DOC best practice manual of conservation techniques for bats

Version 1.0

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Inventory and monitoring toolbox: bats

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Department of Conservation
Te Papa Atawhai

Introduction

Three species of bats are known to occur in New Zealand, representing eight different taxonomic units. All are threatened, and subject to a recovery programme (Molloy 1995; O'Donnell et al. 2010). There is now considerable research effort and active conservation programmes directed towards these taxa. New Zealand bats are challenging to work with because they are rare, cryptic and difficult to catch and study. Working with bats requires the use of many specialised techniques and skills. Additionally, there are currently relatively few people that have in-depth experience of working with bats in New Zealand. Therefore, the New Zealand Bat Recovery Group saw the need to produce a best practice manual that would outline appropriate research, inventory and monitoring, and management techniques and provide ethical standards that should be applied when working with bats. This introduction describes the main objectives of the best practice manual, how the plan was developed, accountabilities for maintaining best practice, and review procedures.

Objectives

The objectives of this manual are to:

- Provide information and resources on the best techniques currently available to manage and undertake research on bats
- Assist DOC staff, external managers and researchers to develop and improve techniques used for the recovery and management of bats
- Provide guidelines for safe, ethical and responsible practices when handling and studying bats
- Help formalise consistent use of best management practices across bat taxa throughout the country
- Provide a mechanism for advocating the continuous improvement of bat conservation management and research

Development of best practice

The practices described in this manual represent a mixture of widely used (globally) techniques plus others adapted or developed for working with New Zealand bats. Most have been trialled and used in New Zealand over the last 10–20 years with approval from appropriate Animal Ethics Committees. Techniques yet to be used in New Zealand are clearly identified in the text.

Accountabilities for identifying and maintaining best practices

The following accountabilities can only be assigned to DOC staff in the context of a best practice manual. However, people outside DOC who can contribute to improving best practice are encouraged to do so via the Bat Recovery Group Leader. Staff in the following positions are responsible for identifying and maintaining best practice techniques for working with all taxa of bats:

- Area Managers
- Rangers

- Programme Managers
- Bat Recovery Group Members
- Technical Advisors
- Science Advisors

Definition of best practice procedures (mandatory, recommended)

Mandatory procedures are those that must be followed because after years of practice they have been identified as the best and most reliable methods. These procedures are indicated by the words '**must**', '**shall**', or '**do not**'.

Recommended procedures are those which to our current knowledge are considered to be best methods or procedures, but may have reasonable alternatives. These procedures are indicated by the words '**should**' or '**may**'.

Review process

The Bat Recovery Group is charged with the responsibility of reviewing and revising this document **annually**, so ensuring that current best practices are promulgated within and outside of DOC.

In some sections, there may be alternative approaches suggested, but further experience may reveal that one method is superior to the alternatives, and may become the single mandatory best practice.

There are also various methods under development which have not yet been written or been agreed upon as best practice; when these methods have been developed the specifications will become available.

Changes from the stated best practice should be approved by the Bat Recovery Group and be carefully documented. Adherence to current best practice guidelines should not prevent innovations, which may result in improved performance, but it is important that such innovation be monitored and tested fully.

Current standard operating procedures and guidelines that commonly apply to animal pest operations are available on the DOC website.[1]

Additional related documents for DOC staff

DOC uses a range of other documents to set standards for research and management that should also be consulted when considering appropriate work programmes related to bats. These include a large number of standard operating procedures (SOPs) and guidelines that can be accessed by DOC staff on the DOC Intranet (Policies and Procedures Tab/DOC 'Knowledge' site). It is not intended to repeat these guidelines in this best practice manual. These documents are regularly revised and updated and new procedures are added to the sites. Examples include:

[1] http://www.doc.govt.nz/publications/science-and-technical/doc-procedures-and-sops/managing-animal-pests/standard-operating-procedures/

- Code of ethical conduct for the manipulation of live animals (olddm-766783)
- Captive management SOP (docdm-266180)
- Translocation SOP (docdm-1089378)
- Animal pests SOP definitions and FAQs (docdm-51708)
- Use of second generation anticoagulants on public conservation lands (docdm-97398)
- Preparing an Assessment of Environmental Effects (AEE) (docdm-95676)

References and further reading

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Introduction to New Zealand bats/pekapeka

Conservation status

Three bat species are known to occur in New Zealand: the long-tailed bat (*Chalinolobus tuberculatus*), the lesser short-tailed bat (*Mystacina tuberculata*), and the greater short-tailed bat (*M. robusta*). All are endemic. In addition, there have been cases of vagrants turning up, though they have mainly been accidental imports with freight (see 'Biosecurity incursions'). The long-tailed bat is a member of the large and widespread family Vespertilionidae, and is represented by seven species in Australasia (Hutson et al. 2001). Long-tailed bats are currently described as a single species, but significant differences in size and genetic diversity have been recorded throughout its geographical range (O'Donnell 2001a; Winnington 1999). The lesser short-tailed bat and greater short-tailed bat belong to the endemic and monogeneric family Mystacinidae. Lesser short-tailed bats are currently described as three sub-species, but significant differences in genetic diversity have been recorded throughout its geographical range (Table 1). Greater short-tailed bats are generally considered to be extinct because there has been no confirmed sighting since 1967 (Daniel 1990). However, unusual Mystacinid-like calls were recorded on Putauhinu Island in 1999, which led to speculation that greater short-tailed bats might still be extant (O'Donnell 1999).

New Zealand bats are protected by the Wildlife Act 1953, and all, except for greater short-tailed bats, are listed as 'Vulnerable' by International Union for Conservation of Nature (IUCN) criteria (greater short-tailed bats = 'Extinct', Hutson et al. 2001). DOC currently identifies six bat taxa of concern (Table 1; O'Donnell et al. 2010). None of these taxa, except the central North Island lesser short-tailed bat, can be considered secure on the mainland, and may face a high risk of extinction in the medium term if conservation management is not successful at reversing their declines (Molloy 1995; O'Donnell et al. 2010). The two single populations of lesser short-tailed bats on Codfish/Whenua Hou and Little Barrier/Hauturu islands are considered more secure.

Table 1. Conservation status of New Zealand bats (Townsend et al. 2008; O'Donnell et al. 2010).

Taxon	Conservation Management Unit* recognised by Bat Recovery Group, 2004† current	Scientific name	Conservation status
1. long-tailed bat (North Island)	1. long-tailed bat (North Island)	*Chalinolobus tuberculatus*	nationally vulnerable
2. long-tailed bat (South Island)	2. long-tailed bat (South Island)	*Chalinolobus tuberculatus*	nationally critical
3. greater short-tailed bat	3. greater short-tailed bat	*Mystacina robusta*	data deficient
4. northern lesser short-tailed bat	4. northern lesser short-tailed bat	*Mystacina tuberculata aupourica*	nationally vulnerable
5. central lesser short-tailed bat	5. eastern lesser short-tailed bat 6. north-western lesser short-tailed bat	*Mystacina t. rhyacobia*	declining
6. southern lesser short-tailed bat	7. southern North Island lesser short-tailed bat 8. South Island lesser short-tailed bat	*Mystacina t. tuberculata*	nationally endangered
7. little red flying fox		*Pteropus scapulatus*	vagrant

Notes:

* Conservation Management Units are defined as population units that are of interest to conservation managers. These may be populations or subpopulations that are worthy of protection because they are distinctive in some way or one of a number of subpopulations vital to maintaining long-term viability of a taxon. They may be distinctive genetically, behaviourally, morphologically, or geographically.

† Based on a revision endorsed by the Bat Recovery Group.

Distribution and populations

New Zealand long-tailed bat

Long-tailed bats are widely distributed from the north of the North Island, through the South Island, to Halfmoon Bay on Stewart Island but there are now significant gaps in this distribution. They are also present on Great Barrier/Aotea, Little Barrier/Hauturu and Kapiti islands (Dwyer 1960, 1962; Daniel 1970; Daniel & Williams 1981, 1984). However, historical anecdotes and monitoring since 1990 indicate that long-tailed bats are now rare or absent at many sites where formerly they were common (e.g. Westland, Nelson-Marlborough, eastern side South Island, Wellington; Barrie 1995; O'Donnell 2000a) and in the few places where intensive monitoring has occurred they are still declining (Pryde et al. 2005).

The size of few long-tailed bat populations is known. Banding studies in some of the larger populations suggest a minimum of 800 bats using Grand Canyon Cave near Te Kūiti (O'Donnell 2002a), 150–200 at Hanging Rock in South Canterbury (Lettink & Armstrong 2003) and > 300 in the Eglinton Valley, Fiordland (O'Donnell 2000b). Average numbers of 86 bats emerging from roosts in Pukeiti, Hawke's Bay (ra = 5–208; Gillingham 1996) and 14 bats in the Waitakere Ranges (ra = 2–24; Alexander 2001) are likely to be underestimates of total population size. Group sizes in plantation forests and urban settings are generally very small (< 10 bats; Dekrout 2009; Borkin & Parsons 2010).

Lesser short-tailed bat

Lesser short-tailed bats were also once widespread. Recent fossils have been found in Waikato, Hawke's Bay and Wairarapa, north-west Nelson, Canterbury and Fiordland. However, there are no current records from much of Northland, Coromandel, Rotorua, Pirongia, East Cape, western Tararua Forest Park, Nelson Lakes, Mt Richmond, Ōkārito, the Longwood Range and Catlins Forest Park—all areas where formerly they were present (Dwyer 1962; Daniel & Williams 1984; Daniel 1990). The species may persist in some of these areas, as none of them have been surveyed (B. Lloyd pers. comm.).

Recent surveys show that populations survive in several areas in the North Island, and at least two areas in the South Island (Lloyd 2005). In Northland, a small population remains in Omahuta and Puketi forests, and individuals have been recorded in Waipoua and Warawara forests. In the rest of the North Island, lesser short-tailed bats have been found from north Taranaki, across the central volcanic plateau towards East Cape and south to the Tararua Range). Significant populations have been found in Waitaanga, Pureora, Waitōtara, Rangataua, Kaimanawa, Whirinaki, and south-east Urewera forests. They occur occasionally in Kaimai-Mamaku, Raukūmara, Ruahine, and eastern Tararua forest parks, as well Waimarino and the western slopes of Mt Ruapehu (Lloyd 2005).

Only two populations of lesser short-tailed bats have been confirmed in the South Island, one in the Ōpārara Basin (north-west Nelson) and the other in the Eglinton Valley (Fiordland National Park). Calls suggestive of short-tailed bats have been recorded at Paparoa National Park and the Dart Valley but their identity has yet to be confirmed. None have been recorded during preliminary surveys of forests elsewhere on the West Coast and in parts of Fiordland National Park and north-west Stewart Island. There are large populations on Little Barrier/Hauturu and Codfish/Whenua Hou islands (Lloyd 2005).

The size of few lesser short-tailed bat populations is known. Numbers of bats in Rangataua Forest fluctuated between 5740 and 6977 in early summer (from 1995–1999). Minimum population estimates from Waitaanga were 2700 bats (B. Williams, pers. comm.); in Eglinton Valley the maximum emerging from a single roost has been > 1500 lesser short-tailed bats (C. O'Donnell, unpublished data); and on Codfish Island 1557 bats (Sedgeley & Anderson 2000). Estimates for the latter two populations are probably substantially lower than the actual population size because there were other roosts occupied in the areas that were not counted. These data and unpublished observations indicate the total population of lesser short-tailed bats in central North Island is currently less than 40 000, and the total New Zealand population is less than 50 000 (Lloyd 2001, 2005).

Greater short-tailed bats

Initially two subspecies of short-tailed bat were described (Dwyer 1962): a smaller one, *Mystacina tuberculata tuberculata*, found throughout much of New Zealand, and a larger one, *M. t. robusta*, restricted to the Muttonbird Islands, off the south-west coast of Stewart Island. Subsequently the subspecies were elevated to species status as the lesser short-tailed bat *M. tuberculata* and the greater short-tailed bat *M. robusta* (Hill & Daniel 1985). Recent fossil remains (i.e. < 20 000 years old) of greater short-tailed bats found in caves, on rock ledges and in swamps show that this species was once found in sites in Waitomo, Hawke's Bay, and Wairarapa in the North Island, and north-west Nelson, Westland, Canterbury, and Central Otago in the South Island. From 1840 until the early 1960s greater short-tailed bats were only found on the rat-free Big South Cape (Taukihepa, 930 ha) and Solomon Island (Rerewhakaupoko, 32 ha), in the Muttonbird Islands, 2–10 km off the south-west coast of Stewart Island.

Major threats to New Zealand bats

A wide range of threats to the continued viability of bat populations have been identified. The range and numbers of bats have declined significantly and in many areas declines are continuing (e.g. O'Donnell 2000a,c; Lloyd 2005; Pryde et al. 2005, 2006). Declines result from a combination of threats, namely predation and competition, habitat degradation, and disturbance.

Predation and competition

Introduced mustelids, rats, possums and cats all prey on, or attempt to prey on, New Zealand bats. Bats are vulnerable to predators throughout the year; in summer when they congregate in large colonies to give birth and rear young, and during winter when they may remain inactive (in torpor) for long periods within roosts.

Mustelids have been recorded killing bats elsewhere in the world (Mumford 1969; Stebbings & Placido 1975; Hill & Smith 1984). In New Zealand, stoats have been seen entering lesser short-tailed bat roosts, although whether they impact on population viability is unknown (Lloyd 2001). The disappearance of greater short-tailed bats from mainland New Zealand coincided with the spread of kiore rats (Worthy 1997), and this species subsequently became extinct during the 1960s when ship rats arrived on its last known refuge, Big South Cape Island off Stewart Island (Daniel 1990). Lloyd (2001) recorded ship rats attempting to enter lesser short-tailed bat roosts during winter, although none were successful at capturing bats. A significant decline in the survival of long-tailed bats in the Eglinton Valley was strongly correlated with irruptions of both stoats and ship rats (Fig. 1; Pryde et al. 2005).

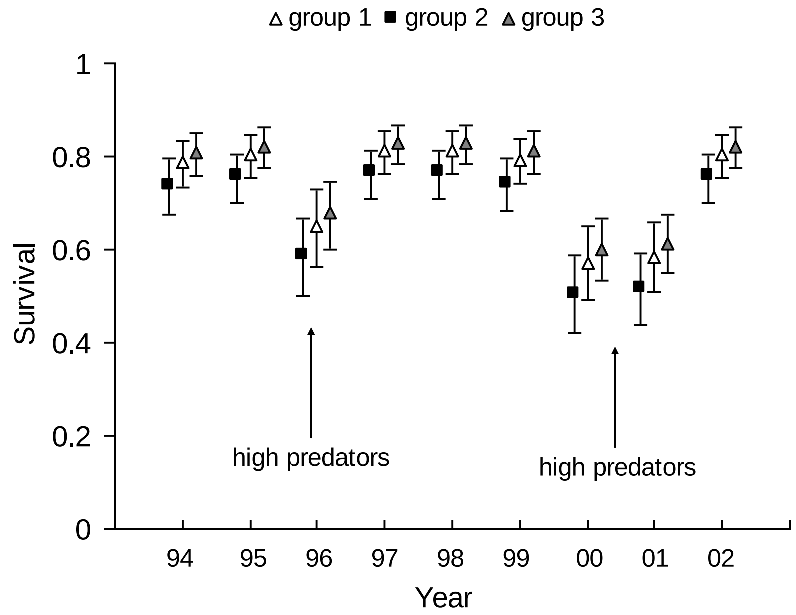

Figure 1. Adult female annual overwinter survival (±SE) of three social groups of long-tailed bats in the Eglinton Valley from 1993–2003. Survival was lower in years when there were high numbers of introduced predators (1996, 2000, 2001) (Pryde et al. 2005).

Feral cats appear to be common predators of bats (Dwyer 1962; Daniel & Williams 1984; O'Donnell 2000a). Cats accounted for 28% of reported long-tailed bat deaths and 26% of lesser short-tailed bat deaths (Daniel & Williams 1984), including juveniles (Daniel & Williams 1981). Similarly, cats accounted for 45% of injured *Chalinolobus* bats handed in for care in Victoria, Australia (Dowling et al. 1994). In South Canterbury, New Zealand, possums were recorded attempting to reach into cavities containing young bats on 50% of nights when roosts were monitored using video cameras (O'Donnell 2000c).

Morepork owls are native predators of bats (Stead 1936; Dwyer 1960; Daniel & Williams 1984), but Griffiths (1996) also observed introduced little owls attempting (unsuccessfully) to catch long-tailed bats near Geraldine, South Canterbury. Little owls have also been reported taking bats in Britain (Speakman 1991).

In the Eglinton Valley, Fiordland, introduced starlings and ship rats made nests in long-tailed bat roosts (Sedgeley & O'Donnell 1999a), although it is not known if these species actually preyed upon or displaced bats from roosting sites. Competition for roosting sites by exotic species may limit availability of roosts to bats (Griffiths 1996). Griffiths found that starlings, house sparrows, feral pigeons and introduced wasps all occupied cavities that appeared to be suitable as bat roosts in the limestone areas at Hanging Rock in South Canterbury. Competition between bats and starlings has been suggested in Europe (Mason et al. 1972; Rieger 1996).

There has been speculation that both exotic and native species may compete with lesser short-tailed bats for food sources. Many of the types of fruit eaten by lesser short-tailed bats are also eaten by many bird species, rodents and possums (Daniel 1976). Molloy (1995) suggested introduced wasps which feed on nectar, fruit and insects may compete with lesser short-tailed bats.

Ecroyd (1995) demonstrated using infrared video surveillance that ship rats and possums all fed on significant amounts of nectar from flowers of wood rose/pua o Te Reinga, *Dactylanthus taylorii*, making it unavailable to lesser short-tailed bats. Possums cause serious damage to native ecosystems by selective browsing; they also feed on buds, flowers and fruits of a wide variety of native trees and shrubs and have also caused mortality of native trees (Cowan & Waddington 1990). By doing this they are modifying bat habitat and possibly competing for some foods with lesser short-tailed bats.

Habitat degradation

Before humans arrived in New Zealand, indigenous forest covered 85% to 90% of the country (McGlone 1989), but it is now reduced to about 14% of its original area (Stevens et al. 1988). Dwyer (1960, 1962) concluded that the decrease in the area of distribution was correlated with the removal of indigenous forest during the last century and the failure of either bat species to survive in open country or urban areas. Disappearance of long-tailed bats from coastal and lowland regions in areas such as Canterbury, Otago and Southland coincided with the loss of forest cover (Hutton & Drummond 1904; Dwyer 1960; Barrie 1995). Early records note the disturbance of large colonies of long-tailed bats while European colonists were burning and felling trees for timber, and clearing land for agriculture or for mining (Buller 1892; Cheeseman 1893). The majority of bat deaths recorded by Daniel & Williams (1984) occurred when the bats' roost trees were cut down. There are still instances of bat roosts being felled for firewood and for timber in Nelson, Buller, and Canterbury in the South Island, and the King Country in the North Island (O'Donnell 2000a,c).

Today, habitat degradation usually relates to loss of old-age preferred roost trees in areas where bat colonies are found, or degradation in important foraging habitat (e.g. Sedgeley & O'Donnell 1999a,b, 2004; Sedgeley 2003). Major threats include: clearance of indigenous vegetation, selective logging of preferred old-aged trees on private land, conversion of indigenous shrublands to pine plantations, firewood cutting, over-grazing of forest remnants so that regeneration is inhibited, and road and quarry works around limestone cliff roosting areas. Long-tailed bats have been recorded in commercial plantation forests, but roost trees are regularly felled as part of normal logging operations. In addition, habitat that has recently been subject to extensive logging has also been converted to pasture (Borkin & Parsons 2010). A variety of river control works can affect primary feeding habitats that occur along waterways. These include water abstraction from foraging sites, construction of dams that may drown foraging sites, and changes in water quality that lead to reductions in invertebrate prey (O'Donnell 2000a,c).

Disturbance

Disturbance by rock climbers, particularly in winter when bats used the same limestone crevices used by climbers, was identified as a potential risk to long-tailed bats at Hanging Rock escarpment in South Canterbury (Griffiths 1996). Disturbance of cave-roosting bats is still a concern at Grand Canyon Cave (D. Smith, pers. comm.). Instances where all bats in this cave have taken flight while people have been watching them have been recorded in recent years (N. Miller, pers. comm.).

Basic ecology

Long-tailed bats

Foraging

Long-tailed bats are relatively small with body mass of adult bats ranging from 8.5 to 12.3 g, and forearm length from 38.7 to 40.5 mm (O'Donnell 2001a). Long-tailed bats have wing characteristics and echolocation calls typical of moderately fast-flying bats that forage along forest edges and gaps (O'Donnell 2000d, 2001a).

Long-tailed bats are solely insectivorous. Although a comprehensive study of their diet has yet to be conducted, existing data suggest they consume a wide variety of aerial aquatic and terrestrial invertebrates (Gillingham 1996; O'Donnell 2005).

Breeding

There is little information on the mating system. Mating most likely occurs in autumn, but the length of embryonic development and mechanism for delaying onset of gestation is unknown. Time of births vary geographically and occur between mid-November to mid-December when females congregate in maternity roost to give birth and raise young. They give birth to a single young once a year (O'Donnell 2001a, 2002b). They have a complex social system. In the Eglinton Valley the population is split among three behaviourally, though not geographically, isolated sub-groups that rarely mix. Bats belonging to each social group are spread over many roosts each day, and composition of bats within these roosts changed from day to day (O'Donnell 2000b).

Habitat use

Long-tailed bats are closely associated with indigenous forest, but have also been recorded in a variety of other habitats. These include logged forests, shrublands, plantation forests and farmland (Daniel & Williams 1984; Dekrout 2009; Borkin & Parsons 2010). They have very large home ranges. Colonies in the Eglinton Valley, Fiordland ranged over 11700 ha. Individual ranges varied depending on age, sex and time of the breeding season (averaging 237–2006 ha, max = 5629 ha; O'Donnell 2001b).

In modified landscapes and predominantly agricultural areas, long-tailed bats have been recorded roosting beneath bridges, in farm buildings, and in caves and crevices in limestone as well as a range of indigenous and exotic tree species (Daniel 1981; Daniel & Williams 1981, 1983, 1984; Sedgeley & O'Donnell 2004; Dekrout 2009). However, more recent radio-tracking studies showed that the majority of roosts used by long-tailed bats are in trees, and maternity roosts are almost exclusively in trees (Gillingham 1996; Griffiths 1996; Sedgeley & O'Donnell 1999a,b, 2004). Numbers of bats using a roost at one time averaged 14–86 bats, and they changed roost site almost every day. Long-tailed bats seldom use an individual roost more than once during a summer, but will reuse many of these roosts from year to year (O'Donnell & Sedgeley 1999).

Roosting

In detailed studies of roosting behaviour in Fiordland and South Canterbury, breeding groups of long-tailed bats selected specific roost trees and roost cavities that were very distinctive from the pool of trees potentially available to them. They roosted inside cavities in main trunks or limbs in some of the largest and oldest trees available. Cavities were high from the ground (usually > 15 m), generally had one entrance, internal dimensions were of a small to medium size (compared to lesser short-tailed bats), and were often formed inside a knot-hole. The cavities provided protection from wind and rain, were dry inside, had relatively thick cavity walls, and internal humidity and temperature were stable compared to ambient (Sedgeley & O'Donnell 1999a,b; Sedgeley 2001).

Figure 2. Long-tailed bat in the hand (photo: C.F.J. O'Donnell).

Lesser short-tailed bats

Foraging

Lesser short-tailed bats are slightly larger than long-tailed bats with body mass of adults ranging from 11.4 to 22.0 g, and length of forearm from 36.9 to 46.9 mm (O'Donnell et al. 1999; Lloyd 2005) (Fig. 3). Lesser short-tailed bats are agile on the ground and have evolved unique morphological adaptations for terrestrial foraging such as the ability to tightly fold their wings within thickened wing membranes, strong hind legs and feet, and small spurs at the base of their claws (Dwyer 1962; Daniel 1990; Jones et al. 2003). Wing shapes in lesser short-tailed bats vary. Data collected in the Eglinton Valley demonstrated that they had wings that made them more manoeuvrable than long-tailed bats within vegetation, which is typical of species known to forage by gleaning insects (Jones et al. 2003). However, lesser short-tailed bats on Whenua Hou did not seem to specialise in any particular flight strategy (Webb et al. 1999. Their echolocation calls are typical of bats that glean from surfaces, and recent evidence suggests that while they hunt by echolocation when flying, they use a combination of prey-generated sound and smell to locate food while on the ground (Parsons

2001; Jones et al. 2003). Lesser short-tailed bats are omnivorous. Their diet consists of flying and non-flying invertebrates, nectar, pollen, plant material and fruit. Adaptations for nectar feeding include an extensile papillated tongue, and a wide gap between the incisors (reviewed in Arkins et al. 1999).

Figure 3. Lesser short-tailed bat in the hand (photo: C.F.J. O'Donnell).

Breeding

Lesser short-tailed bats have a complex mating system that is not fully understood. Male bats occupy singing or mating roosts, and call from them at night to attract females (Daniel 1990). In island populations, singing trees are clustered, and males are thought to compete for possession of them. This behaviour has been described as a lek breeding system (Daniel 1990), but requires further study. Singing begins in spring and early summer but peaks during autumn. Mating takes place in late summer and autumn, and births occur between mid-December and mid-January (Lloyd 2001).

Habitat use

Lesser short-tailed bats have been recorded roosting in a variety of habitats, but only in relatively low numbers outside of unmodified forest (Daniel & Williams 1984). There are no contemporary records of them roosting in caves, but there are historical records that show caves were once used. There are large concentrations of bat remains in limestone caves (Worthy & Holdaway 1994, 2001) and both species of short-tailed bat roosted in granite sea-caves on islands to the south-west of Stewart Island (Stead 1936; Daniel & Williams 1984). Several records for lesser short-tailed bats have been reported from buildings. However, all were from mountain huts or homes adjacent to large areas of indigenous forest (Daniel & Williams 1984). Lesser short-tailed bats have very large home ranges, and large populations have only been found in extensive (1000 ha) areas of largely unmodified old-growth forest (Lloyd 2001). Colonies in the Eglinton Valley ranged over 14710 ha. Individual ranges varied depending on age, sex and time of the breeding season (ranging from 127–6223 ha; Christie 2003).

