regularly and especially in the early morning before the animals become heat-stressed. In practice, this means ensuring that traps are checked within 2 hours of sunrise, but it depends upon the weather. The main determinants of the time it takes to clear traps are the number and spacing of the traps, the number of personnel and the efficiency with which they work. These factors must be appropriately balanced to ensure the welfare of trapped animals is not compromised.

Live Mammal Traps

Live mammal traps include the enclosed Elliot traps and open wire Tommahawk traps. The welfare issues relate to the siting of the traps and clearing them early in the morning.

- In hot conditions traps should be shaded (e.g. with vegetation) and closed during the day. In cold conditions they should be insulated and bedding material provided; and in freezing conditions, closed.
- Avoid ant colonies when siting traps. If ants are present move the trap and the flagging tape 5-10 metres away.
- If ants are present, remove bait from traps in the morning and re-bait as late as possible before dark.
- Clear the traps early in the morning (as above).
- Supply enough bait to give the animals a good feed.
- If females are trapped with their young, transfer them all to an open calico bag and leave the bag on the ground. This minimises the chance that the mother will leave the young behind.

Pitfall Traps

These are buckets dug into the ground, usually with a small fence to direct animals into the pit. Pitfall traps can become hot, or flooded, or overrun with ants.

- Check the trap several times per day.
- Avoid ant colonies when siting traps. Traps at risk of ants entering should be checked more often throughout the day and closed or moved if ants start entering in numbers.
- Place shelter in the trap in the form of soil, leaf litter or large leaves or bark.
- Ensure that there is moisture in the trap, to reduce heat-stress. This can be done using a wet cloth or small lid of water under leaf litter.

- Ensure that the trap does not flood and drown the animals. If rainfall is the main concern, puncture the bottom of the trap to allow the water to drain. If groundwater is the problem, use a watertight bucket or ensure that there is a flotation device in the bucket that animals can use to climb above the water (e.g. floating wood, block of styrofoam).
- In hot conditions shade the trap, for example using a bucket lid propped above the trap.

Camera Traps

The use of remote cameras is increasing as a less invasive method of detecting animals. It is particularly suited to nocturnal, medium to large sized mammals, including wallabies, dingoes and cats.

- Infra-red cameras will not emit a flash, however animal identification from the white-light (flash emitting) cameras is easier and more reliable.
- Be aware of how the bait used will affect non target species.
- Be aware that many animals may damage or remove the bait so it must be anchored securely. Dingoes, large rats, crows, cattle and buffalo are particularly efficient at removing or destroying bait stations.

Bat Traps

These are mist nets and harp traps for bats.

- Harp traps may be left unattended at night and checked early in the morning. Captured bats will roost within the trap. They are easily removed.
- Harp traps used over a number of nights in the same location may attract predators such as Kookaburras or goannas so be aware of this risk.
- Mist nets must be attended constantly, and bats removed as they are captured. They can become injured by the net, or during extraction from the net, which can be difficult.
- If a bat in a mist net seems stressed or has been tangled for a long time, cut it out of the net. It is better to damage and repair a net than to risk an animal death.
- Birds may also be captured when mist netting for bats at night, release and record these captures for subsequent reporting to the Animal Ethics Committee (AEC).

Spotlighting

Spotlighting is used to identify larger nocturnal mammals and birds and to search for nocturnal reptiles and frogs that may not be caught in traps. It can be done on foot or from a vehicle. Most animals seen while spotlighting do not need to be caught but this does not mean that the technique does not cause disturbance.

- Avoid using high powered spotlights; 50W or less is often sufficient.
- Especially with higher powered lights, do not hold the light on the animal for longer than is necessary for identification. If you need to spotlight an animal for longer, use the edge of the beam, not the centre.
- The use of binoculars while spotlighting is difficult but it is a good skill to develop.

Genetic Samples

Genetic studies now contribute to many taxonomic and population biology studies. A small piece of tissue should be taken from a representative sample (generally 5) of the individuals in an area. This is typically done by collecting a small piece of ear tissue (mammals), a tail tip (reptiles) or toe tip (frogs).

- Sit in a quiet shaded place.
- Photograph animal and take measurements.
- Record details on data sheet.
- Use a sharp pair of surgical scissors or a lab-mouse ear notcher. Before and after use, clean the instrument with an alcohol swab to protect animals and samples from cross contamination.
- Secure the animal in a calico bag then expose only the ear (larger animals) or secure by hand (small reptiles and frogs).
- Use the scissors to remove a piece of ear tissue, tail tip or toe clip as per Table 1.
- Place the tissue in small tube of RNALater or 70% Ethanol and place a unique label (preprinted or written in pencil) inside the tube. An identifying number on the top of the tube is also helpful when taking multiple samples.
- Control any bleeding by using gentle pressure, then release the animal at the point of capture.
- If an animal becomes distressed, take action to reduce the distress. This may require abandoning the sampling and or immediate release at the capture site.