Roosting

Three types of roost tree have been described for lesser short-tailed bat: those used by breeding groups and large numbers of bats; those used by solitarily roosting bats; and those used for mating/singing (Daniel 1990; Sedgeley 2003, 2006). In the Eglinton Valley and on Codfish Island/Whenua Hou roost trees used by solitary bats and by singing/mating bats had much smaller stem diameters and internal cavity dimensions than those used by large groups of bats (O'Donnell et al. 1999; Sedgeley 2003, 2006). Roost cavities used by communally roosting lesser short-tailed bats are variable. In Rangatau Forest, central North Island, and on Codfish Island/Whenua Hou, roost entrances are typically on the main trunk and are less than 0.3–7 m above the ground whereas in the Eglinton Valley they can be up to 23 m above ground (Lloyd 2005; Sedgeley 2003). Size of entrances vary greatly, from small holes only just large enough for bats to enter, to enormous splits several metres long (Lloyd 2005; Sedgeley 2003, 2006). All roosts selected by lesser short-tailed bat in the Eglinton Valley are in dry, well-insulated cavities inside some of the largest and oldest trees available (Sedgeley 2003). Lesser short-tailed bats move between a relatively small pool of roost trees (e.g. 20–30 trees in < 150 ha of forest) and use no more than 2 or 3 communal roosts any one time (Lloyd 2001; Christie 2003).

In the Eglinton Valley, Fiordland, several features distinguish lesser short-tailed bat roosts from long-tailed bat roosts. Generally, lesser short-tailed bat roosts are further inside the forest, the roost cavities often have multiple entrances, are lower to the ground, and have much larger entrance and internal dimensions than roosts used by long-tailed bats. Lesser short-tailed bat roosts are used by far greater numbers of bats (several 100s) and could be occupied for up to weeks at a time. Roosts are also re-used on a more regular basis than roosts of long-tailed bats (Sedgeley 2003).

Greater short-tailed bats

Virtually nothing is known about the ecology of greater short-tailed bats (Daniel 1990; Lloyd 2005). They were once present throughout New Zealand but had disappeared from most sites before Europeans arrived; possibly succumbing through predation by kiore brought into the country by the early Polynesians. Greater short-tailed bats were significantly larger than lesser short-tailed bats though their size varied in different parts of New Zealand (Worthy et al. 1996; Worthy & Scofield 2004). Little is known about habitat use, but they were likely confined to forests where they fed on a variety of invertebrate and plant foods (Lloyd 2005).

References and further reading

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Conservation management of New Zealand bats

The purpose of this section is to:

- Describe key drivers for the management of bat populations
- Describe the objectives of the Bat Recovery Programme
- Outline the desired outcomes for bat conservation management
- Identify the triggers for developing bat conservation management projects
- Identify priority sites for undertaking conservation management
- Identify conservation management techniques that aim to sustain or improve the status of bat populations
- Identify management levels within DOC at which bat conservation should be advocated or undertaken

Key drivers

Key drivers of DOC's work on bats include:

- The New Zealand Biodiversity Strategy
    - Goal 3: 'Halt the decline in New Zealand's Biodiversity'. This goal requires that we 'maintain and restore a full range of remaining natural habitats and ecosystems to a healthy functioning state' and 'maintain and restore viable populations of all indigenous species and subspecies across their natural range'.
- The Conservation Act and its attendant Acts
- The Wildlife Act (1953) for legislative protection of bats
- DOC's Strategic Plan (the annual Statement of Intent)

Goals and objectives for bat recovery

Strategic goals:

- Preventing declines and extinctions of New Zealand taxa map directly to the Natural Heritage Outcomes described in the DOC's Statement of Intent

Overall objective of the Bat Recovery Programme:

- To secure key populations of bat taxa from extinction representing the full genetic and distributional range

Specific objectives (developed in 2003—Minutes of the Bat Recovery Group) (in priority order):

1. Maintain the security of at least one population of each taxonomic unit by conducting management programmes at priority sites
2. Ensure no further declines in priority populations at the edge of the range so that genetically distinctive populations are protected

3. Restore populations that have declined in the past
4. Establish new populations

Note that six different taxonomic types are currently recognised (Table 1). Progress towards these goals is described by O'Donnell (2010).

Desired outcome of bat management projects

Areas with significant bat populations should develop bat management projects. The ultimate outcome of bat management projects shall be the maintenance of long-term security of the bat populations in the area where work is being proposed and/or the restoration to environments within the area where numbers have declined. Seven general management techniques are available:

- Statutory advocacy
- Non-statutory advice and education
- Pest control
- Active protection of roosts sites
- Protection of aquatic and terrestrial foraging habitats
- Restoration of roosting and foraging habitat
- Translocations

Aspects of each can be customised for local bat conservation projects. In addition, inventory and monitoring programmes will measure where and when outcomes are being achieved.

Triggers for commencing management

The Bat Recovery Group has identified a number of reasons for identifying an area as a priority for some form of bat work. The major triggers include:

1. The area has been identified as a priority for management of bats by the Bat Recovery Group through its Priority Sites list (Table 2) or objectives of the Recovery Plan (Molloy 1995).
2. Strategic directions for conservation work in a conservancy, as outlined in the Conservation Management Strategy, DOC's current Statement of Intent, or the New Zealand Biodiversity Strategy.
3. Presence of bats in a priority management site for biodiversity (e.g. Ecosystem Optimisation site, Mainland Island, Operation Ark site, Kiwi Zone).
4. Historic evidence of the presence of a nationally important bat population that the Bat Recovery Group is unaware of.
5. The site represents an area of uncertainty (no previous work on bats).
6. There is a major new threat to an area to which DOC can respond; for example, through Resource Management Act processes, district and regional planning, concessions applications.

These triggers are not static. The Bat Recovery Group acknowledges that new information comes to light at regular intervals, which may influence the form and direction of bat management projects. In addition, new drivers may emerge, which require the commencement of new work (e.g. new proposals for developments that may impact on bats). Where the Bat Recovery Group is made aware of new drivers, these will be signalled in the annual Bat Recovery Group recommendations (see olddm-715142 for 2004/05, and olddm-723120 for other years) or in the minutes of the Bat Recovery Group meetings.

Priority populations

Priority populations of bats for management (recognised by the Bat Recovery Group as at 2003 and still current in 2012) are listed in Table 2. These represent 24 populations from the eight recognised taxa for management. These are not all the sites where these bats occur, but represent the best populations, both core populations and outliers at the edge of their range, and the minimum number of sites where management should occur to ensure the security of these taxa. It is envisaged that these lists will be revised once every 5 years by the Bat Recovery Group as our knowledge of populations expands.

Table 2. Recommended priority sites for management of bat populations and main management techniques to be applied at each.

Taxon	Site	Conservancy	Secure	Management area identified	Statutory advocacy	Non-statutory advice	Pest control	Protection of roosts	Protection of foraging sites	Restoration of foraging and roost sites
1. Northern short-tailed bat	Little Barrier	Auckland	Yes	Done	-	-	Island invasion contingency plan	-	-	-
	Mainland	Northland	No	Needed	✓	✓	✓	✓	✓	✓
2. Eastern short-tailed bat	Urewera Whirinaki	East Coast / Hawke's Bay and Bay of Plenty	Yes	Needed	✓	✓	?	?	-	-
3. North western short-tailed bat	Ōhakune	Tongariro /Taupō	No	Done	-	-	✓	✓	-	-
	Pureora	Waikato	No	Needed	✓	-	✓	✓	✓	-
4. Southern North Island short-tailed bat	Tararua	Wellington	No	Done	-	✓	✓	✓	-	Establish second population
5. South Island short-tailed bat	Ōpārara	West Coast	No	Needed	-	✓	✓	✓	-	-
	Eglinton	Southland	No	Done	-	-	✓	✓	✓	-
	Codfish	Southland	Yes	Done	-	✓	Island	-	-	-

Taxon	Site	Conservancy	Secure	Management area identified	Statutory advocacy	Non-statutory advice	Pest control	Protection of roosts	Protection of foraging sites	Restoration of foraging and roost sites
							invasion contingency plan			
6. Greater short-tailed bat	Tītī Islands	Southland	No	Needed	-	✓	✓?	✓	✓	✓
7. South Island long-tailed bat	Geraldine	Canterbury	No	Done	✓	✓	✓	✓	✓	✓
	Eglinton	Southland	No	Done	✓	✓	✓	✓	✓	-
	Dart	Otago	No	Done	✓	✓	✓	✓	✓	-
	Maruia	West Coast	No	Done	✓	✓	✓	✓	✓	-
	Landsborough	West Coast	No	Done	-	-	✓	✓	-	-
	Catlins	Otago	No	Needed	✓	✓	✓	✓	✓	-
	Waikaia	Southland	No	Needed	✓	✓	✓	✓	✓	-
	Stewart Is	Southland	No	Needed	✓	✓	✓?	✓?	✓?	✓?
8. North Island long-tailed bat	Whareorino	Waikato	No	Needed	✓	✓	✓	✓	✓	-
	Ruakurī	Waikato	No	Needed	✓	✓	✓	✓	✓	✓?
	Puketitiri	East Coast / Hawke's Bay	No	Done?	✓	✓	✓	✓	✓	✓?

Taxon	Site	Conservancy	Secure	Management area identified	Statutory advocacy	Non-statutory advice	Pest control	Protection of roosts	Protection of foraging sites	Restoration of foraging and roost sites
	Ōhakune	Tongariro/Taupō	No	Needed	-	✓	✓	-	-	-
	Whanganui	Wanganui	?	Needed	?	?	?	?	?	?
	Whangarei	Northland	?	Needed	?	?	?	?	?	?

Recovery potential

Bats are very long-lived, attempt to breed each year, and populations have good recovery potential if threats are removed. Potential habitats to restore populations are extensive. There is a strong interest in conservation of bats across the breadth of the community, which indicates a strong potential for developing cooperative conservation projects. However, because bats only give birth to single young, once a year, then recovery will be slow and difficult to detect in the short term.

Management techniques

Management techniques for general restoration work, and in the case of DOC, for incorporation into business plan work plans include the following components:

- **Establishing the presence and significance of bats in an area and at what point to initiate management**
  Inventory aimed at establishing the presence of bats or the relative size of populations in particular areas is an important task in areas where there is uncertainty about the status of bats. This is because significant populations that as yet are unrecognised may be discovered, and these have the potential to be core management sites. Staff require a good understanding of the significance of bat populations in an area so that they can determine which of the management actions listed below are appropriate for them. The type and level of management required for new significant populations should be decided in consultation with the Bat Recovery Group.
- **Statutory advocacy**
  Statutory advocacy should focus on requirements aimed at protecting significant bat habitat (particularly in relation to the Resource Management Act 1991). Important actions include ensuring significant bat habitats are recognised and protected using classifications such as Significant Natural Areas (SNAs) and specific rules (e.g. land clearance and firewood cutting rules) in district, city and regional plans. Making submissions on activities that may either benefit or impact on bats is warranted in many situations. Concessions applications and Assessments of Environmental Effects (AEEs) need to include assessments of potential impacts on bats and develop appropriate mitigation techniques.

Specific examples of applications that should be evaluated for impacts on bat communities include:

- Water abstraction proposals that reduce availability of aquatic foraging habitat
- Building or modifying structures (e.g. roads, canals) that might impact roosting or foraging habitats
- Removal of trees from roadsides, reserves, campgrounds, tracks, etc. that may provide roosting habitat
- Forest logging on private land
- Poisoning operations that propose using new, non-approved baits and lures that potentially attract bats
- Gravel extraction proposals on riverbeds where significant bat foraging habitat occurs
- Proposals to dam rivers where significant bat foraging habitat occurs

A very important area of advocacy occurs in relation to making submissions on Sustainable Forestry Management Plans on private land in relation to Forests Amendment Act 1993 requirements. Logging generally targets a significant proportion of trees preferred by bats for roosting. Unless roosting patches are identified and protected, there is a high risk of localised tree selection wiping out a population. Bat detectors can be used to determine the presence of bats. Any sustainable management systems proposed for use in the future need to leave sufficient trees to ensure bat populations survive. Conservation officers with responsibilities for bat conservation should use data on the type and sizes of bat roosts to argue for rules to prevent impacts of logging and forest clearance on bats through their statutory planning procedures and input into Sustainable Management Plans, Personal Use Applications. Sustainable Management Plans need to demonstrate that safeguards are in place so that bat populations are not threatened. Tree preferences are summarised below.

Roost trees for long-tailed bats

Long-tailed bats roost in trees with large stem diameters (diameter at breast height—DBH). Most roost trees are greater than 80 cm DBH, and range up to 250 cm DBH (Fig. 4). Such trees are usually 200–650 years old. Long-tailed bats roost in small- to medium-sized cavities that are usually high up trees (15–20 m from the ground). Bats move to a new roost tree virtually every day, and one group can use over 100 different roosting trees. In Fiordland, long-tailed bats roost in red beech (74%), standing dead trees (21%), silver beech (4%) and mountain beech (1%). In Northland roosts were in the large kauri trees. In podocarp-hardwood forests long-tailed bat roosts have been found in rimu, miro, kahikatea, mataī, and tōtara, from 50–180 cm in diameter.

Roost trees for lesser short-tailed bats

Lesser short-tailed bats tend to use larger roost cavities than long-tailed bats. Lesser short-tailed bats living in mixed beech forest roost in splits and hollows mainly in large diameter red beech trees 40–160 cm DBH (Fig. 5). Bats in podocarp-hardwood forest show a similar dependence on large diameter trees, including Hall’s tōtara, rimu, southern rātā and miro. Most roosts are in trees greater than 80 cm in diameter. However, some are also in smaller trees, which are most often used by solitary bats, groups that require smaller cavities for hibernation, and for mating/singing. Lesser short-tailed bats may use a particular roost for just a day, or continuously up to 6 weeks, before moving to another tree roost.

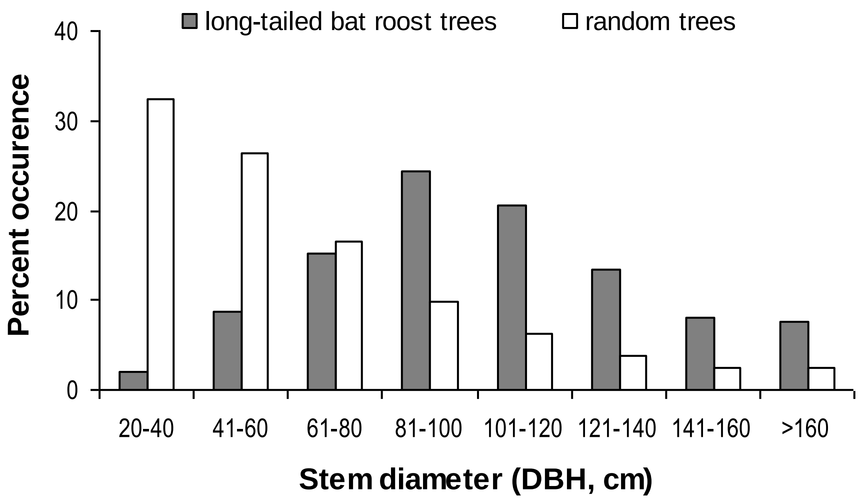

Figure 4. Stem diameters of beech trees used by long-tailed bats in the Eglinton Valley.

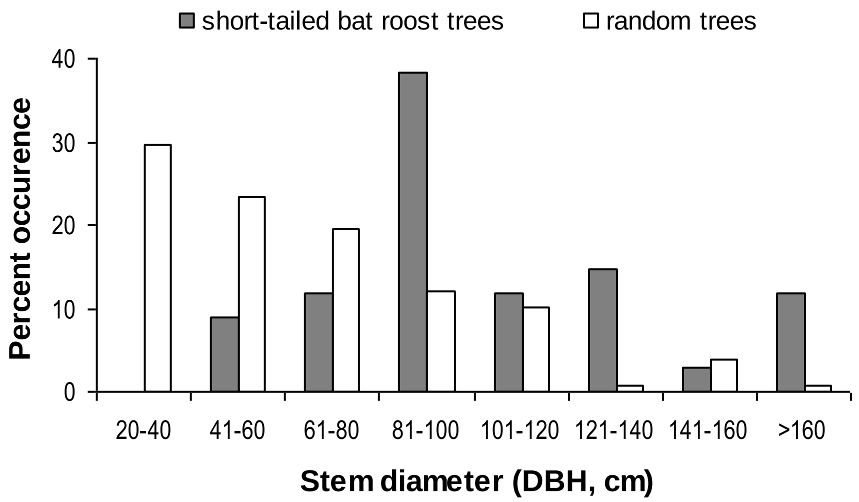

Figure 5. Stem diameters of beech trees used by lesser short-tailed bats in the Eglinton Valley.

Information on preferred trees can be found in Sedgeley & O'Donnell (1999a,b, 2004) and Sedgeley (2003). There is also a DOC fact sheet, 'Protecting old-aged forest trees for New Zealand bats' on the DOC website[2] (O'Donnell 2001).

Non-statutory advice and education

Non-statutory advice should focus on achieving conservation outcomes for bats by increasing awareness of the values associated with bats amongst local communities and encouraging private landowners to protect bat habitat. Actions should include giving public talks, organising field trips, involving the public or specific interest groups in conservation, writing fact sheets and circulating relevant information and fact sheets.

[2] http://www.doc.govt.nz/conservation/native-animals/bats/protecting-old-aged-forest-trees-for-new-zealand-bats/

Non-statutory advocacy and education are powerful tools to:

- Increase landowner awareness of preferred maternity roosts so they don't fell them
- Increase security for existing known roost trees
- Encourage maintenance of existing habitat surrounding roosts
- Encourage enhancement and restoration of sites using replacement plantings
- Undertake relevant statutory advocacy
- Facilitate setting up a local landcare group for bat conservation

For example, in South Canterbury, a DOC-employed rural advocate is working with community groups, and this provides a good model for other conservancies if this particular threat occurs in them. The advocate is working to stop or reduce loss of maternity roosts to wood cutting, clearance and senescence by working with local land owners, district and regional council staff and other interested parties.

A workshop held as part of the National Bat Conference in 1998 identified the major messages that it was important to get across to the public, major target groups and existing resources:

Major messages—what we want to achieve:

- Yes there are bats in New Zealand (promote values/interest)
- Where are bats in New Zealand? (feedback from public/DOC on sightings)
- Conservation of mature trees (major roosting sites are still threatened by logging)
- Need to monitor population trends (advocacy to help this happen)

Target groups (messages may need to have a different emphasis for each group):

- Land owners/farmers/forestry companies
- DOC managers and area offices
- Territorial authorities/councils
- Schools
- Iwi
- Interest groups (e.g. WWF-NZ, tramping groups/Forest & Bird)
- Sponsors
- Service groups
- Politicians
- Journalists
- Research groups

Important resources include:

- DOC fact sheets[3]

[3] http://www.doc.govt.nz/conservation/native-animals/bats/protecting-old-aged-forest-trees-for-new-zealand-bats/

- The WWF's Bat Pack (Jones, J. 1996: Bat Pack: Discovering New Zealand's native bats. World Wide Fund for Nature New Zealand, Wellington).
- Web based information on bats
- Zoo exhibits (flying foxes)
- WWF book
- DOC visitor centres
- Talks by bat workers
- Newspaper/magazine/radio and TV articles
- Summer programmes/visitor programmes (e.g. DOC Southland's walks with the bats)
- *Dactylanthus* advocacy

Pest control to enhance habitat quality and reduce risk of predation

DOC is undertaking, or has undertaken, mammal pest control programmes in several areas where bats are found (e.g. Pureora, Urewera, Waitaanga, Rangataua, Landsborough Valley, Eglinton Valley, and Whenua Hou). Introduced mammal pest species are considered to be a major threat to the continued viability of bat populations and integrated and effective pest control programmes that target possums, rodents, stoats or feral cats are likely to benefit bat populations if effort is intensive enough. Control of introduced pests will benefit bats in two ways:

1. Threats of direct predation on bats will be reduced.
2. Food resources available to bats will be improved.

Possums, rodents and stoats consume large numbers of insects, while possums and rodents reduce the availability of fruit and nectar. Both rats and stoats are killed during 1080 possum control programmes, although the kill-rate is not always consistent. The size of management areas should take into account the home range size of bat colonies (which range over areas of > 10 000 ha), so management areas smaller than this may not protect a colony sufficiently. Selection of habitats by bats for roosting and feeding is not random, therefore selection of management sites would need to include suitable bat habitats.

Integrated pest control should be undertaken in forests that are significant for bats. As a minimum, these should include sites currently recognised as important (Table 2). Such operations need not simply focus on protection or restoration of bat populations. Existing biodiversity projects aimed at improving forest condition (e.g. Hērangi Range, Eglinton Valley, Pureora) will have the potential to benefit bat communities as well as many other threatened species associated with forests (e.g. kiwi, kākā, whio, mōhua, mistletoes).

Contingency plans to minimise the risk of predators arriving on offshore islands, and consequent management actions are important for the protection of bat populations on Whenua Hou, Hauturu and Kapiti islands.

Specific projects may be required to protect *Dactylanthus taylorii* sites or special sites where significant bat populations occur, but no other threatened species (e.g. Hanging Rock area, South Canterbury—O'Donnell 2000a; Grand Canyon Cave, Maniapoto—O'Donnell 2002).

Pest control operations that use toxins need to be planned carefully to ensure there are no risks to bat populations.

Long-tailed bats are unlikely to be at any risk from toxins because the risk of their encountering toxic baits is virtually non-existent. They rarely feed within forests, they feed entirely on the wing on flying insects that would not come into contact with baits. Additionally, they usually hibernate in winter when many bait drops are planned.

The feeding habits of lesser short-tailed bats make them vulnerable to toxins in two ways: either from bats directly consuming toxic baits, or from secondary poisoning by consuming arthropods that feed on baits (Lloyd & McQueen 2000; Sherley et al. 2000). There is one record of a short-tailed bat being found dead on cyanide bait (Daniel 1990). However, feeding trials with captive lesser short-tailed bats, and a trial in which fluorescent dyed non-toxic baits were broadcast in an area inhabited by lesser short-tailed bats, showed that they did not consume carrot- or grain-based baits that are commonly used with 1080 and second-generation anticoagulants (Lloyd 1994). High concentrations of 1080 can persist in arthropods for several days after they have consumed baits (Booth & Wickstrom 1999; Eason et al. 1993). However, no harmful impacts were detected in short-tailed bat populations that were monitored through two aerially broadcast poisoning operations using pollard baits, one on Whenua Hou using brodifacoum (Sedgeley & Anderson 2000), and one in Rangataua Forest in the central North Island using 1080 (Lloyd & McQueen 2002).

Although it is reasonable to assume that these poisoning operations probably did not cause major mortality of lesser short-tailed bats, the trials on Whenua Hou and Rangataua Forest were unreplicated. Several replicate trials would be required in a variety of circumstances before a generalised conclusion can be justifiably drawn about mortality of lesser short-tailed bats during aerial poisoning operations.

Risk to lesser short-tailed bats is greatest where new baits or lures are proposed for use. Such proposals need to be carefully evaluated with non-toxic bait trials. For example, in the 1990s it was shown that lesser short-tailed bats consumed jam-baits that were being used in poison operations at the time. In a recent trial, the survival of tagged lesser short-tailed bats was monitored through a pindone in bait stations operation in the Eglinton Valley (O'Donnell et al. 2011). In this study, survival of bats appeared to be enhanced significantly. Similarly, long-tailed bats in the Eglinton Valley appear to be increasing slowly following a number of 1080 and pindone operations aimed at controlling rats (C. O'Donnell, unpubl. data).

Protection of roost sites

Known roost sites should be identified and access limited and/or regulated to minimise disturbance. Advocacy aimed at protecting old-aged trees on private land should be undertaken. Consideration should be given to potential conflicts between other management work and protection of potential roosts (e.g. felling of standing dead trees for track, hut and campsite maintenance or road widening).

Minimising disturbance of cave and rock-roosting bats is essential. Disturbance by rock climbers, particularly in winter when bats used the same limestone crevices used by climbers, was also identified as a potential risk to long-tailed bats at Geraldine (Griffiths 1996). Disturbance of cave-roosting bats is still a concern. An apparent decline in the number of bats day-roosting in Grand Canyon Cave over 5 years (1992–97), coincided with increased human use of the cave (C. Smuts-Kennedy, pers. comm.). Instances where all bats in the cave have taken flight while people have been watching them have been recorded in recent years (N. Miller, pers. comm.). Disturbance in caves in winter can reduce survival significantly.

Protection of freshwater and terrestrial foraging habitats

Protection of foraging habitats can be achieved through statutory and non-statutory advocacy, through legal protection of significant sites, and through active management of sites already in legal protection. Projects should aim to identify significant foraging habitats in each area, review their protection status and actively pursue formal protection if deemed necessary for increasing overall protection. Surveys using bat detectors should be undertaken in areas near those significant habitats already identified because comprehensive surveys of all areas likely to have bats have not yet been undertaken.

Restoration of roosting and foraging habitat

There is significant loss of maternity roosts to habitat clearance, wood cutting and natural aging of remnant stands of native forest in some areas (O'Donnell 2000b). In South Canterbury, no new roosts are being formed because grazing inhibits regeneration and bats are then forced to use poor quality roost sites resulting in low breeding success.

Restoration of bat communities in environments within an area where numbers have declined can be achieved by restoring forest and wetland remnants on agricultural land (foraging habitat), reducing grazing, replanting roosting tree species, and providing predator-proof artificial roost boxes within forest remnants. Trials are currently being undertaken using artificial 'Schwegler' woodcrete bat houses (these have proven highly successful for forest bat conservation overseas). Tree plantings may be necessary in areas where natural regeneration has been inhibited. Forest remnants that contain roost sites are likely to benefit from fencing and exclusion of stock. In areas where it is not possible to fence extensive areas of habitat, low-cost fencing of small patches around roost sites may be sufficient to afford increased protection.

Translocations

Currently, there are no accepted techniques available for translocating bats to new sites. However, techniques are currently being developed with translocations of lesser short-tailed bats to Kapiti (Ruffell 2006). If this translocation is successful, then translocation may become a useful tool in the future. The Bat Recovery Group will support well-planned trials that are resourced adequately and contain protocols for measuring the success of a trial.

Monitoring outcomes

Monitoring across a whole area and all management operations will not be practical or advisable, therefore programmes should focus on monitoring several representative populations or operations (Table 2) so that the difference that is being made by management can be measured and reported on in accordance with DOC's Natural Heritage Management System.

Who in DOC should be responsible for bat conservation

Bat conservation involves a wide range of staff in DOC, not just delivery staff and those specifically focused on biodiversity. The following list is not exhaustive, but key staff members that should be involved in bat conservation include:

- Area Managers
- Rangers
- Programme Managers
- Bat Recovery Group members
- Technical Advisers
- Statutory planning staff
- Concessions staff
- Staff responsible for evaluating Resource Management Act related proposals, water right applications, Significant Natural Areas, etc.
- Staff responsible for evaluating Forests Amendment Act applications on private land
- Science staff with advisory capacity

Relationships with other recovery programmes

A number of other DOC recovery programmes have direct relevance to the Bat Recovery Programme because they share similar management sites and/or threatening processes (e.g. mōhua, kiwi, kākāriki, kākā, whio, *Dactylanthus* recovery groups). Liaison with such programmes is strongly advised because of the shared benefits of conducting complementary research or management projects. There is also a strong link with Operation Ark, Mainland Islands and Kiwi Zone programmes; all have staff that coordinate and facilitate research on pest control issues in forests and operational responses to predator cycles.

Increasing the quality of management projects by improving knowledge

Staff should seek continual improvement in best management practice for bat conservation. This can be achieved by:

- Sharing information and skills via the Bat Recovery Group (and DOC's listserver L:\Bats)
- Encouraging further research on developing management techniques and increasing our understanding of bat ecology. This can be achieved by encouraging:
  - University students to undertake bat projects
  - Science staff to incorporate bat-related case studies into their programme of strategic research
  - Other Government research providers to increase their efforts with bats research
  - Conservancy and Area staff to incorporate rigorous monitoring of the performance of bat populations into their biodiversity programmes (especially island and 'mainland island'-type projects)

The Bat Recovery Group minutes are the repository of lists of priority research topics.

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Finding bats with bat detectors

Bats, echolocation and bat detectors

Bats and echolocation

Echolocation, also called biosonar, is the biological sonar used by several mammals such as dolphins, shrews, most bats, and most whales. The term was coined by Donald Griffin, who was the first to conclusively demonstrate its existence in bats (Griffin 1958). Many bat species use echolocation to navigate, to orientate and to forage, often in total darkness. Bats generate high frequency sound (ultrasonic) via the larynx and emit rapid ultrasonic pulses through their mouths, or less commonly their noses. By comparing pulses with the information contained in the returning signals (echoes), bats are able to locate, range and identify objects including prey (Fig. 6). Individual bat species echolocate within specific frequency ranges that suit their environment and prey types. Echolocation calls provide an opportunity to unobtrusively survey, monitor and identify bat species (Catto 1994; deOliveira 1998; Russ 1999).

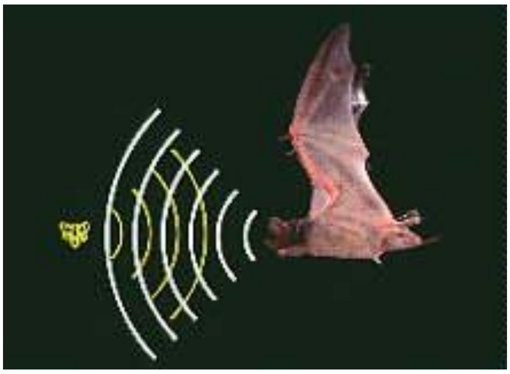

Figure 6. Diagrammatic representation of a bat echolocating.

How bat detectors work

The frequency of bat echolocation calls is generally much higher than humans can hear (ultrasonic). Ultrasound detectors, or bat detectors as they are commonly called, can be used to listen to bat echolocation calls, and are useful tools studying bats. Bat calls are picked up by the detector’s microphone, and transformed into lower frequencies that humans can hear. There are three main types of bat detector, each using a different technique for transforming ultrasound into audible sound:

- Heterodyne
- Frequency division
- Time expansion

Choosing bat detectors

Choosing type of bat detector

Bat detectors that use different systems for transforming bat calls will vary markedly in price; in sensitivity to bat calls; in the quality and information content of calls collected; in methods of storing, visualising and analysing calls; and in their ability to distinguish between calls made by different bat species. Choice of bat detector will ultimately depend on application. This section provides some background information on the three bat detector types to help readers choose the most appropriate detector for specific research or survey and monitoring needs. The section includes basic descriptions of the detectors and discusses some of their relative advantages and disadvantages. For readers requiring more technical information (e.g. how the detectors work; options for storing calls; analysis techniques) we suggest they read Parsons & Obrist (2004). Several companies supply bat detectors commercially (e.g. Stag Electronic, Pettersson Electronik, Titley Electronic, Tranquility and UltraSound advice). Some companies produce several types of detectors.

Heterodyne detectors

Heterodyning is a real-time method (i.e. you can hear the sound from the detector at the same time it is emitted by the bat). Heterodyne (also called narrowband) detectors monitor only one frequency at a time and can be tuned to specific frequencies. These detectors are very sensitive because they 'listen' through a narrow frequency window and can pick up relatively low noise levels. The relatively high sensitivity of these detectors has been demonstrated in the laboratory (Waters & Walsh 1994) and in the field (Parsons 1996). The most common bat detector used in New Zealand is the Batbox III heterodyne bat detector (Stag Electronics, Sussex, UK) (Fig. 7). DOC uses this detector as its standard for surveying using handheld bat detectors along line-transects (see the Toolbox method 'Bats: counting away from roosts—bat detectors on line transects'—docdm-590701). It is also fitted inside the most commonly used automatic bat detector and recording system used by DOC at the time of writing (see 'Automatic bat detector and recording systems' below, and see the Toolbox method 'Bats: counting away from roosts—automatic bat detectors'—docdm-590733).

Advantages:

- They are of relatively low cost compared to frequency division and time-expansion detectors.
- Heterodyne detectors have relatively high sensitivity compared with other detectors. For example, a Batbox III detector can pick up short-tailed bat calls over a greater distance than an Anabat frequency division detector (J. Sedgeley, J. Christie, pers. obs.), and they are twice as sensitive as many other heterodyne bat detectors, especially around 40 kHz (Walsh et al. 1993; Waters & Walsh 1994; Parsons 1996, 1997).

Disadvantages:

- The narrow frequency band of tuneable detectors means all bats calling outside the tuneable frequency range will be missed. Therefore, these kind of detectors are of limited value in countries where there are numerous bat species calling at different frequencies.

- Output from heterodyne systems does not provide enough information for detailed studies of bat echolocation calls. Unfortunately, the limited bandwidth to which the heterodyne detector listens blurs the duration, absolute frequency and frequency-time course of the original call in the heterodyned signal, thus rendering it unacceptable for spectral analysis (Parsons & Obrist 2004).
- It is frequently difficult to distinguish between calls of different bat species using heterodyne detectors. New Zealand long-tailed bats and lesser short-tailed bats call at different frequencies, but there is some degree overlap in their calls. The output from heterodyne detectors does not provide enough information to distinguish between them in all situations (see 'Bat detector frequency and bat identification').
- It is crucial to calibrate heterodyne detectors before using them in the field to ensure the frequency settings are correct (see 'Sensitivity and calibration').

(A) Stag Electronics Bat Box III heterodyne detector. **(B)** Titley electronics Anabat frequency-division detector. **(C)** Pettersson Electronik D980 time expansion detector.

Figure 7. Examples of detectors that use different methods for transforming ultrasound into audible sound.

Frequency division detectors

Frequency division (also called count down or broadband) transform the entire ultrasonic frequency range of a bat call without tuning. Output from frequency division detectors is usually recorded onto a tape recorder, digital recorder such as a MiniDisc, MP3 system or directly onto a computer. Computer software can then be used to visualise and analyse call structure and aid species identification. In Australia the Anabat system is widely used (Fig. 7). It has several options for storing the recorded calls. Calls can be recorded directly onto a laptop or PDA computer or a Compact Flash (CF) card recorder. Calls are examined by firstly digitising them onto a computer using a zero-crossing interface module and then using Anabat or AnaLook software to visualise and analyse the recorded calls (Parsons & Obrist 2004; Reardon 2004).[4]

[4] For more details, see http://users.lmi.net/corben/anabat.htm#Anabat%20Contents

Advantages:

- They enable the entire range of frequencies to be monitored simultaneously (i.e. they can listen for long-tailed bats and lesser short-tailed bats at the same time), thereby increasing sampling effort.
- The Anabat system has several options for storing the recorded calls and can be linked to a delay or time-switch. These features make the system very suitable for remote/unattended surveys.
- Output from frequency division detectors can contain more information than heterodyne detectors, including characteristics such as maximum, minimum and average frequency, duration, and time between calls.
- Output from frequency division detectors contain enough information to clearly distinguish between long-tailed bats and lesser short-tailed bats (trialled with Anabat detectors).
- Frequency division systems are less expensive than time-expansion systems.

Disadvantages:

- Overall, frequency division detectors can be less sensitive than other types of detector (Parsons 1996).
- If the division ratio is set too low, calls of bats using high frequencies may be lost (probably not an issue with New Zealand bat species).
- The methods by which frequency division detectors transform bat calls can lead to misleading output (i.e. will not always accurately represent all the characteristics of a bat call) (Parsons & Obrist 2004).
- In Australia there are several pairs or groups of bat species that cannot be reliable distinguished using the Anabat system (Reardon 2004).
- Frequency division systems (detectors, additional hardware, and software) are more expensive than heterodyne detectors.

Time expansion detectors

Time expansion is not a real-time method of transforming bat calls. Time expansion detectors work by digitising high-frequency output from the microphone at high sampling rates. The signal is then converted back to an analogue waveform using a reduced sampling rate, thus effectively increasing the signal's duration, and so time-expanding it (Parsons & Obrist 2004). The slower speed reduces frequency to audible levels which can then be more easily analysed. Since the signal is stretched out in time it is possible to hear the whole range of frequencies that the bat is using. Again the output is usually stored on a tape recorder, MiniDisc, MP3 system or computer for analysis. Some time-expansion detectors are also capable of producing heterodyned and frequency division output. When combined with a laptop computer and signal analysis software (such as Pettersson Electronik's BatSound) the output from time expansion detectors provides field workers with high quality information on bat ultrasound, and the most accurate reproduction of the bat call (Catto 1994; Russ 1999; Parsons & Obrist 2004) (e.g. Fig. 8). DOC has one time-expansion detector, a Pettersson Electronik D980.

Advantages:

- Time expansion is the only technique that preserves all characteristics of the original signal, making time expanded signals ideal for sound analysis in the laboratory. Since the signal is stretched out in time, it is possible to hear details of the sound not audible with other methods.
- Some time-expansion detectors are also capable of outputting heterodyned, frequency-divided and unmodified high-frequency signals.

Disadvantages:

- Time-expansion systems are much more expensive than heterodyne and frequency division systems.
- At present it is not possible to sample continuously using time expansion.

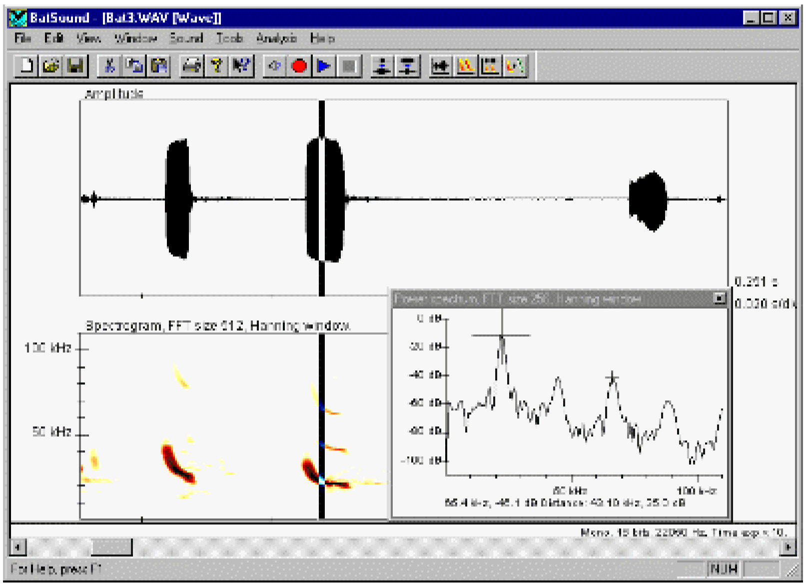

Figure 8. Output from a time expansion bat detector system analysed using BatSound software from Pettersson Electronics.

Automatic bat detector and recording systems

Bat detectors can be used remotely as well as manually. The output from detectors can be recorded and stored. By recording the output from the detector a permanent record of part or whole night’s activity can be kept. Several automated systems have been developed which use different types of bat detectors and methods for storing data. Consequently, relative effectiveness and cost vary (Parsons & Obrist 2004). These days, data are most frequently stored on SD cards. Many systems include timers, delay switches or voice-activated tape recorders that allow units to be left in the field and activated only when a call is detected.

Several automatic systems have been developed by DOC based around the Bat Box III heterodyne detector powered by a rechargeable 12 V sealed gel battery, a voice activated tape recorder, and a talking clock, and are used widely for inventory and monitoring of bats. The first system was relatively simple and inexpensive (O'Donnell & Sedgeley 1994)[5] (Fig. 9). This system has several disadvantages:

- The bat detector, tape recorder, and talking clock all function as separate units and each require different sized batteries.
- Tape recorders pick up a variety of audible environmental sounds, not just ultrasonic sounds coming through the detector.
- There is no timing mechanism to shut off the system during daylight hours. Despite these limitations this system functions perfectly well in accessible locations where the unit can be checked regularly.

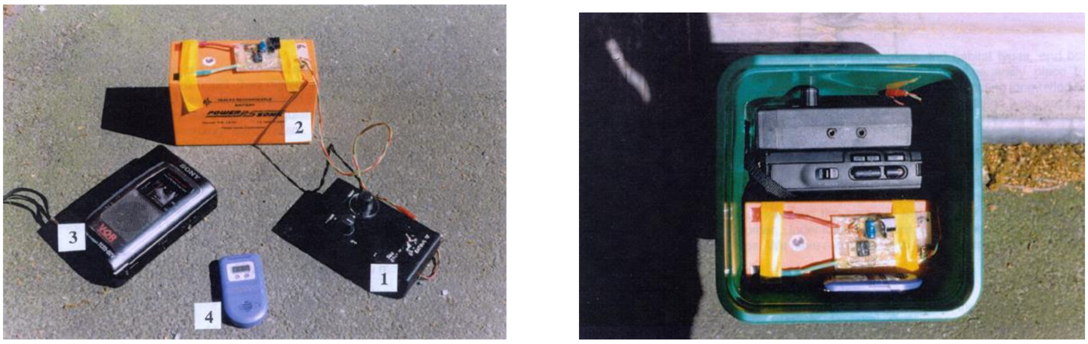

Figure 9. Automatic bat detector and recording system developed by O'Donnell & Sedgeley (1994). The system is relatively cheap but has several disadvantages.

The DOC Electronics Workshop has developed and improved this system. The latest models include an electronic controller which replaces the mechanical talking clock and timing mechanism. The units developed by DOC are called automatic bat monitors or ABMs[6] (Fig. 10).

It is possible for outside agencies to purchase units through DOC. Contact the Electronics Workshop in Wellington for further details.

Figure 10. The DOC automatic bat monitor (ABM).

[5] http://www.doc.govt.nz/upload/documents/science-and-technical/docts05.pdf
[6] For more information, see 'ABM instructions' (olddm-759839).

In Australia the Anabat system from Titley electronics is widely used for remote surveys. It is particularly well suited for unattended bat detector surveys, with several options for storing the recorded calls including on cassette tape or compact flash card recorders. The system is based around a broadband detector which can be linked to a delay or time-switch (Fig. 11). This allows the system to be left in the field and activated only when a call is detected.

Figure 11. Anabat automatic recording system.

Bat detector frequency and bat identification in New Zealand

Call structure and pulse repetition rate in New Zealand bats vary geographically, with habitat type and with the bat's activity (Parsons 1997, 1998). Despite this, the calls of long-tailed bats and short-tailed bats are distinctive and can generally be used to differentiate between the two species. Peak amplitude of long-tailed bat calls is 40 kHz (Parsons 2001). A Batbox III on full volume can detect long-tailed bats on average 43.5 ± 9.8 m away when recorded along forest edges (C. O'Donnell, unpubl. data). Peak amplitude of lesser short-tailed bat calls is ca. 27–28 kHz (Parsons 2001), and their calls can be detected for approximately 20 metres (Lloyd & McQueen 2002).

Call overlap

Unfortunately there is overlap in call structure (fundamentals and harmonics) between long-tailed bats and lesser short-tailed bats, and a significant overlap in frequency when calls are monitored using heterodyne detectors (Parsons 2001). This means that Batbox III detectors set on 27–28 kHz will pick up calls of both long-tailed bats and lesser short-tailed bats, and detectors set at 40 kHz will also pick up calls of both bat species. Therefore, if surveys are conducted in a new area, or in an area where both long-tailed bats and lesser short-tailed bats are known to be present, it cannot be assumed that every call recorded at 40 kHz is made by a long-tailed bat and that every call recorded at 27–28 kHz is made by a lesser short-tailed bat.

There has been work to try and quantify the degree of overlap in calls between the two bat species using paired bat ABM units (one set on 27 kHz and the other on 40 kHz). The proportion of short-tailed bat calls recorded at 40 kHz appears to vary depending on the model of ABM used, the recording situation, the location in New Zealand, and the level of bat activity. For example, in the Eglinton Valley, 24% of a total of 666 short-tailed bat calls recorded at 27 kHz were also recorded at 40 kHz (O'Donnell et al. 1999). On Codfish Island only 3.9% of short-tailed bat calls recorded at 27 kHz were also recorded at 40 kHz (O'Donnell & Sedgeley 1994). In contrast, of 2927 long-tailed bats calls recorded at 40 kHz, < 1% was recorded on 27 kHz (O'Donnell et al. 1999).

Call overlap—putting it in perspective

Fortunately, the calls of the two bat species retain some of their distinctive characteristics at whatever frequency they are monitored. Lesser short-tailed bat calls tend to be relatively short in duration compared to long-tailed bats, and pulse repetition rate in lesser short-tailed bat calls is twice as fast as in long-tailed bats (Parsons 2001). Lesser short-tailed bat calls heard on a detector set at 40 kHz are often very faint, and may require an experienced observer to detect them. With practice and careful listening, it is possible to distinguish between the two species, but it is inevitable some calls will be miss-identified. Listening to reference calls may help with familiarisation.

Bat detectors and ABM units are important tools for determining presence or absence of bats in an area. Both species of bat are categorised as threatened, therefore any record of bats from a new area is valuable, even if call identification is not 100% positive. If species identification is important the site can be revisited and a frequency division or time expansion detector used to determine species. Unfortunately, these types of detectors are not widely available in New Zealand. An alternative is to use paired bat detectors (one set on 40 kHz, the other set on 27–28 kHz) to determine at what frequency most calls are recorded on. The most reliable method to identify bats to species is to examine them in the hand. Bat detectors can be used to determine areas of highest bat activity in which to place mist nets or harp traps.

Recommended frequencies

We recommend the following frequencies be used on the Batbox III detector:

- **To record long-tailed bats, detectors should be set at 40 kHz.**
  At 40 kHz, long-tailed bat calls are often loud, have longer call durations, and a slower pulse repetition rate than lesser short-tailed bats. Long-tailed bat calls have a relatively irregular rhythmical sound which can be described as a series of 'slaps' or 'thwacks'.
- **To record lesser short-tailed bats, detectors should be set at 27–28 kHz.**
  At 27–28 kHz, calls of lesser short-tailed bat are often softer (unless the bat flies very close to microphone) and have a shorter duration and a faster pulse repetition rate than calls of long-tailed bats. Lesser short-tailed bat calls have a more even or regular rhythmic pattern which can be described as a short burst of staccato 'clicks'.
- **To maximise chances of recording both species**
  Reliable identification of both species using one ABM unit can only be achieved by replacing the standard Batbox III heterodyne detector with either a broadband or time-expansion detector, and using a sound analysis computer program to analyse the recorded calls. Otherwise paired ABM units, one set on 27–28 kHz and the other set on 40 kHz should be used.

  Feeding buzzes made by long-tailed bats at 40 kHz could be confused with calls of lesser short-tailed bats. However, feeding buzzes are recorded far less often than the usual characteristic long-tailed bats calls, and seldom occur by themselves. Feeding buzzes can usually be heard at the end of a series of the more usual calls.

- **Examples of calls**
  Examples of calls of long-tailed bats and lesser short-tailed bats obtained using Batbox III detectors are available. The calls were recorded onto audio cassette tapes and converted to Windows Media Player files. The audio file 'Sequence of long-tailed bat calls' (olddm-574297) contains a total of six long bat passes. The bat sometimes sounds like it is going away and then flies back towards the microphone. The recording was made using an automatic system with a bat detector linked to a voice-activated tape recorder. The hissing noise is the sound of the tape recorder switching on and off between events. The audio file 'Sequence of lesser short-tailed bat calls' (olddm-574301) contains seven bat passes. They are of shorter duration and have a faster pulse repetition rate compared with the long-tailed bat calls.

Sensitivity and calibration

It is important to test bat detectors before use, particularly if using old used equipment. Sensitivity between Batbox III units can vary (O'Donnell & Sedgeley 1994; Arkins 1999). The most common causes are under-charged batteries, damaged microphones and miss-aligned frequency dials. It is recommended that DOC equipment is serviced at the end of each field season. The easiest way to check and to calibrate detectors is with the use of a 40 kHz signal generator. If a detector is working adequately, the signal tone should be audible through the detectors speaker at a distance of 40−50 m, providing the detector is pointed directly at the signal generator. The generator can also be used as a guide to re-align the frequency dial (O'Donnell & Sedgeley 1994). The DOC Electronics workshop should be contacted for advice and to find out if detectors have been calibrated before use.

Bat calls and other sounds on the bat detector

Bat calls are heard on the detector as series of clicks as a bat flies into range. A series of audible clicks is defined as a 'bat pass' (Furlonger et al. 1987). Passes are defined as a sequence of two or more echolocation clicks, and a period of silence separating one bat pass from the next. Occasionally it is possible to hear a very distinctive call on the detector that sounds like buzzing, or almost like someone is blowing a 'raspberry'. This call is known as a 'terminal' or 'feeding buzz'. Its purpose is to provide the bat with additional details of the object that it is targeting. As the bat gets closer to an insect, for example, the bat will rapidly increase the pulse repetition rate of its call to provide frequent updating of the distance to the target. It reaches its peak rate as it attempts to grabs its prey.

Bat detectors will pick up a range of high frequency sounds—not just bats. A heterodyne detector will pick up any high-frequency sound that is close to the frequency it is tuned in to. For example, insects can be very noisy on warm summer nights (e.g. cicadas, crickets), and electric fences make a very repetitive clicking noise. Observers should listen for a pattern in the sound to try and distinguish between bat calls and other sounds. Clicks from an electric fence, for example, are very slow compared with bat calls. Non-bat sounds are more likely to be stationary and close to the ground. If the battery in the detector gets low, it can create a feedback noise through the speaker.

Using bat detectors for survey and monitoring

Echolocation calls provide an opportunity to unobtrusively survey, monitor and identify bat species (Catto 1994; deOliveira 1998; Russ 1999). Detectors can be used manually (e.g. using handheld bat detectors to count long-tailed bat calls along line transects) or remotely using automatic systems (described above). For further information on surveying using line transects see the Toolbox method 'Bats: counting away from roosts—bat detectors on line transects' (docdm-590701) and O'Donnell & Sedgeley (2001)[7]. For more information on using automatic systems for inventory and monitoring see the Toolbox method 'Bats: counting away from roosts—automatic bat detectors' (docdm-590733).

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Species identification in the hand

An identification key is not necessary to identify long-tailed bats and lesser short-tailed bats. They are very easy to distinguish in the hand. This section describes differences between the two species focusing on simple external visual characteristics such as fur colour, ear shape and tail shape. There are also numerous differences in morphological measurements, echolocation calls and a range of behaviours. Differences in morphological measurements (and how to take such measurements) are dealt with in 'Handling, examining, measuring and releasing bats'. Differences in echolocation calls are described in 'Finding bats with bat detectors' and differences in general behaviours are outlined in 'Introduction to New Zealand bats/pekapeka'.

In contrast to long-tailed bats and lesser short-tailed bats, there is comparatively little known about greater short-tailed bats. Most of the distinguishing features of greater short-tailed bats are summarised below. A summary of the main differences among the three species can be seen in Table 3. For definitions of various technical and morphological terms see 'Handling, examining, measuring and releasing bats', in particular see the diagram shown in Fig. 44.

Table 3. Summary table of distinguishing features of New Zealand bats. Source: O'Donnell (2001).

<table>
<tr><th></th><th>Long-tailed bat
Chalinolobus tuberculatus</th><th>Lesser short-tailed bat
Mystacina tuberculata</th><th>Greater short-tailed bat
Mystacina robusta</th></tr>
<tr><td>Flying activity starts</td><td>Around sunset</td><td>After dark</td><td>After dark</td></tr>
<tr><td>Roosts</td><td>Native and exotic trees, caves</td><td>Native trees and caves</td><td>Native trees, caves, and seabird burrows</td></tr>
<tr><td>Tail length and position</td><td>Almost as long as head and body. Large 'v'-shaped interfemoral membrane. Post-calcarial lobe present.</td><td colspan="2">Very short, partly free of interfemoral membrane, projecting c. 7 mm on dorsal surface</td></tr>
<tr><td>Fur</td><td>Variable colour, fine and soft. Adult females usually rich chestnut brown upper parts. Males and non-breeders dark brown with blackish heads. Underparts pale brown</td><td colspan="2">Short and velvety, grey-brown, guard hairs over underfur</td></tr>
<tr><td>Jaws</td><td>Fleshy lip-lobule at corner of mouth.</td><td colspan="2">No lip-lobule</td></tr>
<tr><td>Hind legs</td><td>Small, delicate feet. Legs enclosed within interfemoral membrane</td><td colspan="2">Large, robust legs, not fully enclosed by interfemoral membrane</td></tr>
<tr><td>Claws</td><td>Without spurs on toes and thumbs</td><td colspan="2">With spurs</td></tr>
<tr><td>Ears</td><td>Small, broad, rounded</td><td>Large, pointed, extend to or beyond muzzle when laid forward</td><td>Large, pointed, do not reach muzzle when laid forward</td></tr>
<tr><td>Nostrils</td><td>Small</td><td>Prominent and narrow</td><td>Short and broad</td></tr>
<tr><td>Forearm length</td><td>37–46 mm</td><td>39–46 mm</td><td>45–48 mm</td></tr>
</table>

Summary description of greater short-tailed bats

Greater and lesser short-tailed bats share many general characteristics. Overall, greater short-tailed bats are larger and more robust than lesser short-tailed bats (Fig. 12) (Table 4), especially in the skull. Their molars and jaws are proportionately larger. However, proportionately shorter ears, forearm and wing elements in greater short-tailed bats give rise to some overlap between measurements from the two species (Daniel 1990; Lloyd 2005a,b). The potential for overlap is increased in southern New Zealand because size of the greater short-tailed bat decreases dramatically with increasing latitude while that of the lesser short-tailed bat does not (Worthy et al. 1996).

D. Merton

J.W. Manintosh

Figure 12. Greater short-tailed bats.