Table 1: Tissue samples required from vertebrates for genetics.

Animal Group	**Sample Required**
Small mammals	Ear clip (3-4 mm^2, less than 2 x 2 mm)
Limbed lizards	Tail clip (1-2 cm, BUT adjust to size of animal)
Snakes/legless lizards	Tail clip, or 1-2 ventral scale clips
Frogs	Toe clip at 1st or 2nd joint of long toe on hind foot or from finger (2-4 mm, not from thumbs)
Blind snakes	Voucher only

Specimens

The deliberate killing of wild animals for scientific collection remains a contentious issue. In places such as the Northern Territory, where many groups have been poorly inventoried or remain taxonomically unresolved, some collections may be required to confirm species' identity or to provide the basis for the description of new taxa. Animals that are found dead in traps or as road kill and are still intact (not eaten by ants, desiccated or decomposing) should also be vouchered. The following guidelines for collection of specimens should be followed.

- Animals should be killed using appropriate euthanasia techniques. Reilly (2001) provides a detailed description of recommended and acceptable procedures, a summary of which is included in the AEC Guidelines for completing the animal ethics application for research and teaching (including recommended euthanasia techniques). Note that some of these techniques require considerable experience to be used appropriately.
- Maximum use should be made from collected specimens, to minimise the number of specimens required. All specimens should be properly preserved and later deposited in museums.

- Record measurements and wherever possible tissue samples should be taken just after death and stored appropriately for possible subsequent genetic analysis. This needs to be preserved before formalin fixing the voucher specimen. Alternatively, if you open the abdominal cavity to enhance fixation (see below) a small piece of the liver can be preserved for genetic studies as it contains significantly more DNA than ear, tail or toe samples
- Voucher specimens need to be preserved properly to prevent the loss of the specimen, and subsequent collection of the another voucher specimen.
- Impacts upon the local population should be considered – the number of specimens taken should not have a significant impact upon the viability of the remaining population.
- Collection of specimens should be undertaken only when non-destructive techniques (e.g. blood sampling, hair analysis for specific identity) are inapplicable or impractical.

Holding Captured Animals

If an animal cannot be identified at the trap site, it may need to be held for some time.

- Hold the animal in an appropriate container. For small mammals and larger reptiles, calico bags are suitable, for small reptiles and frogs a clear sealed plastic bag is best. Ensure frogs have a little water and green leaves to stay hydrated.
- Minimise the time the animal is held by ensuring that identification or euthanasia takes place as soon as possible.
- Be methodical in processing and releasing animals so that none are overlooked.
- Keep the animals in a place where extreme temperatures are avoided, they will not be trodden on, predators cannot access them, and they cannot harm each other.
- If an animal becomes stressed, take action to reduce the stress. This may require cooling with water, or immediate return to the capture site.
- Return animals to the capture site, to a spot where they will be not be unduly exposed to the sun, cold or predation risk. For nocturnal animals release at dusk or dawn.

Signs of Stress

It is not always possible to tell if an animal is stressed, and therefore at risk of death. Some clinical signs that they may show are:

- Obvious increase in heart rate or breathing rate
- Animal is limp or closes its eyes (mammals)
- Animal may feel hot to the touch
- Eyes open, rigid with fright
- Panting, heat stress

In the Event of a Death or Injury

- Record the process of events leading to any animal death (whether intended or accidental).
- Record what actions were taken to reduce further deaths at the time and for future surveys. This is important information for improving on techniques and for reporting to the AEC.
- All sick, injured and if necessary orphaned wildlife are to be transported to the nearest veterinarian for initial assessment and treatment. Then these animals can be transferred to an official wildlife carer (contact Wildcare Inc. NT on 08 8988 6121, 0408 885 341 or 0412 910 975).

References

Reilly J. (ed) (2001) Euthanasia of animals used for scientific purposes. ANZCCART, Glen Osmond, South Australia – Currently being updated

Relevant CDU AEC Guidelines

Guide to Completing the CDU AEC Project and Permit Application

Other Sources

NHMRC (2013) Australian code for the care and use of animals for scientific purposes, 8th Edition

NHMRC (2014) A Guide to the care and use of Australian native mammals in research and teaching

NHMRC (2008) Guidelines to promote the wellbeing of animals used for scientific purposes: The assessment and alleviation of pain and distress in research animals