A recent study examined dental and skeletal measurements of lesser short-tailed bats and greater short-tailed bats from adjacent populations where the mean size of each is closest. Twenty lesser short-tailed bats from Codfish Island/Whenua Hou were compared with those from eight greater short-tailed bats from the southwest Muttonbird (Tītī) Islands off Stewart Island. The two species had complete size separation in most, and minimal overlap only in a few (five tooth and two cranial measurements), of the measured variables. All greater short-tailed bats were larger than any lesser short-tailed bat, had relatively smaller wing elements and differed in shape of teeth. The percentage increase in size from lesser short-tailed bat to greater short-tailed bat varied from15–20% for many measurements but is as great as 34.5% for molar (M3) width measurement. Forearm length (9.8%) and associated radius length (11.5%) showed the least increase (Worthy & Scofield 2004). Norberg & Rayner (1987) calculated that with a body mass estimate of 24.5 g, greater short-tailed bats had higher wing loadings and aspect ratios than lesser short-tailed bats. This indicates that greater short-tailed bats would have been faster in flight, with more rapid turns (i.e. more agile), but requiring larger turning radii (i.e. less manoeuvrable).

Table 4. Greater short-tailed bat measurements (mm). Data obtained from Lloyd 2005a,b; Daniel 1990; Hill & Daniel 1985; Dwyer 1962.

	Total Length	Snout-to-vent length	Wingspan	Forearm	Tibia	Ear length
Sample size	–	8	–	8	8	8
Minimum	70	65.4	290	45.3	18.2	17.7
Maximum	90	72.4	310	47.5	19.1	18.6
Mean	–	68.6	–	46.4	18.7	18.3

No one has handled a live greater short-tailed bat since the 1960s, so it is difficult to know how distinctive one would appear in the hand. The greatest differences between greater and lesser short-tailed bats are largely skeletal, therefore it may be difficult to distinguish between live specimens of the two species. Forearm length is one of the easiest measurements to take from a living bat (see 'Handling, examining, measuring and releasing bats'), but it has greater potential for size overlap between the two species than many other measurements. If a caught bat is suspected to be a greater short-tailed bat, perhaps the most reliable method would be to test the ear length against the muzzle—the ears of greater short-tailed bats do not reach their muzzle when laid forward. It is extremely important to take as many measurements possible, take photographs with a scale indicated, and if possible take tissue samples (see 'Collecting samples').

Comparisons between long-tailed bats and lesser short-tailed bats

Long-tailed bats and lesser short-tailed bats are very different in the hand. Long-tailed bats are relatively small and delicate, the body mass of adult bats ranges from 8.5 to 12.3 g, and their forearm length from 38.7 to 40.5 mm. Their fur is chocolate brown, or chestnut brown, and they generally have a placid temperament when handled. Lesser short-tailed bats are stocky in appearance and can be a third larger than long-tailed bats. Body mass of adult bats ranges from 11.4 to 22.0 g, and length of forearm from 36.9 to 46.9 mm. Lesser short-tailed bats have larger, more pointed ears, fur colour that has been variously described as grey-brown, beige or golden and can have a relatively aggressive temperament when handled (O'Donnell et al. 1999; Lloyd 2001; O'Donnell 2001).

Fur colour

Long-tailed bats

Fur colour is variable in long-tailed bats, and changes with age. Adult male and female long-tailed bats can have different fur colour. Adult females usually have rich chestnut upper-parts, sometimes with white tips to the fur. Males, and 1–3-year-olds of both sexes, are darker, with dark brown upper-parts and blackish fur around the head (O'Donnell 2001) (Fig. 13). Under parts are pale brown in both sexes often paler about the pubic region. The fine, soft dorsal hair is up to 7 mm long, with no differentiation into over-hair and under-hair (Dwyer 1962). The limbs, wing and tail membranes are almost naked and blackish-brown in colour (Fig. 13).

(a) C. O'Donnell.


(b) R. Morris


Figure 13. Fur colour in long-tailed bats. (a) Adult female long-tailed bat, note chestnut fur colouring. (b) Male/juvenile, note darker fur colour.

Lesser short-tailed bats

The fur of lesser short-tailed bats is generally grey-brown; it is short, dense, and velvety, sometimes appearing frosted. Guard hairs are present over under-fur. Fur colour can also vary with age. Occasionally the fur of adult females can sometimes appear to be almost golden coloured, whilst juveniles are often much duller. The bare skin of the ears, wings, nose, legs and tail is grey-brown. (Fig. 14).

B. D. Lloyd

R. Morris

Figure 14. Fur colour in adult lesser short-tailed bats.

Ears

Long-tailed bats

Long-tailed bats have smaller ears and a shorter tragus than lesser short-tailed bats. The ears of long-tailed bats are rounded distally, and the outer margin of the ear continues along the face, beneath the eye, as an antitragus, which terminates just behind the lip-lobule (Fig. 15). The more pronounced tragus extends from within the ear above the antitragus. It is narrow at the base, but widens and is rounded at the tip distally (O’Donnell 2005).

B. Ebert

R. Morris

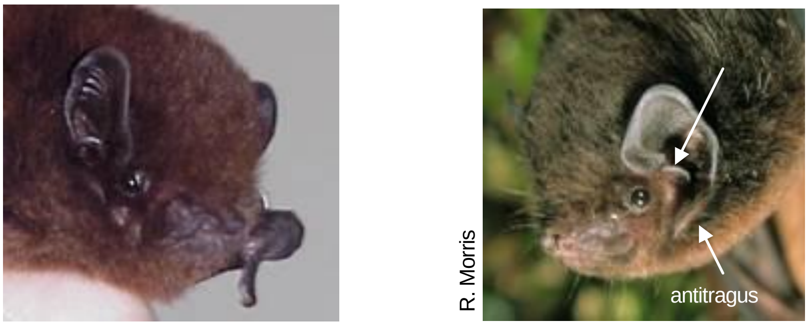

Figure 15. Ears of long-tailed bats. The ears are smaller and more rounded than those of lesser short-tailed bats. The tragus in long-tailed bats is small and rounded.

Lesser short-tailed bats

The ears of lesser short-tailed bats are larger than those of long-tailed bats (c. 18 mm long × c. 9 mm at base), oval, and simple, with a long (c. 10 mm) pointed tragus (Fig. 16a) (Lloyd 2005a,b). When handled lesser short-tailed bats can hold their ears in an upright position or can partially curl their ears, or fold them relatively flat against their heads (Fig. 16c,d).

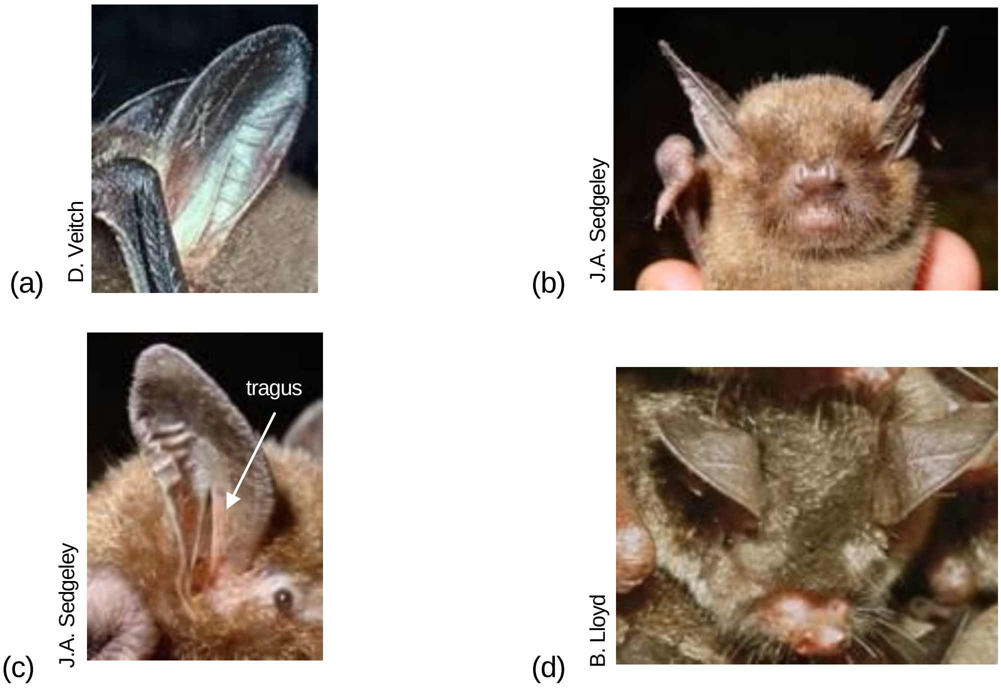

Figure 16. (a) The ears of lesser short-tailed bats are longer than ears of long-tailed bats and the tragus is long and pointed. (b) Ears fully erect. (c) Ear partially curled. (d) Ears flat.

Legs and tails

Long-tailed bats

The tails of long-tailed bats are fully enclosed within a large v-shaped tail membrane. The tail is almost as long as the head and body length (Fig. 17). Long-tailed bats have a calcar that extends from the heel as a strong process and supports almost half of the posterior border of the large tail membrane. A small, rounded post-calcareal lobe is present near the base of the foot (O'Donnell 2005). The legs and feet of long-tailed bats are thinner and more delicate than those of lesser short-tailed bats. The legs of long-tailed bats are also fully enclosed within the tail membrane (Fig. 17).

Figure 17. Tails of long-tailed bats. The tail is long and fully enclosed within a large v-shaped membrane. Note the dark colour of the tail and wing-membrane.

Lesser short-tailed bats

Lesser short-tailed bats have relatively short tails. The basal section of the tail lies enclosed within the tail membrane, but the tip projects freely c. 7 mm from dorsal surface of the membrane. The tail membrane is much shorter and more rounded than in long-tailed bats. Lesser short-tailed bats have a long curved calcar, but no posterior lobe. When lesser short-tailed bats are not flying the tail membrane is tightly furled away and the tail protrudes (Fig. 18). The legs of lesser short-tailed bats are unusually robust, and their feet are stout and broad (c. 6 mm) (Fig. 18). They also have a very fine talon, only just visible to the unaided eye, at the base of the inside curve of each claw which is a unique characteristic of the genus (Lloyd 2005b).

(a)


(b)


(c)


Figure 18. Tail and hind legs of lesser short-tailed bat. (a) Dorsal view showing tail membrane tightly furled away and tail protruding freely. (b) Dorsal view showing tail membrane almost fully extended. (c) Ventral view showing tail membrane almost fully extended.

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Inventory and monitoring methods for counting bats

A large number of inventory and monitoring methods are now available for bats. Choosing a method depends on the objectives or questions leading to the work you are doing, and their relative suitability for long-tailed bats or lesser short-tailed bats.

All inventory and monitoring methods for bats (Table 5) are available on the DOC website: http://www.doc.govt.nz/publications/science-and-technical/doc-procedures-and-sops/biodiversity-inventory-and-monitoring/bats/

Table 5. Inventory and Monitoring Toolbox method specifications for counting bats.

Introduction to bat monitoring
Bats: counting away from roosts: bat detectors on line transects
Bats: counting away from roosts: automatic bat detectors
Bats: counting away from roosts: visual counts
Bats: trapping away from roosts: inventory and species identification
Bats: exit counts at roosts: cameras and recorders
Bats: exit counts at roosts: simple visual counts
Bats: trapping at roosts: estimating population size
Bats: trapping at roosts: estimating survival and productivity
Bats: roost occupancy and indices of bat activity: infrared beam counters
Bats: roost occupancy and indices of bat activity: field sign
Bats: roost occupancy and indices of bat activity: automatic bat detectors
Bats: counting inside roosts
Bats: casual reports

The first step is reading the ‘Introduction to bat monitoring’ (docdm-590958), which describes the principles behind the inventory and monitoring objectives, and includes comparative tables and decision trees to guide you to the most suitable and cost-effective method to use to answer specific inventory and monitoring questions.

Not all methods are suitable for both bat species, and not all methods are appropriate for both inventory and monitoring. Therefore, the comparative tables are organised as follows:

- Methods for inventory of long-tailed bats
- Methods for monitoring long-tailed bats
- Methods for inventory of lesser short-tailed bats
- Methods for monitoring lesser short-tailed bats

The tables are further arranged by methods that count bats away from roosts, at sites or both. There are 14 methods; 4 methods for use away from roost sites, 8 at roost sites, and 2 that can be used in both situations. All methods are listed on each table and are linked to specifications that provide full details.

Each method is scored for its relative precision in answering the specific inventory and monitoring objectives; ✓✓✓ = Good; ✓✓ = Medium; ✓ = Poor; × = Not recommended; — Not applicable. The relative costs or resources required (equipment costs, personnel costs, skills required) for each method are also assessed and ranked as Low, Medium or High. Once you have been guided to a method(s), read the method specifications carefully to ensure it meets your study objectives.

Catching bats

New Zealand bats are protected fauna, and it is illegal to catch, handle or keep them without appropriate permitting and ethical approvals (see 'Permitting, ethics approval and training'). Permission for catching and handling bats will only be given if there is a valid reason for doing so, since all catching methods will disturb bats, and if used carelessly, will cause injury.

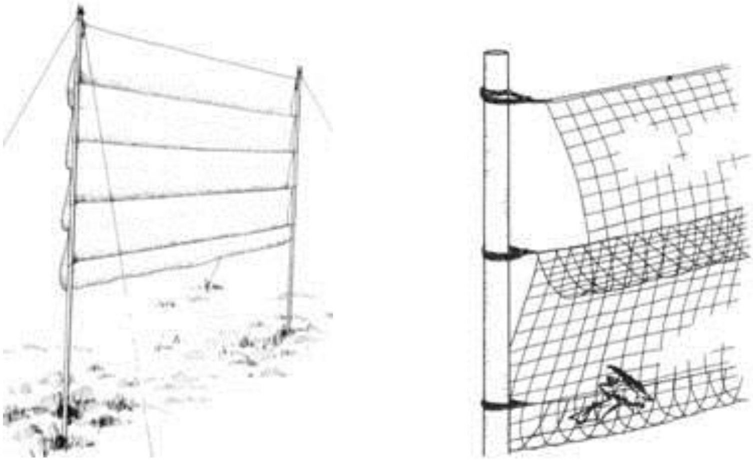
Bats are most frequently caught to enable positive species identification, to obtain morphological measurements, to mark them for a population study, to obtain genetic samples, to obtain a sample of their droppings for dietary studies, and to attach radio-transmitters to find roost sites or study home-range and habitat use. Bats may be caught both in free flight and at their roosts using a variety of techniques.

Techniques that are in common use and have proven successful for catching New Zealand bats are described in some detail below. The relative efficiency of these techniques for catching long-tailed bats and lesser short-tailed bats is also discussed. Methods that have not been trialled in New Zealand, but are proven techniques for other bat species will be mentioned briefly.

Mist nets

Description

Mist nets are fine nylon or terylene netting most commonly used in New Zealand for catching birds. Mist nets can only be obtained from the DOC Banding Office in Wellington. Sizes vary a little according to brand, but they are generally available in standard lengths of 6 m, 9 m, 12 m and 18 m, and usually 2 m high. The height of the net is divided into pockets or bags by 3–5 horizontal strings (shelf-strings) running the length of the net. Bats are usually trapped in these loose pockets of netting (Fig. 19). A tightly tensioned net with no pockets is much less likely to catch bats because they may simply bounce off. Mist nets are available in different mesh sizes, but the best size for catching bats is 36–38 mm. Nets with smaller sized mesh are easier for bats to detect with their echolocation calls, and larger sized mesh might allow bats to fly through. Nets commonly used to catch birds are adequate for catching bats (30–40 mm mesh size), but specially designed bat nets may be preferable. The Banding Office supplies specialised bat-nets that have a mesh size of 38 mm, and reduced sized pockets to lessen entanglement of bats.



T.P. McOwat, reproduced with permission, *Bat Workers' Manual*, © JNCC 2004. ISBN 1861075588

Figure 19. Mist net set up with poles. The height of the net is divided into pockets by five shelf-strings running the length of the net. Bats are usually trapped in these loose pockets of netting.

Advantages:

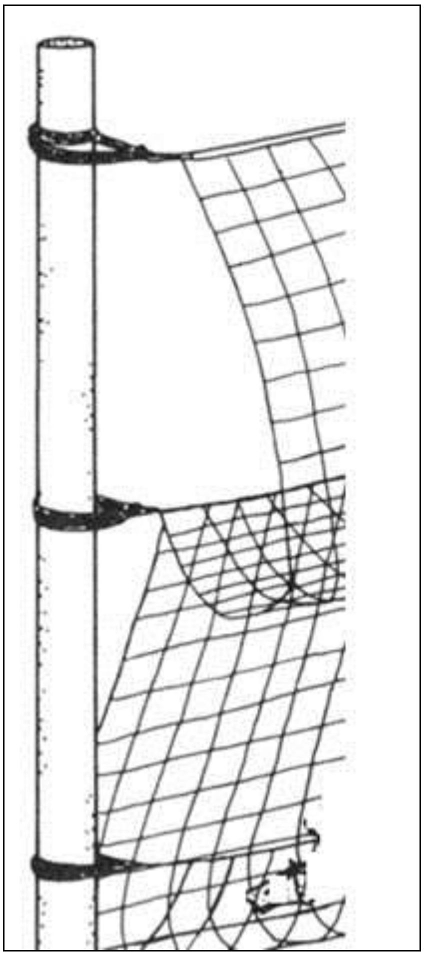
- Mist nets are much less expensive than commercially available harp traps.
- They are lightweight, and are easily transportable when folded away into small bags. At the time of writing bird mist nets ranged in price from $94–$260, and bat mist nets from $190–$350 each (excluding GST).
- Mist nets are very effective for catching lesser short-tailed bats.

Disadvantages:

- Long-tailed bats can easily detect mist nets, and consequently capture rates for this species are often very low.
- Bats can also become badly tangled in nets, so the technique may be more stressful on bats than other methods.
- Nets require continuous supervision.
- Large mist net rigs that incorporate several nets stacked on top of each other can be time-consuming to construct, and require a reasonably open area in the forest to set up.
- Nets are difficult to repair.

Setting the nets

Mist nets can be set using poles, or with rope/string. They can be set one net high, or several stacked one on top of the other (Fig. 20). If more than two nets are set on top of each other it is recommend a pulley system is employed so bats caught in the top portions of the net can be lowered easily (Kunz & Kurta 1988; Dilks et al. 1995).

(a) 

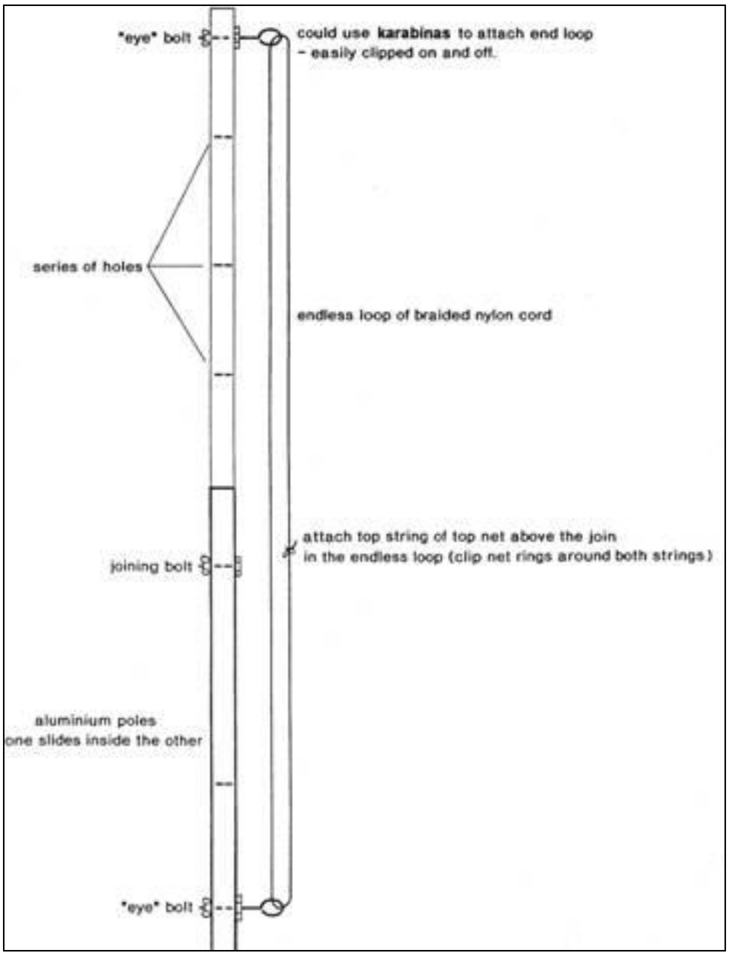
(b) 

Figure 20. Attaching mist nets to poles. (a) Wrapping end loops around pole. (b) Aluminium pole with simple pulley system attached for raising and lowering the net. Adapted from Dilks et al. 1995.

Pole systems

Aluminium tubing makes ideal mist net poles since it is lightweight and strong. Tubing of differing diameters can be cut into telescoping sections and then joined together to create the desired height. This can be most simply achieved by using two lengths of tubing, one with a smaller diameter that can slide inside the larger. The length of the pole can be varied by bolting through the larger diameter tube at different heights. The addition of an eye-bolt at the top and bottom of the pole and an endless loop of cord (to which the nets are tied) creates a simple pulley system for raising and lowering the nets. (Kunz & Kurta 1988; Dilks et al. 1995) (Fig. 20). An alternative method is to glue a shorter length (25 cm) of smaller diameter tubing into the top of a section of 1 m tube so it protrudes 15 cm. This can then be inserted into the base of the next section to produce net poles of the required height. This method has been used successfully to create poles 6 m tall (Churchill 1998). Poles need to be pushed firmly into the ground and guyed securely to take the strain of the net. Alternatively 'pole-holders' made from a short length of larger diameter tubing can be used to hold the mist net poles in position. One end is sharpened to make into a 'spade-point', and it can be hammered or pushed into the ground (Churchill 1998).

Rope systems

Cord or string can be used instead of poles (Fig. 21). An endless loop of cord is passed over a branch and under a tree root or log. The addition of a karabina or metal ring at the top and bottom through which the endless loop passed will allow the cord to run smoothly for raising and lowering nets. This technique will only work if there are branches available in the right places and the correct distance apart, otherwise it can sometimes be difficult to achieve suitable net tensioning.

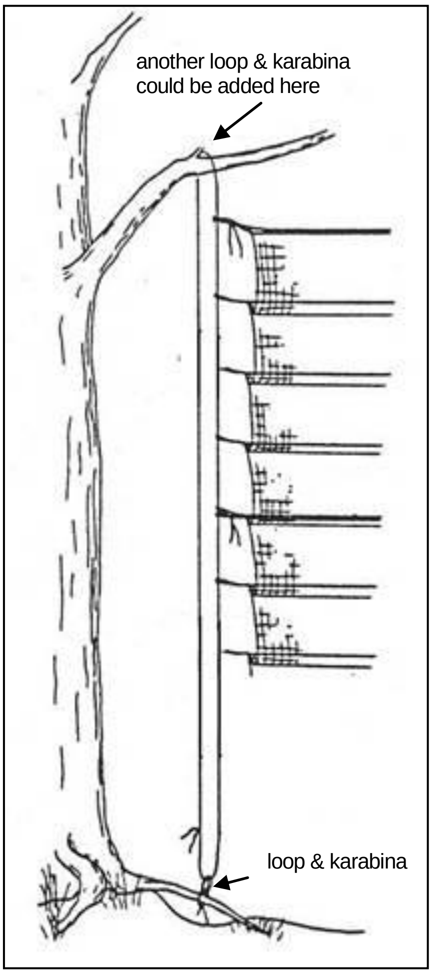


Figure 21. A simple mist net rig using string instead of poles.

The problem of having no supporting branches in the right places can be overcome by suspending the vertical endless loops from a horizontal top rope. Karabinas, metal rings or small pulleys are attached to the top rope at the required net-length apart. This method is outlined in detail in Dilks et al. (1995) and Kunz & Kurta (1988) (Fig. 22).

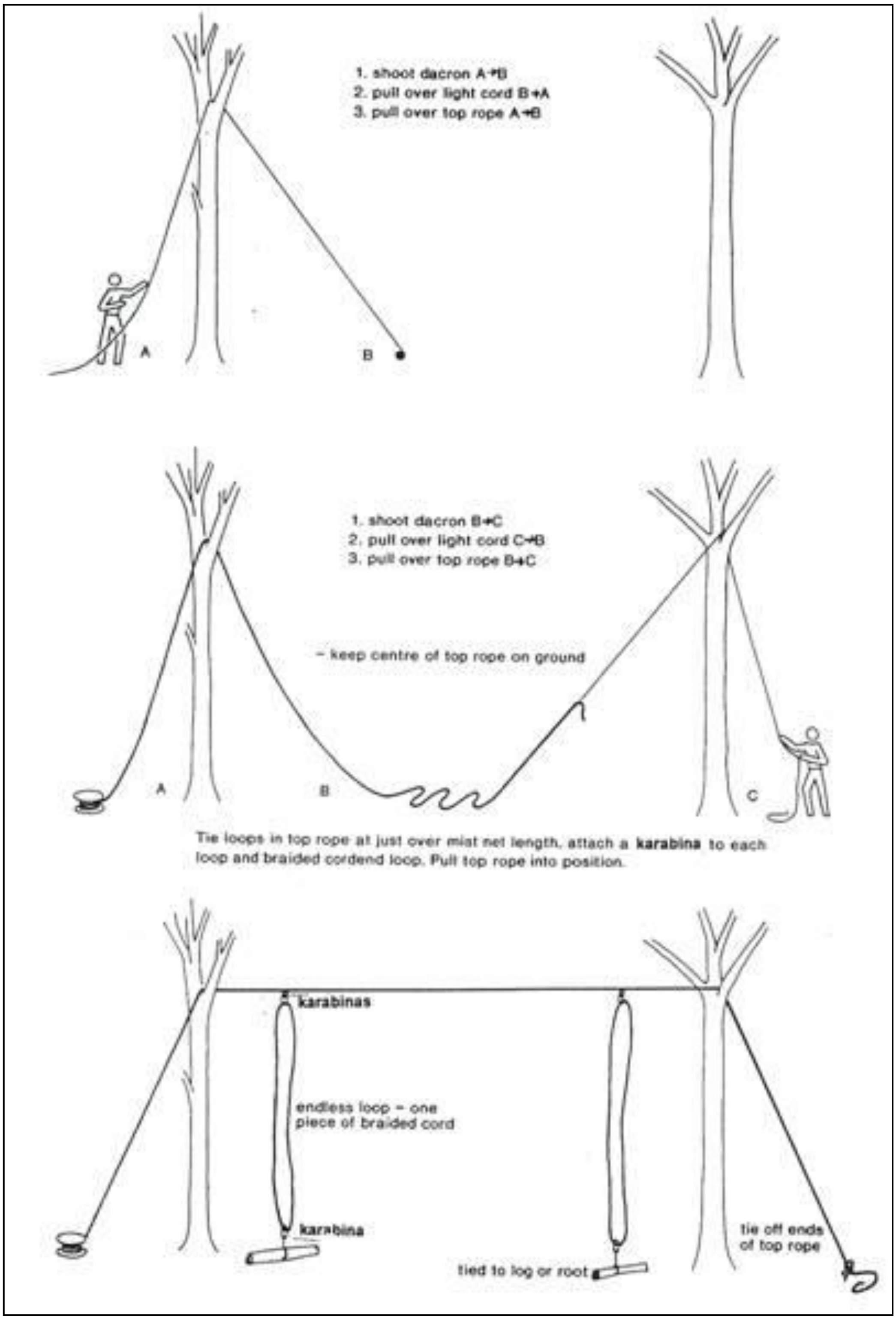


Figure 22. Steps for erecting a canopy rig (Dilks et al. 1995).

If nets are to be stacked one on top of the other they need to be joined so there are no gaps between them. Gaps can be prevented by over-lapping net attachment by one pocket. However, this will create a double layer of mesh which will be easier for bats to detect, and if caught, will result in the bats becoming more tangled. Nets can be tied together with short lengths of sewing thread or string, wire bread-bag ties, or even grass (Fig. 23). Alternatively, for a more permanent solution a number of nets can be sewn together. This technique is useful if sites are netted regularly using the same set-up and number of nets. It is recommended that plastic or metal snap-lock rings (e.g. shower curtain rings) are used for attaching stacked nets to cord pulley systems (Fig. 23).

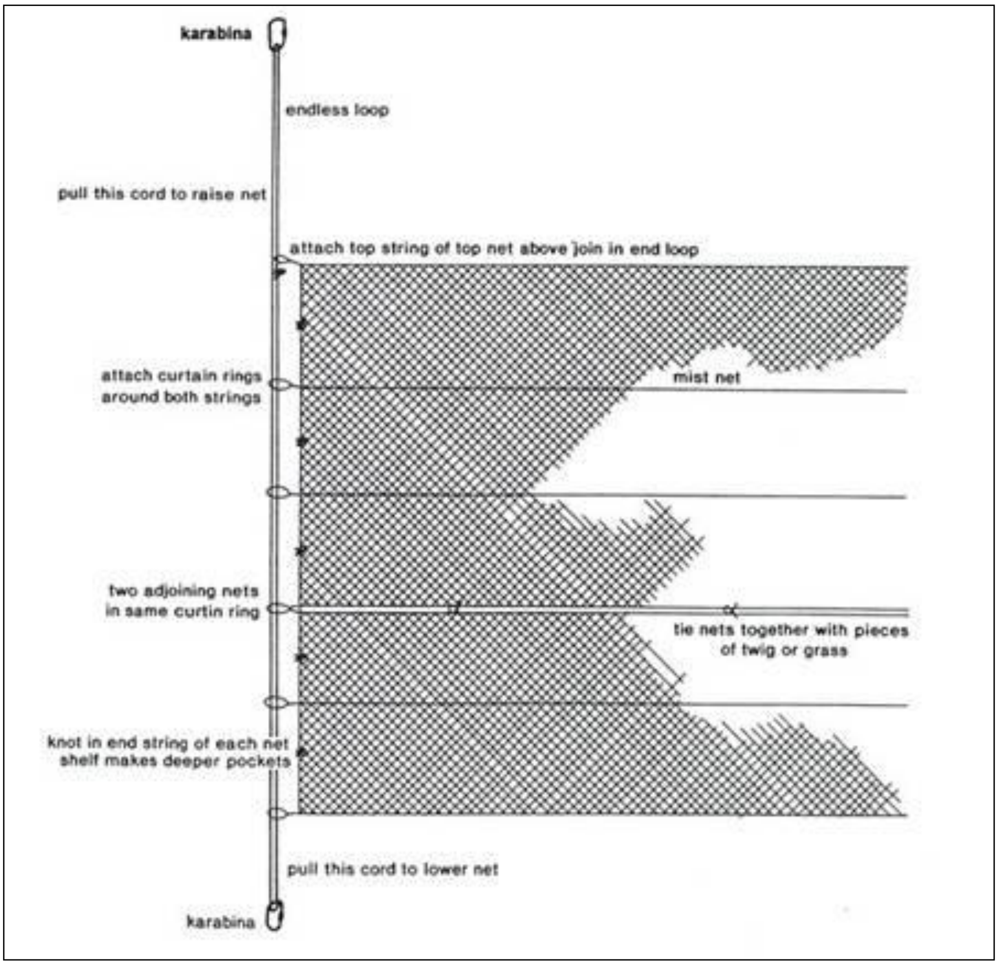


Figure 23. Method for attaching nets to cord pulley system (Dilks et al. 1995).

Monitoring nets

Nets **must** be monitored at all times. A good position for observation is sitting at one end of the net. The observer **should** minimise light and noise disturbance by sitting quietly in the dark and check the length of the net every few minutes with a torch. Using a bat detector will give an indication of how much bat activity there is, and how often to check the net. It is preferable for the detector to be turned down low, or to have an earphone fitted to minimise noise. It is possible to catch a bat in the net that is not heard on the detector, so detectors should not be relied on as the sole means of determining if a bat has been caught. Mist netting in areas of high bat activity **must** be undertaken by a minimum of two people. Two or more people are useful if more than one bat is caught in a net at a time, and for lowering higher nets. Bats **must** be removed from nets as soon as possible to prevent unnecessary stress on the bats, to reduce entanglement, and to prevent bats chewing large holes in the net.

Extracting bats

Before handling any bats it is important to read the sections on 'Handling, examining, measuring and releasing bats' and 'Health and safety'.

Removing bats from a mist net can be difficult and time consuming, requiring patience and skill. It only becomes easier with training and practice. Prior training in removing birds from mist nets is very useful, since the techniques are basically the same. Most people consider that removing bats is more difficult because bats can become far more entangled than birds. Bats can chew through the net and in their struggles can spin and bunch up the net, so it becomes difficult to find their entry point.

When a bat hits the net, it is important to grasp it as quickly and as carefully as possible so its struggles won't entangle it further or allow it to escape. It may be necessary to lower the net to reach the bat. Do not try and lower the net by pulling down on the net close to the bat. Lower the net from each end, and avoid lowering the bat so it rests on other net pockets or on the ground and becomes further entangled. Lesser short-tailed bats can become aggressive when tangled in nets and may bite (sometimes drawing blood) when handled. Although it is a requirement that gloves **must** be worn when handling lesser short-tailed bats, it is almost impossible to remove bats from a fine mist net wearing a pair of gloves. As an alternative, one glove (or a bat bag) could be worn on the hand used to restrain the bat, and the other hand could be left bare to manipulate the net (Fig. 24).

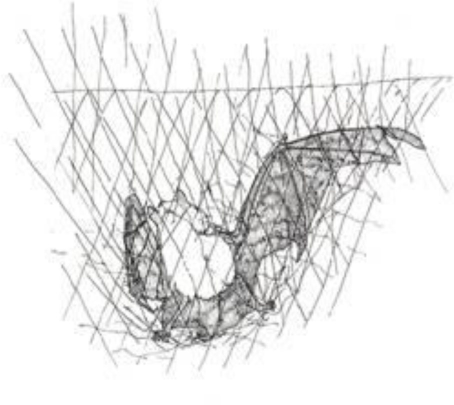

C.F.J. O'Donnell

Figure 24. Using a bag to restrain a lesser short-tailed bat in a mist net.

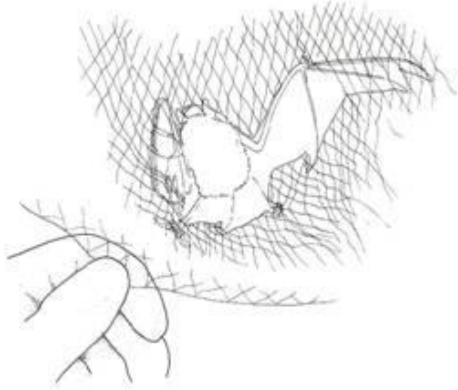
The first thing to do before attempting to remove the bat is to determine which side of the net the bat flew in. Open the pocket and look for parts of the bat that are not covered in netting. It is usually easiest to start with the least tangled part of the bat.

Generally it is easiest to clear a bat in the following order: firstly, gently tease the net away from its feet and tail, and then ease the bat away from the net and into the hand working the net away from the stomach and body. Next, extract one wing at a time. Sometimes the whole wing can pass through the mesh of the net, and the strands have to be lifted over the forearm and thumb without straining the delicate finger bones. Finally, remove the head, checking carefully that there is no netting caught in the bat's teeth. Six basic steps to remove bat from a net are illustrated in Fig. 25.

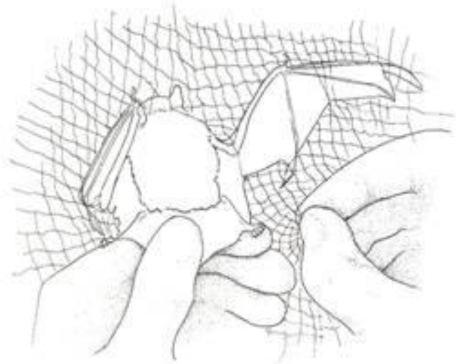
Two ‘real life’ examples of removing lesser short-tailed bats from nets are shown in Fig. 26. A short video of taking a long-tailed bat out of a mist net is also available (see ‘Extracting a long-tailed bat from a mist net’—docdm-22907).



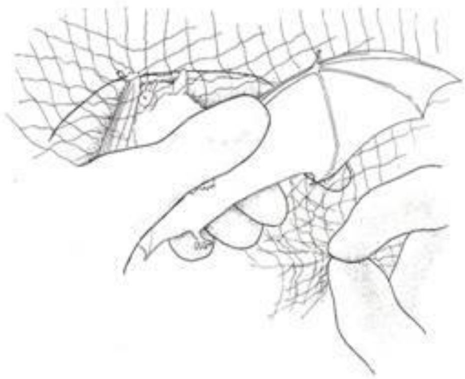
1 In the net.



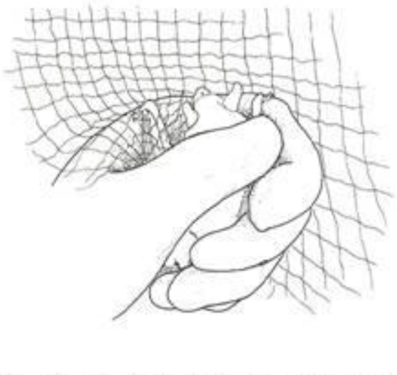
2 Opening the bag.



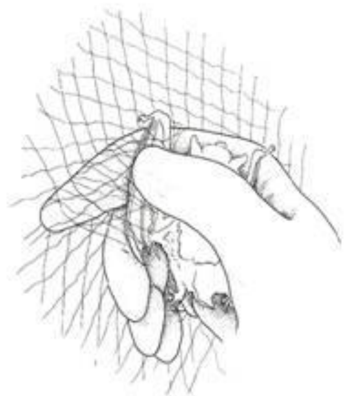
3 First clear the feet and tail.



4 Hold the body firmly and free the extended or less-tangled wing.



5 Next free the head. At this stage the bat must be held firmly to avoid re-entanglement. Once the head is free, change the grip to restrain the movement of the head and prevent the bat biting.



6 Wings may need to be opened to disentangle them.

T.P. McOwat, reproduced with permission, *Bat Workers’ Manual* © JNCC 2004. ISBN 1861075588

Figure 25. Six basic steps to remove a bats from mist nets.

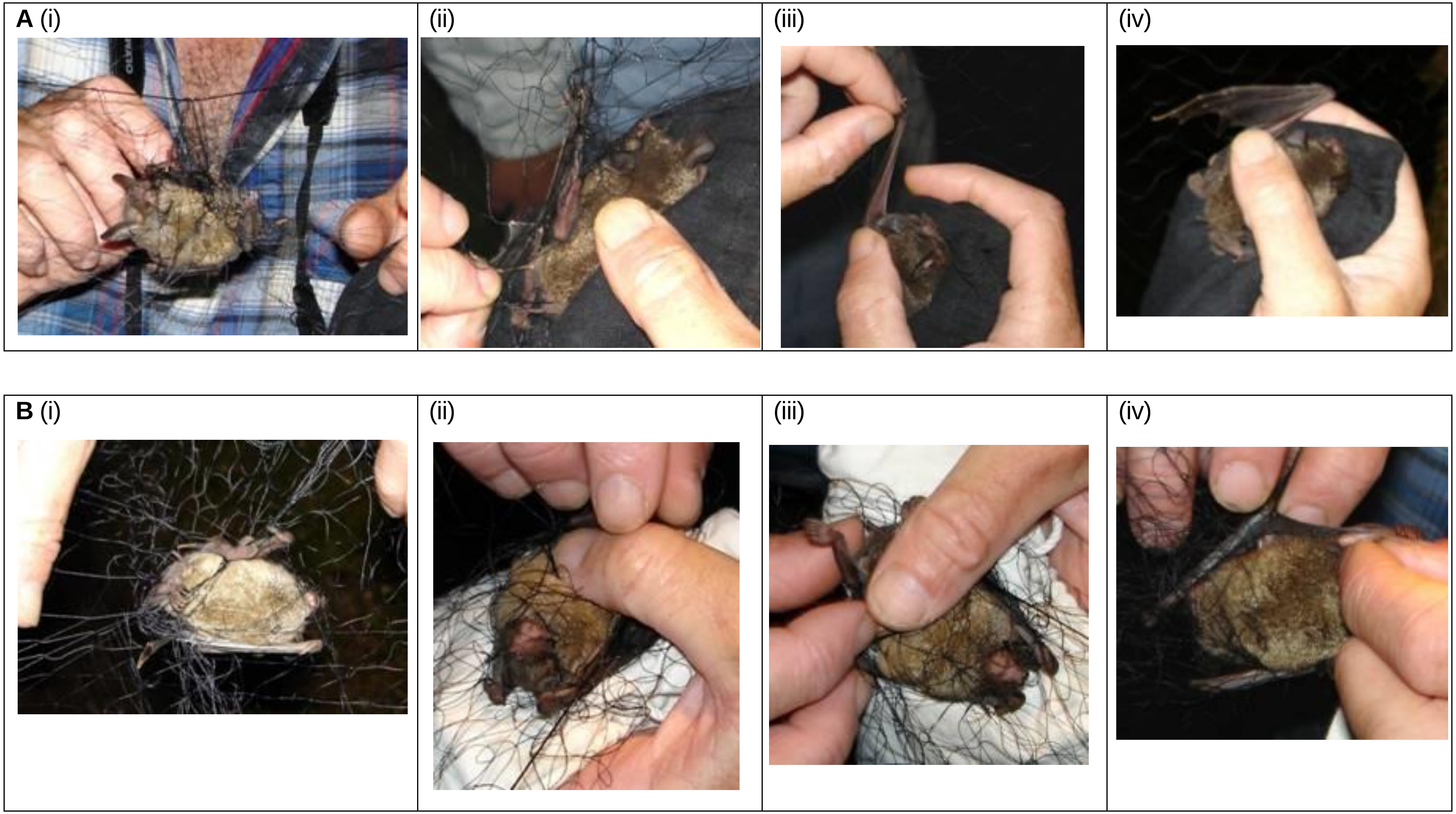


Figure 26. Two examples of how to remove a lesser short-tailed bat from a mist net. **A**: (i) In this sequence the belly of the bat is tangled but its back is fairly free. (ii) The bat is secured in the bag and its feet disentangled. (iii) Its forearm is freed by gently easing the net over its wrist and thumb, finally (iv) half opening the wing to check if there are any more net strands looped around it. **B**: (i) In this sequence the belly of the bat is free. (ii) The bat is secured in the bag, and (iii) its feet are worked free. (v) Lastly, the net is gently pulled over the bats wings and head.

Occasionally bats become so badly entangled they cannot be freed quickly. In these situations the net **should** be cut using fine scissors or a sewing ‘unpicker’ so the bat is not further stressed. Sometimes it is only necessary to cut a few strands to assist the usual process of manipulation and extraction. However, if a bat appears to be distressed it **must** be removed as quickly as possible. Captured bats should be transferred into a soft cloth holding bag (see ‘Guidelines for temporarily keeping bats in captivity for research purposes’).

Nets **must** be checked thoroughly for bats before they are furled or dismantled. Bats can look very small when in a net, especially if they are at the top of a high rig, and can be mistaken for leaves. Twigs, leaves, insects, etc. **should** be routinely removed from nets, especially before nets are furled or folded away.

Differences in capture success rates between species

Long-tailed bats

Capture rates of long-tailed bats in mist nets are often very low, even in areas of high activity (e.g. < 0.01 bats/net-hour in the Eglinton Valley, O’Donnell & Sedgeley 1999). Long-tailed bats can easily detect and fly through small holes in nets. Capture rates can be improved by setting nets close to foraging areas such as ponds, and by using high mist net rigs of 3–10 nets tall. Nets set for long-tailed bats do not need to touch the ground. The highest capture rates for long-tailed bats in the Eglinton Valley occur in February when young bats begin to fly for the first time. These bats are relatively poor fliers, and are less adept at avoiding nets.

Lesser short-tailed bats

In areas of reasonably high bat activity lesser short-tailed bats can sometimes be caught within minutes after opening nets. Lesser short-tailed bats do not seem to be able to detect or avoid nets in the same way as long-tailed bats. Lesser short-tailed bats spend a proportion of the time foraging on the ground, sometimes flying at heights of less than 1 m. To catch this species the bottom of the net should be close to or touching the ground, and it is seldom necessary to set nets more than two nets high (Fig. 27).

C.F.J. O’Donnell

Figure 27. Lesser short-tailed bat caught in a mist net at ground level.

Harp traps

Description

Harp traps are specialised traps developed in America and Australia specifically for catching bats (Constantine 1958; Tuttle 1974; Tidemann & Woodside, 1978). Harp traps typically consist of a 2 m × 2 m square frame of metal tubing that supports two banks of vertically strung monofilament fishing line. A canvas collecting bag is attached beneath the frame (Fig. 28). Bats fly into the lines, slide down them and land in the bag. The trap works on the principle that the banks of fine lines confuse the echolocation calls of bats. The bats may be able to fly through the first set of lines, but find it difficult to avoid the second set. The lines are tensioned so that bats do not become entangled. The collecting bags are usually lined with polythene/plastic which is attached at the top and extends downwards for about ½ to ¾ of the height of the bag. The polythene is slippery and prevents bats from climbing out, but allows them to crawl up the canvas bag beneath the polythene (Fig. 29). Harp traps are usually placed on the ground, but can be suspended above the ground and used in a variety of situations.

It is possible to construct a harp trap using guidelines found in Tidemann & Woodside (1978), but commercially produced harp traps are available from Australia (by Faunatech Ltd). The standard commercially available traps are 4.2 m^2 in size, and have two banks of fishing line. Smaller traps, and three- or four-bank traps can be made to order.

Figure 28. Harp trap.

Figure 29. Bats in harp trap bags.

Advantages:

- Harp traps are reasonably portable over short distances, and are easy to set up.

- They do not require constant supervision, so it is possible to run several traps at once and throughout the night.
- Harp traps can be used judiciously in front of tree roosts or in cave entrances to catch a large number of bats in a short time.
- Bats can always be removed quickly and easily from the trap collecting bag, and appear to be less stressed than bats that get tangled in mist nets. Bats often roost quietly in the bag rather than struggling to escape.
- The catching part of the trap (the two banks of fishing line) is easily replaceable, and relatively easy to repair.

Disadvantages:

- Harp traps have a relatively small catching area compared with mist nets.
- They can be heavy to carry over long distances, particularly if multiple traps are required in an area where there is no vehicle access.
- Commercially available harp traps are more expensive than mist nets. At the time of writing Australian made harp traps cost approx. AU$1,400 (including accessories—re-stringing kit, carrying bag) plus AU$235 for shipping and insurance.
- Lesser short-tailed bats have been observed climbing out of trap bags.

Setting up harp traps

Assembly

Harp traps can be easily assembled by two people in a few minutes, and one person with practice. However, the commercially available traps which are made up of several different parts can be confusing to assemble for the first time. Fortunately all of the traps purchased from the Australian company Faunatech come with a very detailed information manual that includes instructions for assembly and maintenance. If you borrow a trap, try and ensure you borrow a manual too.

Once the trap is assembled it is important to ensure the strings are tensioned correctly. The trap should be adjusted so the fishing lines are firm when pressed with an open hand. If lines are too loose bats might get tangled, and if too tight they might bounce off. Remove and replace any broken lines. The collecting bag also has to be adjusted. After it is hung onto the trap, the end ties of the bag should be tied firmly around the hip-mounts or the legs of the trap. The aim is to prevent the bag being loose and saggy, which will allow bats to escape. The bag should form a narrow ‘V’ shape; however, if the bag is tied off too tightly it reduces the angle of the capture zone, and bats may not slide down into the bag.

The commercial traps come with four telescoping legs, so the traps can be placed on the ground and easily adjusted to the desired height.

Suspending a trap from objects

Harp traps can be suspended above the ground in a variety of situations such as in cave entrances, above rivers, under bridges, and outside roost trees (Fig. 30). Ropes can be attached for hoisting and lowering the trap.


Figure 30. Harp trap suspended outside long-tailed bat roosts in a tree and in a rock crevice.

It is best to avoid a centrally located lifting rope (as originally illustrated in Sedgeley & O'Donnell 1996) because it may warp the frame and affect the line tensioning. Instead, a bridle or a bracket **should** be attached to the top of the trap, and the centrally spaced lifting rope attached to the bridle (Figs 31 & 32). It is also necessary to secure the top and bottom of the trap to prevent the trap coming apart. This can be achieved by firmly tying lines from the top of the trap (the line-carrier heads) to the hip mounts at the bottom (where the legs are normally fitted) (Fig. 33). Guy ropes should be attached to the trap to aid in positioning the trap once it is in the air, to secure the trap in its final positions, and to assist in the lowering process.

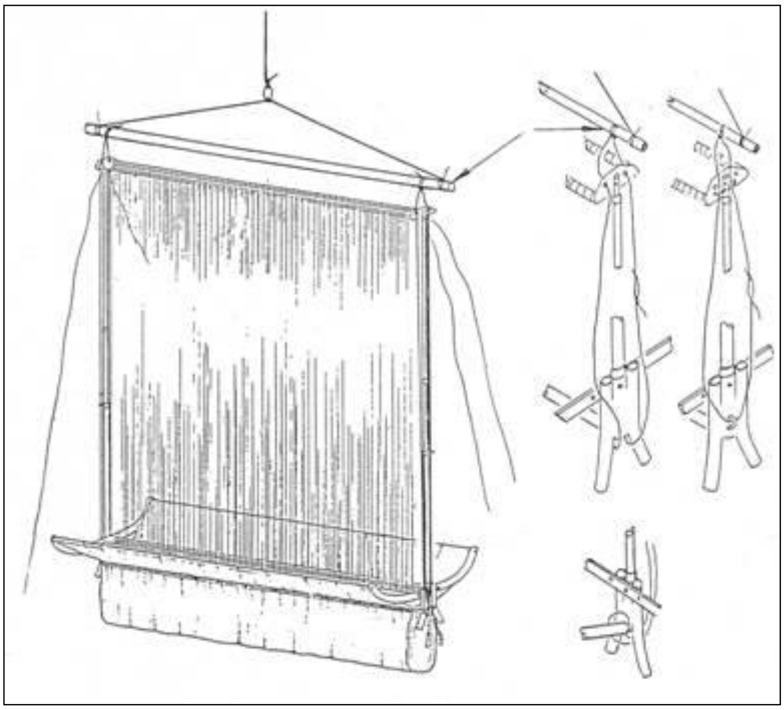


Figure 31. Attaching bridle to suspend harp trap. Illustration from harp trap instruction manual.

W. Simpson

J.A. Sedgeley

Figure 32. Bracket for suspending harp trap.

Figure 33. Securing sides of trap.

Monitoring traps

Harp traps do not require continuous monitoring because after the bats hit the fishing line, they fall into and are held by the collecting bag. Harp traps positioned on the ground can be set up at dusk and checked at dawn, providing bats are released before it is too light. However, it is preferable to check the traps several times during the night so bats are not kept for unnecessarily long periods of time. In Australia there have been records of bats in harp traps being preyed upon by mammals and snakes. Predation at traps left unattended for long periods has not been recorded in New Zealand, but is a potential problem since both possums and morepork have been observed visiting traps.

During the breeding season traps **must** be checked several times each night, to enable captured lactating females to be released to return to their young as soon as possible. Long-tailed bats give birth from mid-November to mid-December, and lesser short-tailed bats from mid-December to mid-January. Young begin to fly at 5–6 weeks old (O’Donnell 2001; Lloyd 2001). Harp traps placed outside roosts **must** be monitored continuously from dusk onwards.

Extracting bats

Before handling any bats it is important to read the sections on ‘Handling, examining, measuring and releasing bats’ and ‘Health and safety’.

After bats fall into the collecting bag there is usually an initial period where the bats flap or run around the bottom of the bag. Eventually, the bats tend to crawl under the plastic and move upwards towards the top of the bag. Once they reach the top they usually settle down to roost, and if several bats are present they will often form roosting clusters. Bats can easily be lifted out from the bottom of the bag or from beneath the plastic and placed in cloth holding bags.

Sometimes if there are only one or two bats in the trap, or if conditions are fairly cold bats will go into torpor. This usually means the bats will simply be very sluggish and slow moving when they are handled. However, very occasionally long-tailed bats that are coming out of torpor have been observed to extend their wings, open their mouths and sometimes squeak loudly. This behaviour may be a form of aggressive display designed to scare off predators while the bat is trying to warm up sufficiently to fly away. If this behaviour occurs it may be necessary to gently fold the bats wings to its sides to enable it to be transferred into a holding bag.

Differences in capture success rates between species

Several studies with infrared cameras show that bats can detect and avoid harp traps, and this ability varies among different species (reviewed in Gration 2002). There are no studies comparing differences in capture rates between long-tailed bats and lesser short-tailed bats.

Long-tailed bats

Long-tailed bats have been caught successfully in the range of locations and situations.

Lesser short-tailed bats

Free-standing harp-traps do not appear to be as successful at catching lesser short-tailed bats, even when set in areas of high activity. For example, traps set in roosting areas on Codfish Island never caught a bat, and have only caught bats on several occasions in the Eglinton Valley. Lesser short-tailed bats may not necessarily be better at avoiding traps; rather, they may be better at escaping from them. Lesser short-tailed bats seem to be able to jump or climb out of the collecting bags. Several individuals were observed climbing out of a trap set up outside a roost tree on Codfish Island.

Maximising capture rates in foraging areas and flight paths

Bats can frequently detect and avoid harp traps and mist nets. There are several ways of increasing the probability of catching bats.

Where should harp traps be placed?

All types of traps will be more effective if they are set in areas of high bat activity such as on flight paths, or in favoured foraging areas. These areas can be identified by using automatic bat detector units to record nightly activity levels. Bats frequently use gaps in the forest and long-tailed bats in particular will fly along tracks and the forest edge. Nets and harp traps set across these gaps may be effective. In South Canterbury and in the Eglinton Valley nets and traps set across small pools and streams amongst trees where long-tailed bats frequently foraged proved to be effective (Fig. 34). More than one harp trap can be used to fill gaps. Care should be taken when trapping over water. Traps should be set low to the water to catch bats feeding on insects or drinking, but not so low that a bat caught in the bottom pocket of the net or in the collecting bag of the harp trap would hang in the water. Nets will sag over the course of a night and may have to be readjusted. It is important to be aware of rising water levels.

There is some thought that bats can come to learn the position of traps and therefore capture rates will decline once the element of 'surprise' has been lost. Rather than run traps in the same place continuously, we recommend traps are moved around, and locations are given 'rest' nights.

Positioning—camouflage and confusion

Mist nets placed in front of vegetation are less likely to be detected by bats. Whilst this is a good method, it is important to consider that if wind direction or wind strength changes the net may be blown into this camouflage and become tangled. Harp traps and mist nets can also be positioned so vegetation 'funnels' the bat towards the trap/net, e.g. at the end of an enclosed track, beneath a large branch over-hanging water, or between a clump of trees and the bush edge. Harp traps work well when placed in small gaps in the vegetation. Spaces around the traps can be filled with branches, vegetation or even shade cloth material (Fig. 34a).

(a) J.A. Sedgeley

(b) J.A. Sedgeley


Figure 34. Harp traps set in long-tailed bat foraging areas. (a) Pond used by foraging long-tailed bats in South Canterbury. (b) Old forest track in Eglinton Valley.

Configuration of two or more nets, or a combination of nets and a harp trap, may confuse bats' echolocation. Commonly used configurations include a V pattern, perhaps with a harp trap (see 'Harp-trapping at roosts' below) positioned between the nets at the base of the V, a T pattern or a Z pattern. A net placed diagonally across a track may be less obvious to a bat than one placed at right angles (Kunz & Kurta 1988; Reardon & Flavel 1987; Churchill 1998).

Lures

It has been suggested that bats can be lured into a net by flicking a small pebble upwards into in the air in front of the net as the bat flies overhead. If the pebble is well-aimed the bat will, in theory, swoop down to investigate and may get caught in the net. We know of no examples of this working in New Zealand. Playing back acoustic calls has been used to increase capture rates in bat species in the UK (Hill & Greenaway 2005). Several researchers in New Zealand have used Audubon bird squeakers to lure lesser short-tailed bats into nets. The theory is that the squeaker produces a call which imitates lesser short-tailed bat singing. The use of squeakers may be more effective during the summer/autumn breeding season when singing activity peaks. The effectiveness of this technique, and whether it is biased towards males or females, is untested. The technique appeared to attract bats of both sexes into mist nets during summer months in the Eglinton Valley. The most effective method was to use the squeaker while sitting on the ground close to the net. The ideal position was near screening vegetation which helped camouflage the net and the person using the squeaker, and midway along the length of the net to maximise catching potential. Squeakers have not been successful at luring long-tailed bats; they generally scared the bats away.

Trapping at roost sites

For some research studies and management projects it is necessary to catch a large number of bats; for example, mark-recapture studies examining population size, productivity and survival; and translocation projects. The most efficient way to catch a large number of bats is to trap in roosting areas and directly at roost sites. However, trapping in these areas constitutes a much higher level of disturbance to bats than trapping in foraging areas, and can potentially cause bats to abandon their roosts. Several studies/management projects have involved catching long-tailed bats and lesser short-tailed bats as they emerge from their roosts (e.g. Sedgeley & O'Donnell 1996; Sedgeley & Anderson 2000; Lloyd & McQueen 2002; O'Donnell 2002). None of these studies recorded bats abandoning their roosts, but this does not mean bats may have been affected in other, less observable ways. Trapping at roost sites must only be undertaken if there is valid reason for doing so.

Mist netting at roosts

Nets **must not** be set directly (less than 5 m) outside occupied communal roost trees. There is the potential to catch many bats in a very short period of time (lesser short-tailed bat roosts may be occupied by thousands of bats). It is extremely difficult to quickly extract large numbers of bats that are in a net at one time. Bats can become very tangled, and removal can be very time consuming. Bats that remain in a net for a long period become distressed and can chew nets, and sometimes will bite themselves and other bats. The sounds of bats squeaking in the net will also draw in others. If a net is inadvertently placed in a position where too many bats are being caught, a soft cover such as a sheet, or a tent fly, could be thrown over the net to prevent further captures. The net should not be laid on the ground because this is likely to increase the degree of entanglement of bats (and damage the net).

Placement of nets farther away but still in the vicinity of a roost can work well. Lesser short-tailed bats were caught successfully in nets placed 10–100 m from an occupied roost in Rangataua Conservation Area (Lloyd & McQueen 2002), and 200 m from roosts in the Eglinton Valley (O'Donnell et al. 1999). A large mist net rig (10 nets high) has been used in front of the main entrance of a long-tailed bat roost in Grand Canyon Cave. The mist nets did not completely block off the cave entrance, and could be quickly lowered by a pulley system. However, bats at Grand Canyon Cave easily detected and avoided the nets, and capture rates were very low.

Harp-trapping at roosts

Caves

Harp traps can be very effective when placed in confined spaces such as cave or mine entrances. At Grand Canyon Cave (a large tunnel-shaped cave with two entrances), four traps were used in the larger main north entrance. Two were stood on a large mound of earth, and two were suspended above them (Fig. 35). A line of four standing traps were used to cover the smaller southern entrance (Fig. 36). Despite their much smaller capture area, the harp traps in the northern entrance captured many more bats than the large mist net rig (O'Donnell 2002).

(a) J.A. Sedgeley

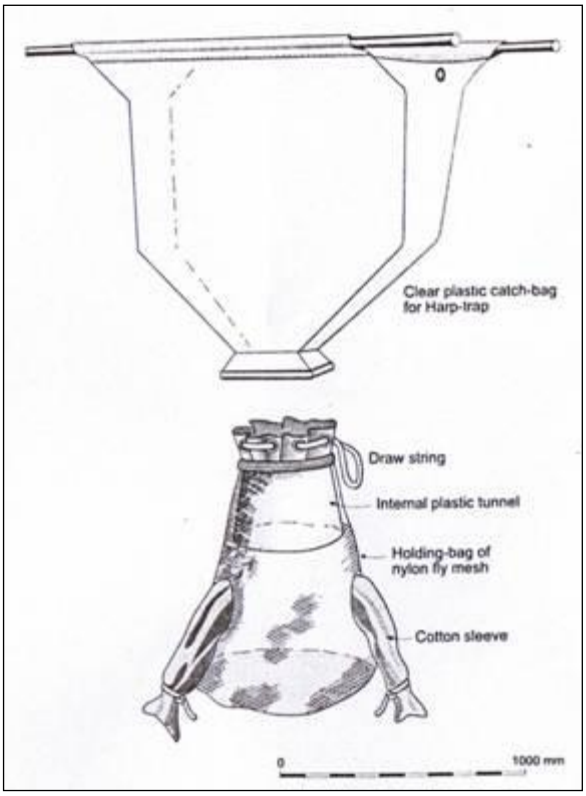
(b) J.A. Sedgeley

Figure 35. Two traps suspended in main (north) entrance of Grand Canyon Cave. Views from (a) inside, and (b) outside.

Figure 36. Four harp traps set at rear (south) entrance to Grand Canyon Cave.

Tree roosts

Trapping directly outside of tree roosts can be very effective with correct positioning. Harp traps have been used to catch up to 100% of long-tailed bats emerging from trees (including maternity roosts) in the Eglinton Valley, and did not appear to affect roosting behaviour in these bats, or cause them to abandon their young (Sedgeley & O'Donnell 1996). Harp traps have also been used successfully outside lesser short-tailed bat roosts in Rangataua Forest, Eglinton Valley and on Codfish Island (Sedgeley & Anderson 2000; Lloyd & McQueen 2002). However, these traps were not used at lesser short-tailed bat maternity roosts, and only a relatively small proportion of the total number of bats using the roost was captured. Large numbers of bats inside a collecting bag at one time may be unduly stressful for the bats, so traps must be monitored continuously outside roost sites. The Eglinton bat studies aimed to catch no more than 100 bats in a trap at one time. A small infrared video camera mounted on the trap bag was useful for counting how many bats fell into the trap. In the Rangataua Forest a specialised bag was used to allow ongoing removal of bats and continuous trapping (Lloyd & McQueen 2002) (Fig. 37).



Lloyd & McQueen, *New Zealand Journal of Ecology*, 2002.

Figure 37. Specialised harp trap bag used to allow ongoing removal of bats during continuous trapping outside roost trees.

The optimum trap placement for catching long-tailed bats exiting a roost appears to be trap hung parallel to the roost with the edge of the collecting bag as close to the exit-hole as possible, often leaning onto the tree (Fig. 38). In this way bats often fall directly into the bag. If bats hit the lines near the top of the trap it is possible for them to recover and fly off before they fall into the bag. A trap placed perpendicular to a roost entrance was effective at catching lesser short-tailed bats as they returned to their roost site after foraging (Lloyd & McQueen 2002). Position of tree branches in relation to the roost hole can sometimes make it difficult to fit in a large-sized harp trap. Standard-sized commercial traps can be made smaller by shortening the sides of the trap and rolling up the line carriers. The line carriers secured into their new position (taped with insulating tape), and then the fishing lines have to be carefully re-tensioned and pushed back into position (Sedgeley & O’Donnell 1996). Alternatively a specially made small-sized trap can be used.


J.A. Sedgeley

Figure 38. Harp trap placed outside long-tailed bat roost. Arrow indicates the bottom of the roost entrance.

When to set nets and traps

Time of day

Nets should be open by dusk, but not too early, so capture of birds can be avoided. Long-tailed bats emerge from their roosts on average 2–30 minutes after sunset, but can emerge as early as 58 minutes before sunset. Lesser short-tailed bats emerge later, usually about 30 minutes after twilight (O'Donnell et al. 1999; Lloyd 2001, O'Donnell 2001). Bat activity is generally highest in the first 2 hours after sunset, but bats can be caught at any time of night. Nets do not have to be dismantled during the daytime; however, they **must** be furled to prevent capture of birds. To do this, walk mid-way along the length of the net, grasp the net and spin it to create a tightly furled tube. The net **must** then be tied with tape or string at intervals along its length to prevent it spinning loose. If several nets are stacked, the nets need to be lowered and all nets gathered together before spinning (Kunz & Kurta 1988; Dilks et al. 1995). If nets are left set up for several days they will begin to sag, and often require readjusting.

Harp traps can be set at any time, and do not need to be dismantled during daytime because they are very unlikely to catch birds. They can easily be moved to one side or laid on the ground if there are track access issues. If harp traps are left set for several days it is good practice to bring in the collecting bags and dry them out if they become overly damp or wet.

Weather conditions

We do not recommend that mist netting be undertaken in windy or rainy conditions. Bat activity is reduced during these conditions, and bats can easily detect nets that are moving in the wind or covered in water droplets. Harp traps can be firmly guyed, so are not usually affected by wind. The polythene liners in the harp trap collecting bag will protect bats to some degree from drizzle or very light rain, but it is not good practice to leave bats inside cold and wet bags. If caught bats become wet and cold they may enter torpor and subsequently become difficult to release.

Seasonality

Bat activity is usually reduced during cold conditions and during winter months, and capture rates may be correspondingly low. For example, long-tailed bat activity levels in autumn, winter and spring are < 5% of summer levels (O'Donnell 2000). However, activity levels in relation to temperature do vary throughout the country. Activity levels of lesser short-tailed bats in the Eglinton Valley were very low during winter, whereas on Codfish Island bats were active to temperatures as low as −2°C (Daniel 1990; Sedgeley 2001). On Codfish Island 399 bats were caught in a very short period of time during winter using a combination of harp-trapping and mist netting (Sedgeley & Anderson 2000).

Other trapping methods

These methods have not been trialled in New Zealand, but are proven techniques for other bat species.

Hand nets

Simple hand nets, the type that can be obtained from entomological suppliers (butterfly or dragonfly nets), are commonly used to capture bats at roost sites overseas (Kunz & Kurta 1988; Churchill 1998; Finnemore & Richardson 2004). Nets should be made of a fine mesh and need to be deep enough to prevent bats from escaping. Nets with an open mesh-like mist netting material should not be used because bats become entangled very easily (Finnemore & Richardson 2004). Hand nets may have limited application in New Zealand because they seem most effective when used inside roosts such as mines, caves, and houses. However, they could be used as a type of bag or cone trap if held directly below a tree roost entrance hole (see 'Cone and bag traps' below). Hand nets could be used to catch bats in caves if they are roosting within reach. The net can be carefully placed over a bat before it flies. In places where bats are roosting high from the ground, nets with telescoping poles can be employed, and a thin stick may be used to touch the bat gently and cause it to fall into the net (Kunz & Kurta 1988; Finnemore & Richardson 2004). As soon as a bat falls into a net, the frame should be rotated so the bat cannot fly out. Hand nets should not be used to catch bats in free-flight. If the hoop of the net strikes a bat it can easily break bones and cause other injuries.

Cone and bag traps

These traps are mostly used for catching large numbers of bats in a short period of time, and work best at roost sites where bats are emerging from a small hole. Traps can be fairly simple, consisting of little more than a large cone made of plastic or nylon material with a collecting bag at the narrow end (Finnemore & Richardson 2004).

Trip lines

Trip lines are commonly used in Australia as a cheap and simple method for catching bats over water. Monofilament fishing line (about 3 kg breaking strain) is criss-crossed across a water body multiple times at heights of 10 to 15 cm (Churchill 1998) or 6 cm (Reardon & Flavel 1987). Bats collide with the lines when they fly down to the water to drink, and fall into the water. Many bats cannot fly off from the surface of the water, and swim to the edge before taking off in flight. A person waiting at the edge can pluck the bats out of the water by hand or with a hand net. Bats can swim surprisingly quickly.

General care and maintenance of nets and traps

This section discusses general maintenance; it is important to also read the section 'Animal health considerations'.

Mist nets

Mist nets and harp trap bags **should** be thoroughly cleaned of twigs, vegetation, bat droppings and insects, and should be carefully dried before storage. Twigs can be removed relatively easily by breaking them into small fragments. Nets should be stored in small cloth or plastic bags when not in use. Nets can often get ripped, have holes chewed in them by bats and have shelf-strings break. Holes in nets can be repaired using nylon thread or monofilament fishing line. They should also be disinfected (e.g. with TriGene) if used in different study areas.

Harp traps

Harp trap bags **should** be thoroughly cleaned of twigs, vegetation, bat droppings and insects, and should be carefully dried before storage. Harp trap lines can snap or lose tensioning and parts of the frame and legs can corrode. It is important to undertake regular repairs and maintenance so traps can be set up quickly and easily. Well maintained traps will improve capture rates and reduce possibilities for bats to become entangled. The harp trap manual provides detailed information on re-stringing, maintenance and repair.

Temporarily holding captured bats

This section describes methods for temporarily holding bats for short duration as part of the catching process. See 'Guidelines for temporarily keeping bats in captivity for research purposes' for information on holding bats for longer periods.

Bag design

The most common devices for temporarily holding bats are soft cloth (cotton) bags with either draw-strings or ties. Bags used for holding birds are fine, but **must** be washed (e.g. with TriGene) before using for bats (and again afterwards). Strands from fraying material can become tangled around bats claws and teeth. Unless bags have perfectly sewn seams with no frayed edges, turn them inside-out so the seams are on the outside. Bags made out of a light coloured material are best because it makes it easier to see bats inside, and the bags can easily be numbered with a permanent marker or tag pen. Bats can easily squeeze out of tiny spaces. Draw-strings or ties should be long enough to wrap around the bag at least once to secure the opening tightly. They should also be long enough to hang the bag up.

Holding captured bats

After bats are captured it may not be possible to process them (weighing, measuring, transmitter attachment, etc.) immediately. Bags containing bats **must** never be put on the ground where they might be accidentally trod on or sat on. Ideally bags **should** always be hung up in an allocated place. It is recommended all bags are numbered, and a record is kept of the number of bats in each bag, their age and sex, and time each was caught. Bats can be prioritised for processing if it is known exactly what type of bat is contained inside each bag. Bats should not be held in cloth bags for more than 1 hour, and it is preferable to release them as soon as possible.

A small number of bats of the same species can be put into a bag together, but long-tailed bats and lesser short-tailed bats **must** never be mixed together in the same bag. The two species have never been recorded roosting together under natural conditions, and differ in size and temperament. There may be animal health implications resulting from holding the two species together.

Animal health considerations

Little is known about potential disease risks in bats. However, when catching bats common sense should be used. General principles include:

- Disinfect or wash nets and harp trap bags between study areas to minimise possibility of transfer of parasites and diseases between sites.
- **If possible,** use different nets for birds and bats.
- Regularly empty bags of twigs, leaves, droppings, etc.
- Dry bags thoroughly in the sun, or wash them with Napisan, Virkon or TriGene when moving between different sites.
- **Don’t** use bags that have been used for holding birds unless washed first.

See ‘Health and safety’ section for more guidance.

Also see ‘Psittacine pox internal prevention and emergency response plan for DOC’ (olddm-32604) for further information on disease surveillance and hygiene precautions.

Additional equipment

A head-torch is essential for taking bats out of nets and bags. A spotlight or floodlight is useful for checking nets and is essential for tall mist-net rigs. It is very difficult to take a bat out of a net without the aid of a head-torch. Always have available some sharp fine scissors or an 'unpicker', and cloth bags to put captured bats into.

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Handling, examining, measuring and releasing bats

There are two important considerations to take into account when handling any live animal: (1) to avoid stress and injury to the animal, and (2) to avoid stress and injury to the handler. This section describes how to safely handle and release bats. It describes how to age, sex, and assess reproductive condition individual bats; and discusses differences between species. Techniques for taking a range of morphological measurements and biological samples are also described.

Handling

Justifications for handling bats

Before catching and handling bats it is necessary to ask the question: Do I really *need* to handle bats? **New Zealand bats are fully protected fauna, and it is illegal to catch, handle or keep them without appropriate permits and ethics approvals** (see 'Permitting, ethics approval and training'). Permission for catching and handling bats will only be given if there is a valid reason for doing so, since all catching and handling methods will disturb bats, and if used carelessly, will cause injury. Reasons for handling bats include:

- Species identification
- Marking
- Research purposes
- Management purposes (e.g. translocation)
- Public relations and educational purposes
- Rescuing injured and sick bats[8]

Safety precautions and the use of gloves

Bats overseas suffer from a number of diseases. Although there have been no records of transmission of these diseases to humans in New Zealand, handlers should avoid being bitten and take some precautions if bitten. Anyone planning to work on a project that will involve routine handling of bats **must** read the section 'Health and safety' for more details about the potential health risks of handling bats, particularly in relation to lyssavirus. Long-tailed bats rarely pierce the skin if they bite, but lesser short-tailed bats can give painful bites. Although bites may cause little injury, a reflex withdrawal of the hand by the handler may harm the bat. Gloves **should** be worn for handling lesser short-tailed bats, but gloves are often clumsy, particularly if they are thick. A good compromise is to wear a single relatively thin glove on the hand used to control the bat, and keep the hand used for manipulation purposes bare (e.g. for extracting bats from a mist net or taking measurements). Depending on what part of the bat is being measured it may be possible to keep the bat inside the bag and gently extrude the part to be measured.

[8] The full legal implications for handling and temporary holding native bats have not been tested. A permit may not be strictly necessary to help a sick or injured bat, and we do not wish to discourage people from helping them. However, bats **should** be handed in to either a vet or wildlife care group or DOC as soon as possible. Vets **should** be encouraged to report any bats coming into their care to DOC. We do not recommend that untrained people keep sick, injured or abandoned bats. These bats **should** be tended by those that are expert in such matters.

J.A. Sedgeley

C.F.J. O'Donnell

Figure 39. Gloves should be worn to handle lesser short-tailed bats. A bag or a glove can be used on one hand to help protect the handler from bites and the free hand left bare for manipulation.

Always wash hands after handling bats. If a bat bite punctures the skin, or a bat scratches a handler, basic hygiene precautions **must** be followed. Clean the affected area carefully with a sterile alcohol wipe or with a liquid antiseptic such as 'Savlon', or soap and water, as soon after the bite or scratch as possible. If wounds are cleaned carefully, the risk of any infection will be greatly reduced. **The risk of sick or dead bats carrying disease is higher, so obviously sick bats or dead bats must be handled with gloves.**

Recommended grips

New Zealand bats are fairly small and fragile, particularly the finger bones in their wings. Both species can be very wriggly in the hand, and can bite. Therefore, it is important to develop a technique to hold a bat safely to both prevent injury to the bat and to avoid getting bitten (Fig. 39). It is generally easiest for workers to hold the bat in the non-dominant hand and take measurements with the dominant hand. We generally recommend bats be held loosely in the palm or across the fingers of the hand, with the fingers curled around the body, and the thumb gently placed on its back or behind the head. The bat may be held with the head protruding between the thumb and forefinger, which can be used to keep the bats jaw shut (Racey 2004) (Fig. 40). Do not apply excessive pressure to the neck or back. This method appears to minimise stress caused to the bat. This grip is probably the best one to use when measuring forearms and for opening wings (Figs 39 & 41).

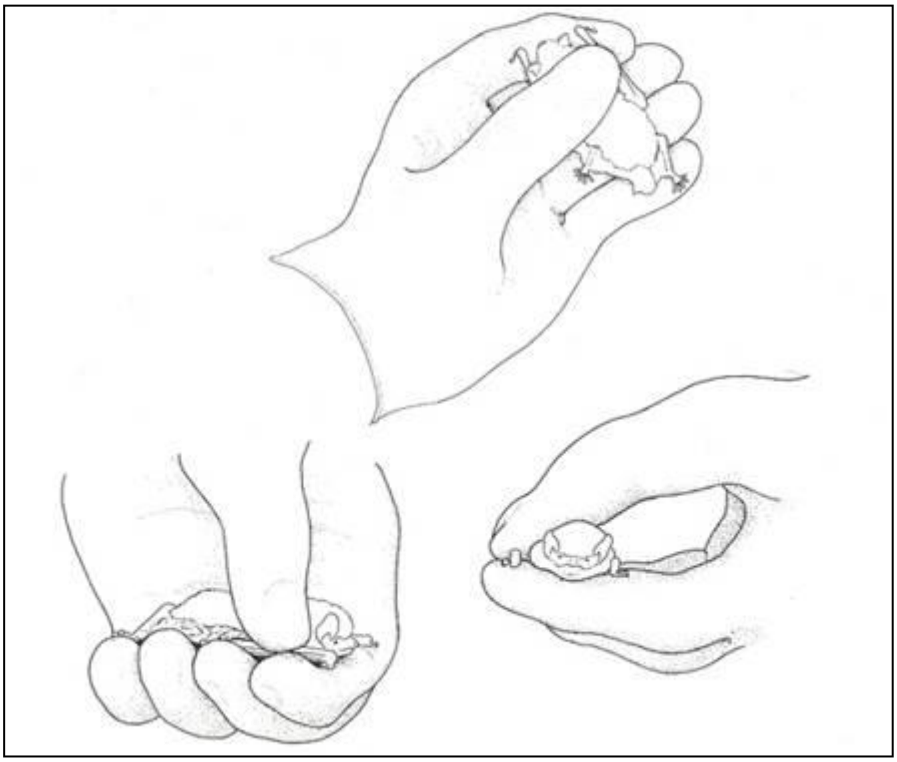

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Figure 40. Palm grip recommended for general handling and measuring purposes.

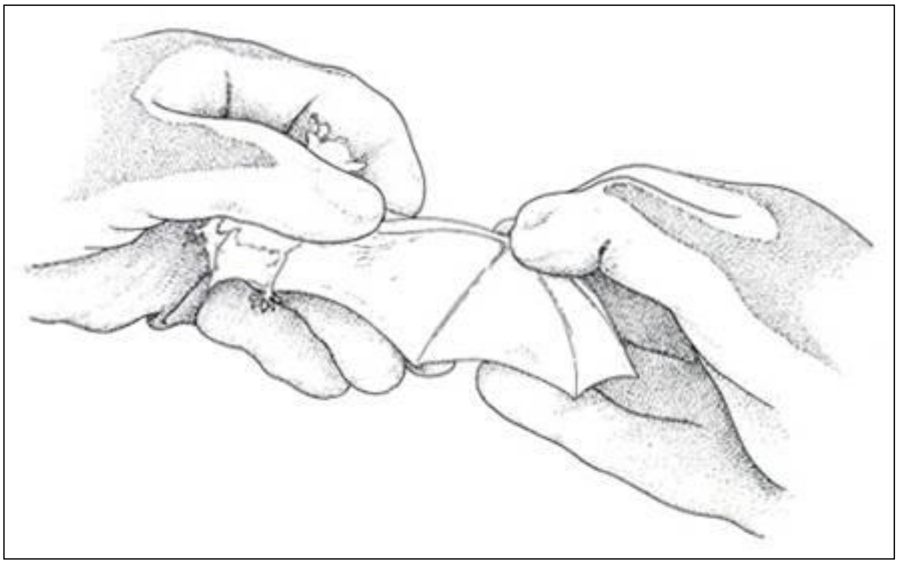

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Figure 41. Using the palm grip to examine a wing.

Photos: J.A. Sedgeley

Figure 42. Using the palm grip on New Zealand bats.

An alternative grip is commonly used in other countries for restraining larger bat species. This grip is also useful for holding a bat still for close examination or photography (Figs 42 & 43). However, this grip appears to be more stressful for the bat compared to the palm grip, and **should** only be used for specific purposes. Care **must** be taken not to strain the forearms and flight muscles.

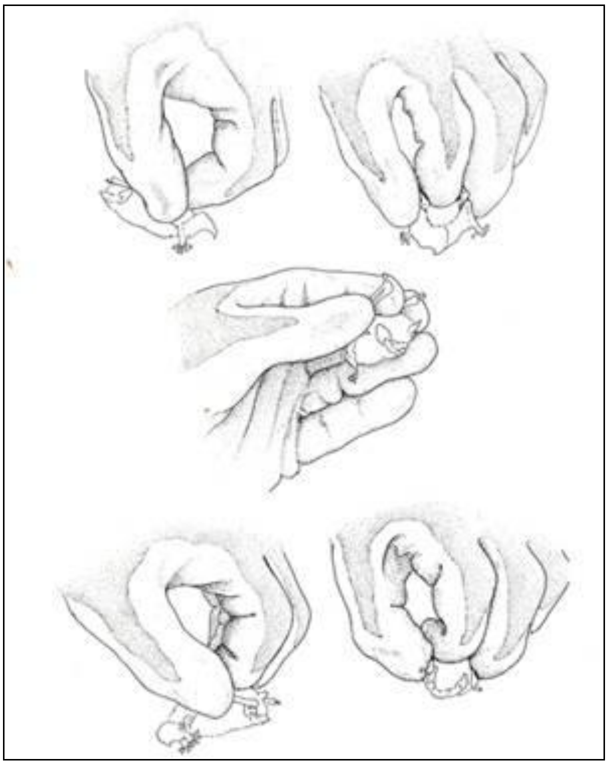

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Figure 43. Variations of an alternative grip recommended for holding a bat still for close examination. Care must be taken not to strain the forearms and flight muscles.

Handling time

Even experienced animal handlers may require practice to become both confident and competent in handling bats. It may be better for an inexperienced handler to do their measuring indoors (a room, a tent, a vehicle) to prevent bats escaping, particularly if crucial data is to be collected. However, if removal to 'indoors' is likely to increase handling and captivity time, it may be better to defer important data collection until the handler is more experienced. Accuracy in obtaining measurements is also likely to improve with practice.

Bats **must** be processed (identified, measured or marked) as soon as possible after catching. They can be held for a short time after capture in a cotton bag (see 'Catching bats'). Handling duration **must** be kept to a minimum while taking measurements. Prolonged handling can distress the bats. Lesser short-tailed bats appear to be more sensitive to prolonged and insensitive handling than long-tailed bats. Very occasionally lesser short-tailed bats will convulse during a lengthy handling period. If this happens, the handler **must** immediately cease examining or measuring the bat, place it somewhere quiet to recover, (e.g. back in a bag by itself) and then release it as soon as possible. Lactating female bats **must** be processed and released as quickly as possible because they may be feeding young**.**

What should we measure and why

Measurements are usually taken for two main reasons: (1) to aid in species identification, (2) as part of a larger project studying some aspect of bat morphology or biology. Detailed measurements are not necessary to distinguish between the two extant New Zealand bats species (lesser short-tailed bats and long-tailed bats). They have a number of highly characteristic external features such as fur colour, ear shape and tail shape that make them easy to distinguish visually in the hand. These characteristics are described in 'Species identification in the hand'.

However, there are other circumstances where the ability to take a variety of measurements is useful. For example, four exotic species of bats from two Microchiropteran families have arrived dead in New Zealand as stowaways in cargo several (Daniel & Yoshiyuki 1982; Daniel & Williams 1984; O'Donnell 1998), and there is the possibility of rare vagrants turning up. There is also the remote possibility that the greater short-tailed bat is still extant. In these situations, the ability to take standardised measurements will greatly aid in species identification.

Research projects usually involve taking a variety of standardised measurements. For example, a project might involve recording variation in bat size between populations, or recording changes in growth rates or body condition within a population. Alternatively, a project might aim to document basic aspects of life history such as the timing of breeding. Figure 44 shows the main features of a bat and the terms to describe them. Many of these terms are used in the sections below.

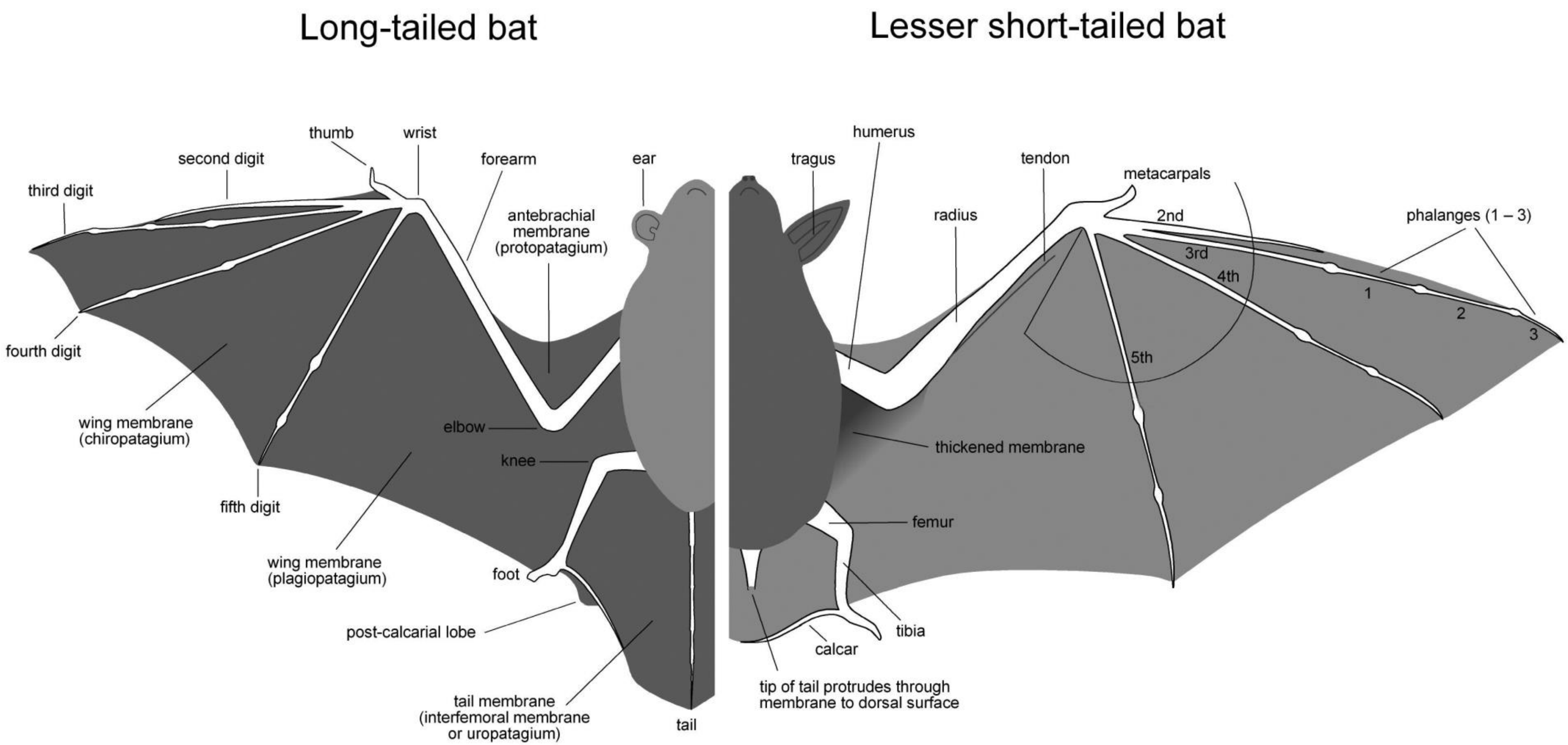

Figure 44. The main features of a bat.

Ageing bats

The most reliable and easiest way to age a bat is to examine the finger joints in its wing. There are other features to examine, but these can be much more subtle and difficult to observe. The terms baby or pup are often used to describe bats from birth to age of first flight. The term juvenile is used to describe the bats from the age of first flight until the joints in the finger bones become ossified/fused. The term sub-adult is used to describe young bats that have achieved fully adult skeletal development, but have not reached breeding condition/sexual maturity.

Wing joints

At the time of first flight, bats' bones are not completely ossified. This can be seen most clearly in the finger-bones. If held up to the light, the cartilaginous ends of the finger bones in babies and juveniles are apparent as pale bands either side of the joint. As the cartilage is replaced by bone, the joint becomes more rounded and knuckle-like (Fig. 45) (Hutson & Racey 2004). The joints usually appear fully ossified by the autumn which means this technique can only be used to reliably distinguish juveniles from adults and sub-adults for up to about 12 weeks after birth.

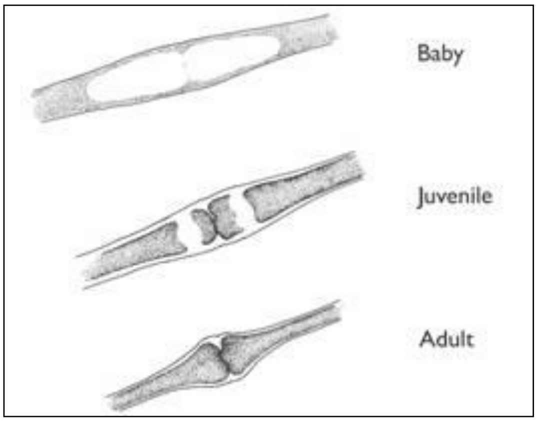

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Figure 45. Development of the joints of the finger bones. Joint development can be used to reliably age bats. Joints of baby bats and juveniles appear thin and tapered with paler bands. Joints of adults are more knobbly.

The easiest way to examine finger joints is to illuminate the wing from behind by holding the wing over a torch (Figs 46 & 47). A torch with a reasonable diameter head is best. It is important to be aware of how hot the torch gets. A spotlight, for example, could burn the bat's wing.

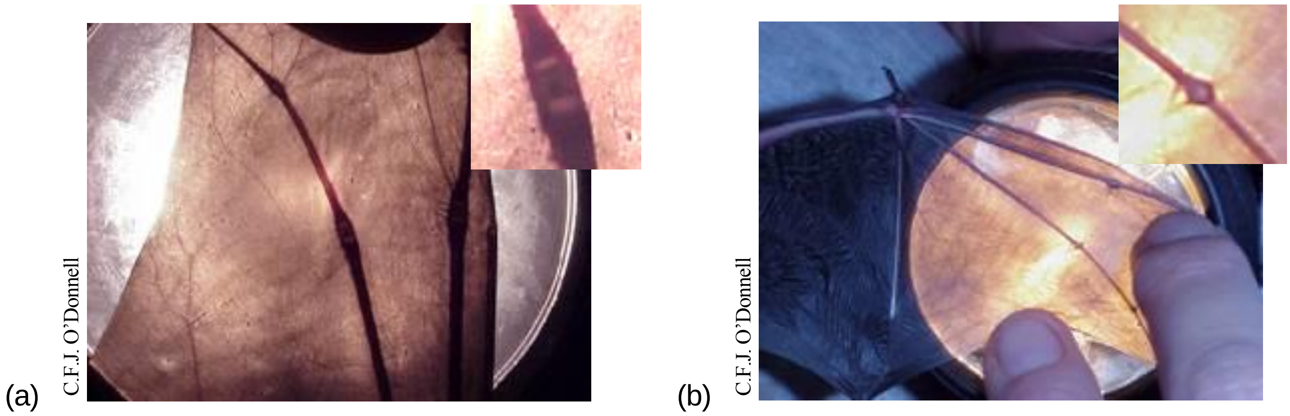


Figure 46. Technique for examining finger-joints in long-tailed bats illustrating differences between (a) adult, (b) juvenile.

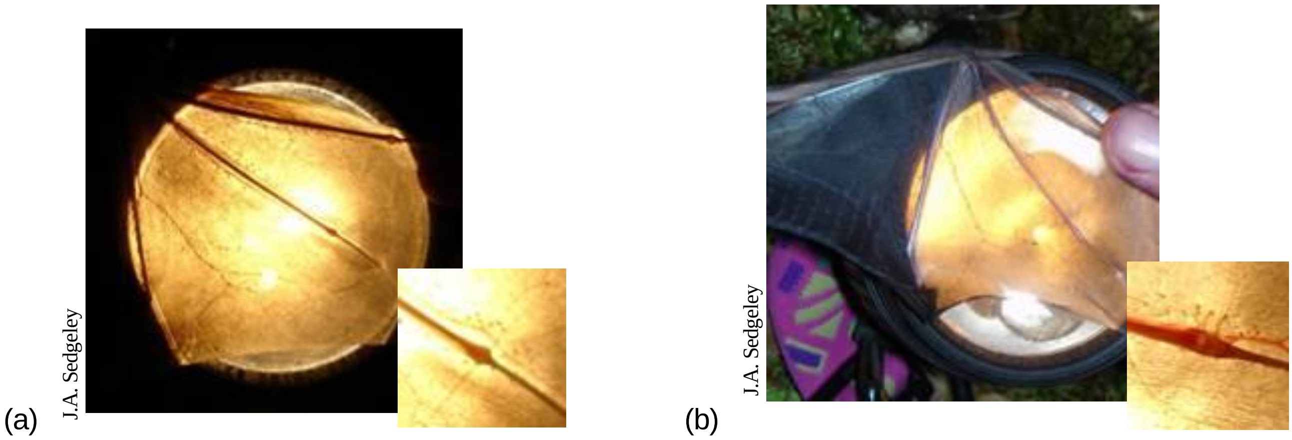


Figure 47. Technique for examining finger-joints in lesser short-tailed bats illustrating differences between (a) adult, (b) juvenile.

Other features

Juveniles will fly before they are fully grown and so the length of their forearms, wing-depth at fifth digit and body mass, at least in the early stages, are usually smaller than in adults (Anthony 1988) (see sections 'Wing depth at fifth digit (or fifth digit length)', 'Ear length and tragus measurements', and 'Wing measurements recorded for New Zealand bats' below). There are also subtle differences in the wing membranes of juveniles. The membranes are often darker in colour, are clean and unblemished and feel soft and almost sticky, although wings of adult lesser short-tailed bats can also feel sticky (Hutson & Racey 2004). Fur colour of juvenile long-tailed bats is usually darker than adult fur, but this is not always obvious. Juvenile bats often have a greater number of mites or bat flies than adult bats.

When to look for juveniles

Young begin flying at 5–6 weeks old. Young long-tailed bats have been recorded flying in South Canterbury in the second week of December and in Hawke's Bay on 6 January. In the Eglinton Valley, flying juveniles are caught from mid-January onwards, but date of first flight can vary by as much as 17 days annually (O'Donnell 2002; O'Donnell 2005). Lesser short-tailed bats give birth some time between mid-December and mid-January throughout New Zealand (Lloyd 2005), so flying young are unlikely to be caught until January and February.

Sexing and assessing reproductive condition

Sexing

Male bats have a conspicuous penis (Fig. 48). Female lesser short-tailed bats have a pronounced clitoral pad above the vagina, but it is smaller and more domed than a penis (Fig. 49). Females have a single anterior pair of mammary glands and nipples located c. 4 mm from the armpit. Nipples are relatively obvious in females of both species if they have recently given birth and are suckling young (Fig. 50). They are much less obvious once they begin to recede after lactation and in females that have never given birth (Fig. 51). The easiest way to examine the nipples of female bats is to hold the bat on its back, hold the wing open, and gently blow onto the bat to part its fur.

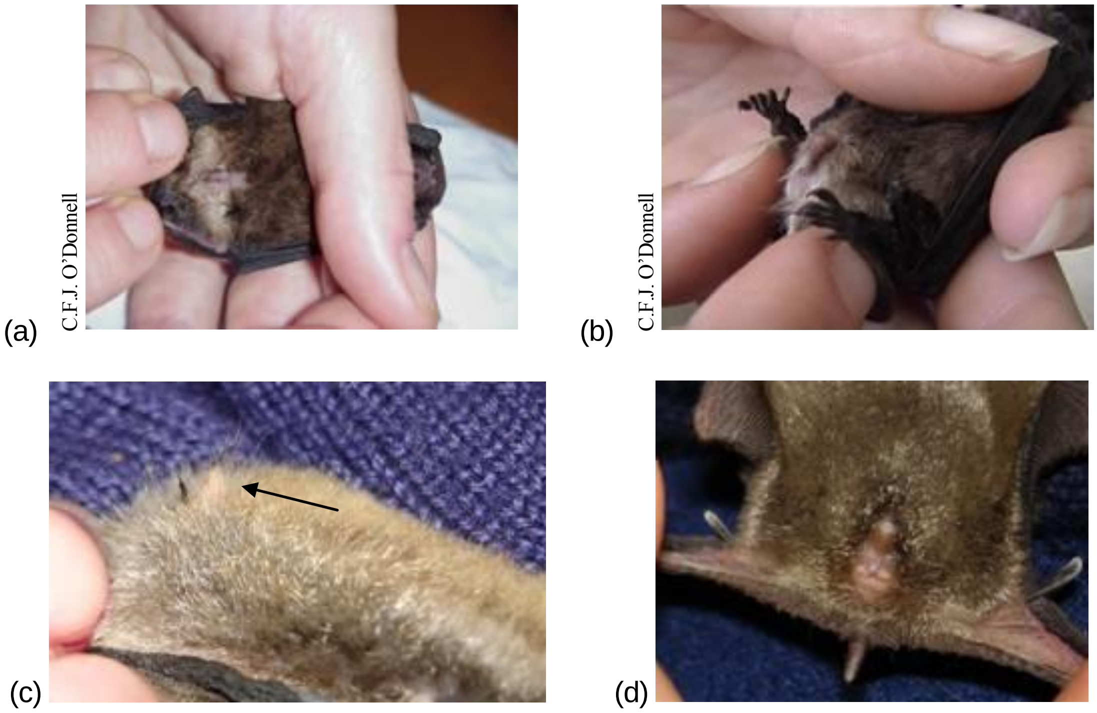


Figure 48. Genitalia of male bats (a) (b) long-tailed bats, (c) (d) lesser short-tailed bats.

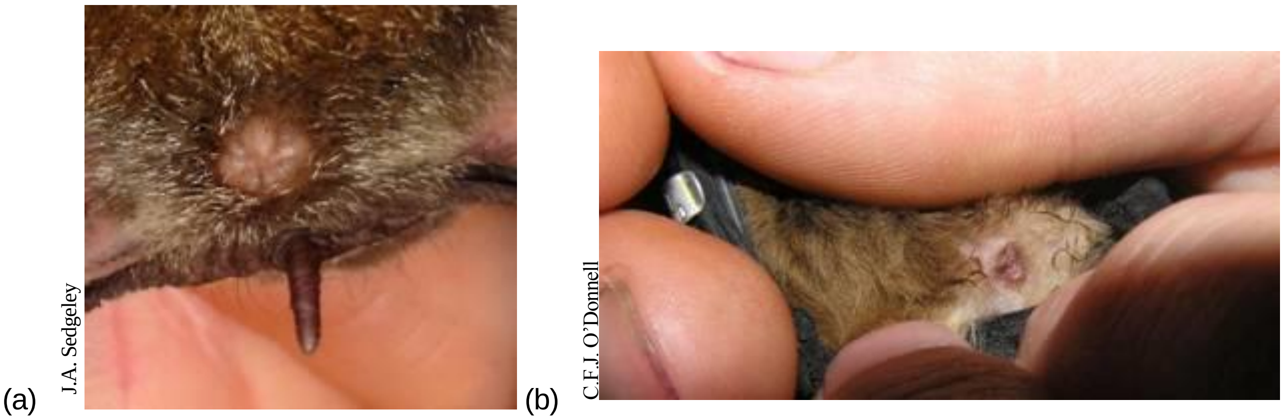


Figure 49. Genitalia of female bats (a) lesser short-tailed bat, showing clitoral pad, (b) long-tailed bat.

Reproductive status of females

Female bats that have never given birth are fairly easy to distinguish from females that have given birth by the state of their nipples. Females in the later stages of pregnancy and lactating females can also be identified.

(a) J.A. Sedgeley

(b) J.A. Sedgeley

Figure 50. Female bats showing conspicuous bare nipples which indicate they are lactating (a) long-tailed bat, (b) lesser short-tailed bat.

Nulliparous females

Females that have never given birth are termed nulliparous. They have very tiny nipples that can be difficult to find beneath their fur, and sometimes the nipples have tufts of hair on them (Fig. 51). In the Eglinton Valley, nulliparous female long-tailed bats were usually 1–2 years old.

Anonymous

Figure 51. Nipples of female long-tailed bats that have never given birth (nulliparous females).

Parous females

Females that have given birth are termed parous. They can be non-breeding parous females, or breeding parous females. Technically, a female that has never given birth before, but is pregnant, is still termed nulliparous. Early pregnancy can be difficult to diagnose in bats. Gentle palpation of the abdomen can diagnose pregnancies that are between half and two-thirds progressed. The lower abdomen becomes very distended, and it is possible to feel the single baby lying transversely by slight lateral pressure from two fingers. Care is needed not to confuse a female with a full stomach with a pregnant female. After parturition the vulva may appear blood-stained and swollen (Hutson & Racey 2004).

During pregnancy and lactation, the nipples become enlarged and protuberant and often change to a darker colour (keratinised). While the young is suckling, there is often an area of bare skin around the nipple (Fig. 50). Milk can sometimes be extruded from the nipple by gentle finger pressure on the base of the nipple. After lactation, the nipples regress markedly, but in long-tailed bats retain a larger and often darker appearance than nipples of females that have never given birth and suckled young. However, nipples of lesser short-tailed bats seem to recede more than those of long-tailed bats and tend to be flattened and pink in colour (O'Donnell 2002; Lloyd 2005).

Female long-tailed bats have occasionally been recorded carrying small babies with them when they go out to forage (Fig. 52). This seems to happen more frequently when only one or two females in a colony have given birth. As more females give birth, and as the babies grow larger, the babies are left behind to cluster together inside in the roost. Long-tailed bats change roost site almost every day, so female bats probably have to carry their young to new roost sites until the juveniles are old enough to fly.

C.F.J. O'Donnell

Figure 52. Female long-tailed bat caught in a harp trap with a young baby attached to her nipple.

When to look for reproductive females

Long-tailed bats and lesser short-tailed bats mate in autumn and gestation is delayed until spring. Births occur once a year, with females giving birth to a single baby (O'Donnell 2002, 2005; Lloyd 2005).

Long-tailed bats

Female long-tailed bats are visibly pregnant from early to late November and give birth from mid-November to mid-December depending on the population (O'Donnell 2005). The earliest birth dates have been recorded in South Canterbury (despite its southern latitude) and the latest in the Eglinton Valley. Births in South Canterbury begin in the first of November c. 1 month earlier than other populations. In Hawke's Bay, average birth date was about the last week of November. In the Eglinton Valley, births occurred throughout December, but most were highly synchronous, with 70% during a 10-day period in mid-December. Two bats that were visibly pregnant in the first week of January were first-time breeders. Sex ratio at birth was equal. Females first gave birth at 2–3 yrs old (O'Donnell 2002).

Lesser short-tailed bats

Lesser short-tailed bats give birth some time between mid-December and mid-January throughout New Zealand. Approximately 80% of reproductively mature females breed every year (Lloyd 2005).

Reproductive status of males

In some bat species, testes of juveniles are smaller than adult males, or the testes of sexually active bats may descend on a seasonal basis. These changes have not been observed in New Zealand bats. However, reproductive status of long-tailed bat males can be determined by assessing distension of the epididymides (sperm storage vessels) after spermatogenesis (sperm production) (Racey 1988; O'Donnell 2002; Hutson & Racey 2004).

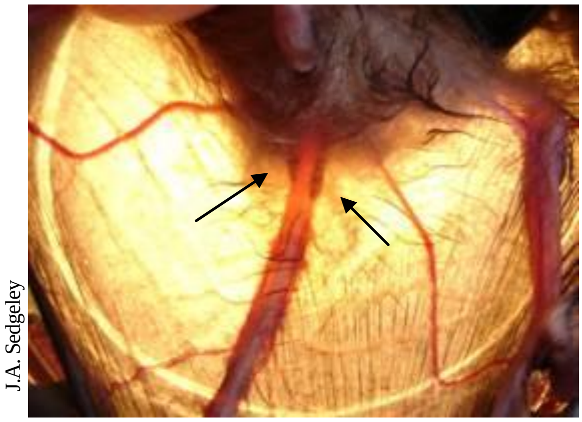


Figure 53. Location of epididymides in long-tailed bats. Colour and distension of epididymides can be used to assess breeding condition.

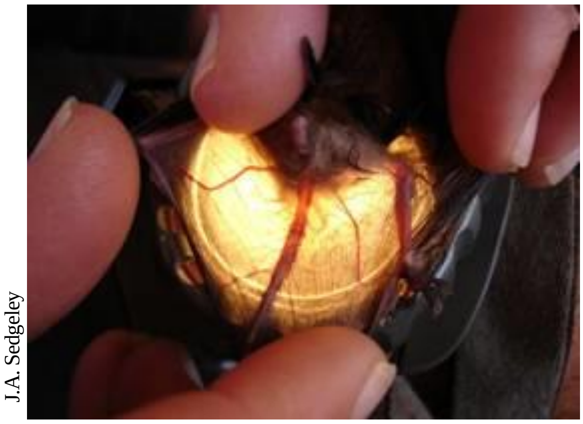


Figure 54. Positioning long-tailed bats on a torch to examine epididymides.

Determining reproductive status of long-tailed bats

The epididymides are attached to the testes and lie either side of the tail (Fig. 53). The easiest way to examine them is by illuminating the tail from behind by holding the tail over a torch. A torch with a reasonable diameter head is best (Fig. 54). It is important to be aware of how hot the torch gets. A spotlight, for example, could burn the bat's wing.

Reduction of pigmentation accompanied by varying degrees of distension of the epididymides can be used as a criterion of sexual maturity. The testes and epididymides of long-tailed bats are covered with a sheath of peritoneum (the tunica vaginalis). In juvenile and sexually immature male long-tailed bats the sheath of peritoneum is densely pigmented so the epididymides appear black and can be clearly seen through the skin (Figs 53 & 54). After spermatogenesis, sperm is released from the testes of adult males and passes through the epididymides into the caudae (tails of the epididymides). The caudae become distended and stretch the sheaths of peritoneum separating the black pigment cells so the epididymides appear clear, pale grey or white.

Determining reproduction condition in lesser short-tailed bats

Unfortunately, it is not possible to determine the reproductive status of live male lesser short-tailed bats using visual signs because neither testes nor epididymides are visible externally at any time of the year. Other techniques used to assess reproductive status in male bats overseas included surgical examination, and examination of urine for the presence of sperm. Neither of these techniques have been trialled in New Zealand.

When to look for reproductive males

Distended caudae epididymides are typically recorded in bats from late summer to early autumn, peaking in early autumn. Some bat species achieve sexual maturity in their first autumn; others may take many years to become sexually mature (Hutson & Racey 2004). In the Eglinton Valley, distended epididymides have been recorded in long-tailed bats that are 1 year old (mean = 1.6 yr, O'Donnell 2002). Sometimes juvenile bats have pale coloured epididymides even though they are not sexually mature (Racey 1988; Hutson & Racey 2004). We recommend that before checking the condition of the epididymides the bat is aged by assessing the ossification of its wing-joints (see 'Ageing bats').

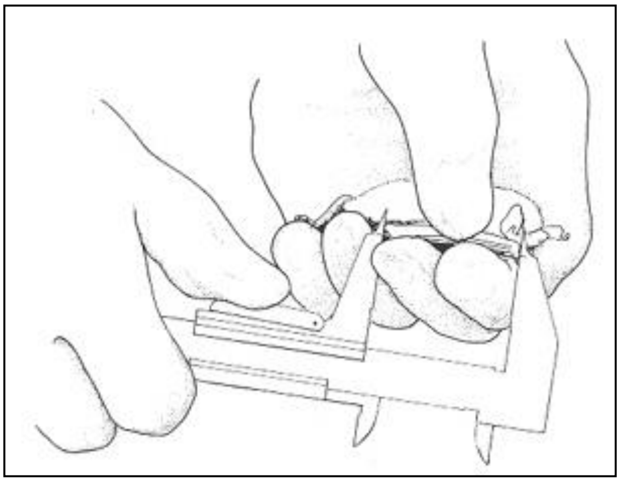
Common measurements

Linear measurements are usually taken with a vernier or dial caliper, but a ruler can be used. Body mass is usually measured with a spring balance. A magnifying hand lens may also be useful for some measurements. When taking any measurements it is important to record a range of background information, which **should** include: date, time, place of capture, method of capture, and reason for capture. Important information to support any morphological measurements **should** include: species, sex, age and possibly reproductive condition. The fate of each individual captured **should** also be recorded, e.g. released, banded, radio-transmitter attached, wing-biopsy punch taken, and in special permitted circumstances, bat collected. It is useful to develop a recording form (landscape A4 works well) with blank boxes to fill for use in the field. It is very rare for more than one person measuring a particular characteristic to come up with exactly the same result. It is therefore important to routinely record who took the measurement so that any biases can be considered when undertaking analyses.

How to measure forearm length

Forearm length is one of the few consistent measurements that can be taken of bats. It is probably the easiest measurement to take, is least variable, and generally proportional to the size of the bat. Despite being considered one of the least variable measurements, several studies have shown relatively large differences between observers measuring the same bat. In comparative studies that require high degree of precision it may be worthwhile limiting forearm measuring to one person.

Forearm length is taken by measuring the maximum length from the elbow to the wrist when the wing is in the folded position. This measurement is ideally taken with callipers, although a short steel ruler with an end stop can be used as an alternative. It is best to hold the bat in the non-dominant hand, and use the dominant hand for manipulating the callipers (Fig. 55). The elbow of the bat rests on the movable jaws of the callipers, the callipers are adjusted to the correct maximum length when you can see or feel a slight movement of the skin of the bat's wrist against the fixed jaw (Hutson & Racey 2004). Caliper measurements are usually taken to 0.1 mm accuracy.



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J.A. Sedgeley

Figure 55. Taking forearm length measurements.

Examples of forearm length recorded in New Zealand bats

Long-tailed bats

Forearm length in long-tailed bats varies throughout New Zealand ranging from 36.7 to 46.0 mm. Forearm length also varies among different sex and age classes (O'Donnell 2005). See Tables 6 & 7 for further details.

Table 6. Average measurements (mean ± 1 SE) of adult long-tailed bats from different localities. Adapted from O'Donnell (2005).

Location	**Sample size**		**Mean forearm length** (mm)	
	Male	Female	Male	Female
Waitākere Ranges	7	6	39.3 ± 0.3	39.4 ± 0.3
Ruakurī	31	70	39.5 ± 0.2	39.8 ± 0.1
Grand Canyon Cave	166	170	39.3 ± 0.1	40.1 ± 0.1
Puketitiri	53	145	39.9 ± 0.8	40.3 ± 0.9
Maruia	2	3	39.3 ± 0.4	39.5 ± 0.6
South Canterbury	19	74	38.7 ± 0.2	39.6 ± 0.1
Eglinton Valley	120	439	39.9 ± 0.9	40.5 ± 0.6

Table 7. Average measurements (mean ± 1 SE) of different age and reproductive classes of long-tailed bats from the Eglinton Valley. Adapted from O'Donnell (2005).

Age–sex class	**Forearm length (mm)**	
	Sample size	Mean
Reproductive female	601	40.5 ± 0.6
Non-reproductive female	217	40.5 ± 0.9
Adult male	157	39.9 ± 0.9
Juvenile female	109	40.1 ± 0.1
Juvenile male	140	39.8 ± 0.1

Lesser short-tailed bats

Measurements recorded for forearm length in adult lesser short-tailed bats from throughout New Zealand range from 39.1 to 46.9 mm. Forearm length varies among subspecies among different sex and age classes (Lloyd 2005; O'Donnell et al. 1999). See Tables 8 & 9 for more details.

Table 8. Size variation among populations and subspecies of lesser short-tailed bat. Forearm lengths (mm) are for adults. Adapted from Lloyd (2005).

Subspecies	**Population**	**Forearm length** (mm)		
		Sample size	Mean	Range
M. t. aupourica				
	Little Barrier Island	34	41.16	39.9–42.0
	Omahuta	8	39.97	39.3–41.1
M. t. rhyacobia				
	Urewera	5	44.18	43.4–45.7
	Whirinaki	43	44.20	42.3–46.2
	Kaimanawa	74	44.31	40.9–46.6
	Pureora	56	42.29	40.1–44.1
	Waitaanga	29	41.98	39.9–44.2
	Rangataua	547	44.01	40.1–46.9
	Waitōtara	9	41.79	40.4–43.7
M. t. tuberculata				
	Tararua	1	42.60	
	North-west Nelson	5	41.48	40.8–42.8
	Codfish Island	286	42.27	39.4–45.1
	Eglinton Valley	31	43.44	41.2–45.5

Table 9. Average measurements (mean ± 1 SD) of forearm length for different age and reproductive classes of lesser short-tailed bats from the Eglinton Valley (O'Donnell et al. 1999).

Age–sex class	**Sample size**	**Forearm length (mm)**	
		Mean	Range
Parous female	9	44.3 ± 1.27	41.3–45.5
Nulliparous female	18	43.1 ± 0.88	42.2–44.6
Adult male	4	43.0 ± 0.39	42.6–43.5
Juvenile female	6	42.9 ± 0.33	42.5–43.4
Juvenile male	7	41.8 ± 0.89	40.7–43.7

How to measure body mass (weight)

Body mass in bats varies greatly. It can vary over a 24-hour period and seasonally. This level of variation will put great limits on the usefulness of weight data. Body mass is really only useful in long-term studies of growth and body condition (Hutson & Racey 2004).

The best way to measure body mass is to use a spring balance such as a Pesola. A 50 g Pesola (long scale) is ideal and can be read to an accuracy of to 0.1 g. There is a range of Pesola balances available (e.g. 30 g, 60 g, 100 g or larger), but these balances may give less accuracy because some of the scales can only be read to 0.5 g. Spring balances can be obtained through the DOC Banding Office. Small electronic kitchen-style scales can also be used but are seldom practical in the field because they require a flat firm surface. Active bats **should** be placed in a small cloth bag, and the difference between the empty bag and the bag plus bat recorded. The bag needs to be small enough to confine the bat and prevent a lot of movement. A lighter bag will improve measurement accuracy too. There are alternatives such as cloth cones (the bats inserted down into the narrow end) or narrow sections of elasticated tights/pantyhose that can be wrapped around the bat. Plastic bags **should** be avoided because bats seemed to get stressed despite being held inside for a very short time. It is important to regularly re-weigh the empty bags, particularly if measuring a large number of bats. The bag will change weight if it gets damp or filled with debris or bat droppings. Ensure the balance and the bag are free of obstruction and wait until the bag and bat have stopped moving around to take the measurement.

When reading the scale it is important to hold the ring at the top so the balance swings. If the body of the balance is held while taking measurements the readings will be affected. It may also be necessary to calibrate the balance; for Pesolas this is done by turning the flat screw at the top.

Examples of body mass recorded in New Zealand bats

Body mass can change by as much as 30–50% with time of year, with reproductive condition, and with foraging success. Mass also varies among bats of different age and sex classes. Body mass of all bats should increase in autumn (late March) as they accumulate body fat reserves to help them survive through winter months. Weights of reproductive females vary according to time of the breeding season, but weights of males are relatively stable except during autumn (Hutson & Racey 2004).

Long-tailed bats

Adult mass of long-tailed bats from around New Zealand (for pre-feeding and non-breeding bats) range from 7.1 to 12.5 g; and non-flying young from 3 to 6 g. Bats of either sex can weigh up to 3 g more following successful foraging spells, but return to basal weights by dawn. There is also variation among age and sex classes (O’Donnell 2002, 2005). See Tables 10 & 11 for further details.

Table 10. Average body mass (g) (mean ± 1 SE) of adult long-tailed bats from different localities. Female mass *excludes* pregnant and nulliparous females for all study areas except Puketitiri (original data not available). Adapted from O’Donnell (2005).

Location	**Sample size**		**Mean body mass** (g)	
	Male	Female	Male	Female
Waitākere Ranges	7	6	9.5 ± 0.5	9.6 ± 0.7
Ruakurī	31	70	8.5 ± 0.1	9.8 ± 0.1
Grand Canyon Cave	166	170	9.0 ± 0.1	9.8 ± 0.1
Puketitiri	53	145	9.6 ± 0.1	11.9 ± 1.2
Maruia	2	3	8.7 ± 0.1	10.3 ± 0.3
South Canterbury	19	74	8.7 ± 0.2	10.2 ± 0.7
Eglinton Valley	120	439	9.3 ± 0.1	10.5 ± 0.1

Table 11. Average body mass (mean ± 1 SE) of different age and reproductive classes of long-tailed bats from the Eglinton Valley. All weights are *pre-feeding*. Adapted from O'Donnell (2005).

Age–sex class	**Sample size**	**Mean body mass** (g)
Pregnant female	125	12.3 ± 0.1
Lactating female	324	10.5 ± 0.1
Post-lactating female	152	10.6 ± 0.1
Non-reproductive female	217	10.1 ± 0.1
Adult male	157	9.3 ± 0.1
Juvenile female	109	9.1 ± 0.1
Juvenile male	140	8.9 ± 0.1

Lesser short-tailed bats

Body mass recorded for adult lesser short-tailed bats from throughout New Zealand, (excluding pregnant and lactating females) range from 9.8 to 21.1 g. There is also variation among the subspecies and age and sex classes (O'Donnell et al. 1999; Lloyd 2005). Female body mass can increase by up to 35% during late pregnancy (Lloyd 2005). See Tables 12 & 13 for further details.

Table 12. Size variation among populations and subspecies of lesser short-tailed bats. Body mass data are for adults, *excluding* pregnant and lactating females. Adapted from Lloyd (2005).

			Body mass (g)	
Subspecies	**Population**	Sample size	Mean	Range
M. t. aupourica				
	Little Barrier Island	34	11.94	10.1–13.6
	Omahuta	8	12.03	11.0–13.1
M. t. rhyacobia				
	Urewera	5	13.72	12.3–16.1
	Whirinaki	29	14.75	13.1–17.2
	Kaimanawa	27	14.65	10.6–17.3
	Pureora	55	13.70	11.7–16.4
	Waitaanga	20	12.06	10.0–13.7
	Rangataua	448	14.51	10.4–20.5
	Waitōtara	5	13.62	12.4–15.3
M. t. tuberculata				
	Tararua	1	14.10	
	North west Nelson	4	12.68	12.2–14.0
	Codfish Island	283	14.60	11.5–19.0

Table 13. Average measurements (mean ± 1 SD) of body mass for different age and reproductive classes of lesser short-tailed bats from the Eglinton Valley. Adapted from O'Donnell et al. (1999).

Age–sex class	**Sample size**	**Body mass** (g)	
		Mean	Range
Parous female	9	19.0 ± 1.54	16.9 – 22.0
Nulliparous	18	15.6 ± 1.18	14.0 – 18.3
Adult male	4	14.7 ± 0.90	13.6 – 15.8
Juvenile female	6	14.8 ± 0.65	14.0 – 15.5
Juvenile male	7	14.3 ± 0.92	13.2 – 16.0

Body condition index

Because body mass of individual bats can vary so much, weights are only really useful in long-term studies of growth and body condition. There is little point in amassing data in a casual way. A more accurate description of body condition can be derived using both body mass and forearm measurements. This procedure corrects for differences in skeletal size between bats, without loss of mass units (O'Donnell 2002). The body condition index is calculated by dividing body mass by the individual's forearm length, then multiplying by the mean forearm length for the total sample of bats that is being examined.

Other measurements

Many features on a bat can be measured (see Fig. 44). Some measurements may only be useful for specific and detailed morphological studies. Several published identification keys include a wide range of measurements for each bat species (e.g. Churchill 1998). Knowledge of how to take these measurements will aid in species identification. Some of these measurements can be variable and inconsistent because they involve soft parts such as wings or ears that can be extended to varying degrees. This variation may limit the usefulness of measurement data. Limiting the number of people measuring will reduce the variability of measurements in long-term studies. A number of the more common measurements are listed below. Examples of some measurements taken from New Zealand bats are also given (Tables 10 & 11).

List of features that can be measured

- Head and body length (nose tip to anus)
- Head length (from junction with neck to nose tip)
- Body length
- Tail length (anus to tail tip)
- Length of fifth digit/finger (from inside of wrist to tip of finger) (Fig. 56)
- Wingspan (wing tip to wing tip) (Fig. 57)
- Length of metacarpals and phalanges
- Tibia length
- Foot (heel to toe tips excluding claws)
- Calcar length (from base of ankle to tip, can also compare with the total length of the edge of the tail membrane).
- Ear length (from notch at base of the pinna to tip) (Fig. 58)
- Tragus width (the greatest width) (Fig. 58)
- Tragus length (maximum length from base to tip, ignoring any curves) (Fig. 58)
- Outer canine width[9] (distance between the upper surfaces of the canines at the gum line)
- Abnormalities (unusual colouration, injuries, deformities)

[9] This measurement is extremely difficult to take in live bats.

Wing depth at fifth digit (or fifth digit length)

The easiest and most reliable way to take this measurement is to use a measurement from the outside of the wrist to the finger tip. This is best done on a flat surface (Hutson & Racey 2004) (Fig. 56). Some researchers take the measurement from the inside of the wrist to the tip of the finger. Whichever measurement is selected it should remain consistant throughout the study. Measurements of fifth digit in long-tailed bats (O'Donnell 2005) have been taken using the first method.

Figure 56. Measuring the fifth digit of a long-tailed bat. Note the bottom edge of the wing is curling upwards. To obtain a more accurate measurement the wing tip needs to be gently smoothed down so it is flat.

Wingspan

Wingspan is not a particularly useful field measurement because too much variation in measuring technique is possible (Hutson & Racey 2004). However, it is a measurement often quoted in books and identification keys (e.g. Churchill 1998). The measurement is taken from wing-tip to wing-tip usually with the bat laid out on a firm surface (Fig. 57). It is very difficult determine how far to extend the wings, and care **must** be taken not to over-extend and injure the bat. **This technique is not recommended for lesser short-tailed bats.**

Figure 57. Taking measurement of wingspan in a long-tailed bat.

Ear length and tragus measurements

Ear length is a difficult measurement because bats often fold down their ears when being handled. It is important to ensure the measurement is taken from an ear that is fully erect. Ear length is measured from the notch at the bottom of the inside of the ear to the tip of the ear. Tragus width is taken at the greatest width and tragus length is the maximum length from base to tip ignoring any curved edges (Hutson & Racey 2004) (Fig. 58).

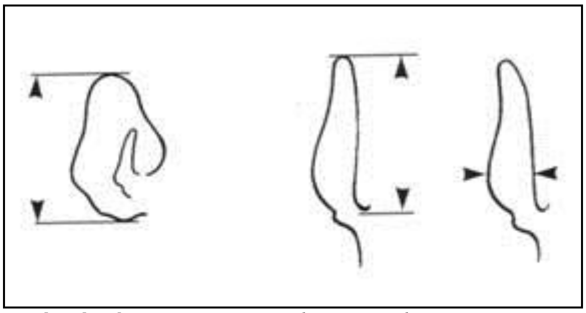

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Figure 58. Where to take ear and tragus measurements.

Wing measurements recorded for New Zealand bats

The following measurements have been recorded for long-tailed bats throughout New Zealand: adult head and body length 42–63 mm; tail 30–46 mm; ear 7.2–11.0 mm (Dwyer 1962; O'Donnell 2001, 2005; C. O'Donnell, unpubl. data). Tables 14 & 15 provide further details of body length, length of fifth digit and wingspan (O'Donnell, 2005).

Measurements for adult lesser short-tailed bats throughout New Zealand include: wingspan 280–300 mm; body length 60–70 mm; ear length 17–19 mm; tibia length 14.5–17 mm (Lloyd 2001, 2005).

Table 14. Average wing measurements (mean ± 1 SE) of long-tailed bats from different localities. Adapted from O'Donnell (2005).

Location	**Sample size**		**Wing depth at fifth digit**		**Wingspan**	
	Male	Female	Male	Female	Male	Female
Waitākere Ranges	7	6	49.9 ± 0.5	49.2 ± 0.7		
Ruakurī	31	70	50.0 ± 0.2	50.9 ± 0.1		
Grand Canyon Cave	166	170	50.2 ± 0.1	51.4 ± 0.1		
Puketitiri	53	145			276.4 ± 7.0	282.7 ± 7.0
Maruia	2	3	51.5 ± 0.5	53.0 ± 1.0		
South Canterbury	19	74	51.6 ± 0.3	52.9 ± 0.2		
Eglinton Valley	120	439	52.5 ± 0.3	53.5 ± 0.1	273.7 ± 1.4	277.3 ± 1.2

Table 15. Average measurements (mean ± 1 SE) of different age and reproductive classes of long-tailed bats in the Eglinton Valley. Adapted from O'Donnell (2005).

Age–sex class	**Body length (mm)**	**Wing depth at fifth digit (mm)**	**Wingspan (mm)**
Reproductive female	56.5 ± 3.5	53.5 ± 0.1	277.3 ± 1.2
Non-reproductive female	53.8 ± 2.4	53.5 ± 0.2	277.3 ± 1.8

Adult male	52.6 ± 1.4	52.5 ± 0.3	273.7 ± 1.4
Juvenile female	45.5 ± 3.2	51.4 ± 0.2	263.2 ± 2.2
Juvenile male	50.7 ± 2.9	51.1 ± 0.2	263.1 ± 1.7

Wing-tracing

Morphological data can be obtained using wing-tracings. Standard aerodynamic measurements can be calculated from wing-tracings using conventions outlined in a paper by Norberg & Rayner (1987) (wing loading, aspect ratio and wingtip shape index, etc.). A combination of wing-morphology and echolocation characteristics can determine the range of habitats in which a bat can fly and the different foraging strategies it can use. Wing-morphology differs significantly between long-tailed bats and lesser short-tailed bats. See Webb et al. (1999), O'Donnell (1999), Jones et al. (2003), and Lloyd (2005) for detailed descriptions.

The most common technique to obtain these measurements is to make wing-tracings by drawing around the bats wing. The bat is placed face down on a sheet of paper attached to a hard surface such as a clipboard. It is important to draw one wing, including head and tail, and keeping the leading edge of wing as straight as possible (Fig. 59).

C.F.J. O'Donnell

Figure 59. Holding a long-tailed bat in position to take a wing-tracing.

Digital cameras can be used as an alternative to tracing around the wing. This technique is being used by researchers in Australia and Britain and it is proving to be much quicker and easier. The Australian technique involves lying a bat face-down with its wings gently extended and taped with sticky tape. Initially two bits of tape were used on each wing (Fig. 60), but one piece of tape over the forearm proved to be sufficient. The digital camera was mounted on a tripod with the camera facing directly down, with the camera 60 cm above the bat. A plastic board (a thin flexible chopping board) that had a piece of graph paper and a scale was used so that the measurements could be calibrated. The board made it easier to get the tape on and off quickly and could also be cleaned. Standard morphological measurements were taken from the photograph using the software program 'ImagePro Plus' (Lindy Lumsden, pers. comm.). **Note this technique has not been trialled in New Zealand and ethics approval would be required to tape down the bat.**


L. Lumsden

Figure 60. Taping an Australian free-tailed bat *(Mormopterus* sp.) to obtain wing-measurements.

Collecting samples

Skin for DNA analysis

See the section on '[Taking tissue samples for genetic purposes]'.

Ectoparasites and associated insects

Many arthropods live on bats for at least part of their lives. New Zealand bats are host to several ectoparasites. Both long-tailed bats and lesser short-tailed bats host several species of mite. Long-tailed bats are hosts to the long-tailed bat flea *(Ponnibius paciflcus)* and lesser short-tailed bats are hosts to an undescribed species of tick belonging to the genus *Argas* (*Carios*). Lesser short-tailed bats are also host to the endemic and threatened New Zealand batfly (*Mystacinobia zelandica*) which is not a parasite, but feeds on bat guano throughout its life cycle for more details). Although superficially similar to other batflies, the New Zealand batfly evolved separately, and is placed in its own family (Lloyd 2005). Arthropods associated with bats have become very specialised in their morphology, physiology, life cycle and ecology and are therefore interesting to study in their own right. Additionally, relatively little is known about the relationships between many of the parasites and their hosts; for example, what role they might play in disease transmission (Hutson & Racey 2004).

Parasites and batflies are found by inspecting the flight membranes, feet, ears, face, and anus, by blowing through the fur. They can be removed with fine forceps or tweezers, or a fine paintbrush, and are best stored in 70–80% ethanol. Some of the insects are very agile, but can be immobilised with a small dab of ethyl acetate. Some mites may be firmly attached and there is the risk of leaving mouthparts embedded in the host. If a parasite is firmly embedded it is preferable to leave it where is. There is a risk of the embedded parts causing infection, and it is very difficult to identify a parasite with its mouthparts or head missing (Hutson & Racey 2004). Stored specimens should be clearly labelled with full data including date, locality, name of collector, and details of the bat host (e.g. species, age, sex, position on body). See 'Collection, storage and transport of diagnostic samples from birds and reptiles' (olddm-718668) for more details.

Preserving dead bats

Dead bats **should** never be discarded immediately. They may be useful for a variety of purposes, e.g. as a voucher specimen, exhibitions, for disease screening, education, and training, obtaining DNA samples, or for other scientific purposes. Dead bats are most frequently found close to long-term roost sites, or are recovered from pet cats. It is an unfortunate fact that any project involving catching and handling of bats may at some time result in accidental injuries and deaths. It is important that all bat deaths are documented and the body sent off for post-mortem examination and/or to a museum. Carefully documenting injuries and deaths in bats can only aid in improving our catching and handling techniques. A post-mortem examination may reveal that the bat did not in fact die from mishandling.

DOC has protocols for processing dead animals and these should be adhered to. For full details see the DOC standard operating procedure 'Wildlife health SOP' (olddm-766252; 'Wildlife health SOP index page'—olddm-757175). The 'Wildlife health SOP' provides clear guidelines on when it is appropriate to send off specimens for post-mortem examination. If the bat is to be sent for autopsy it **should** be sent to a veterinary pathology laboratory according to the instructions for submitting samples in 'Collection, storage and transport of diagnostic samples from birds and reptiles' (olddm-718668) and submitting bodies 'Massey University IVABS pathology sample submission instructions' (olddm-921158).

Generally, a dead animal should be chilled to refrigerator temperature (approx 4°C) as soon after death as possible, and despatched for diagnosis on the earliest available transport. Freezing interferes with examination of tissues, and some aspects of microbiological culture, and should be a last resort if the dead body cannot be delivered within approximately 24 to 36 hours. Alternatives are fixing the body whole in alcohol, or field dissection and submission of fixed tissues for histopathology. Non-DOC people should deliver the body to the local DOC office and provide information for the 'Wildlife submission form' (olddm-677719).

DOC maintains a contract with Massey University's Institute of Veterinary, Animal and Biomedical Sciences (IVABS) for the post-mortem of wildlife—where possible, make use of this service. Massey University tends to keep all specimens after autopsy so if the specimen is to be returned to the sender (DOC only) or to be sent on to a museum it should be accompanied with a letter requesting its return.

If it is decided to send a specimen directly to a museum (Te Papa in Wellington, or Canterbury Museum in Christchurch) it is best to preserve the bat in one of three ways in the following order of preference (1) frozen; (2) in 70–80% ethanol; (3) in methylated spirits. The latter is useful in remote situations when freezing is not an option and no ethanol is available. If there is likely to be a delay before getting the bat to the museum, it will aid preservation if the abdomen is opened to allow the preserving fluid to penetrate, and the mouth propped open with a piece of matchstick. All specimens should be clearly labelled with date, time, location, species sex, and contact details of the collector. If mailing the specimen to the museum, the outside of the envelope should also be clearly labelled describing the contents as either a specimen preserved in alcohol, or as a frozen specimen. Do not mail specimens at times when there is unlikely to be appropriate staff available at the museum to process them (e.g. weekends and public holidays).

Releasing bats

This section identifies the best ways to release bats that have been captured and held for a relatively short time for identification and discusses implications of release for those that have been held over a longer period.

Where and when to release

Although bats are known to be able to home considerable distances (Guilbert et al. 2007), it is always preferable to release them as close to point of capture (or where they were found) as possible. Every attempt **should** be made to do this. This ensures that the bat is in familiar territory and is able to locate a suitable roost site or foraging area rapidly. Unless there are unavoidable circumstances, bats **should** always be released either at night, or at dusk or dawn. If it is not possible to release a bat at these times it is preferable for the bat to be placed in an artificial roost box, or in a crevice or tree hole so it can emerge by itself when it gets dark.

How to release

Warm and active bats will often fly directly out of a holding bag once the top is opened. If releasing by hand, the bat **must not** be thrown into the air because it may not be ready to fly. Simply hold out your arm and open your hand and wait for the bat to fly off in its own time (Fig. 61). Having a bat detector or light switched on will help confirm the bat has flown away rather than falling to the ground. Don’t shine the light directly into the bat’s face.

Young bats that have recently started flying and heavily pregnant bats sometimes need a bit of extra lift to take off. To do this, find an open area to stand in, free of obstructions, and hold your hand high in the air. Sometimes it may be necessary to put a bat onto a tree so it can crawl up even higher before it takes off. If a bat becomes torpid and is unwilling to fly, it should be warmed for a few minutes before release. Torpidity during capture and handling procedures seems to occur more frequently in long-tailed bats than in lesser short-tailed bats. It generally occurs when a bat has been by itself in a holding bag (rather than with a group of bats in the same bag), and when bats have been captive in a harp trap bag for several hours. Bats can be warmed up by holding the bat inside loosely cupped hands, or popping the bat inside a bag under your clothing and close to warm skin. Care should be taken when handling lesser short-tailed bats because they may bite through gloves or a bag as they become active.

D. Geddes

Figure 61. Releasing a long-tailed bat from hand.

Success rates of returning to the wild

The success with which bats can be returned to the wild may depend on the length of time for which they have been held captive and other factors such as their flying ability (Racey 2004). Radio-tracking and banding studies show that bats that are caught, handled, examined and released on the same evening, or the evening following capture, integrate back into their colonies. Bats are long-lived animals, so it seems reasonable that that may have good long-term memories for home range, and the ability to home (Racey 2004; Guilbert et al. 2007). Healthy wild bats that have been kept in captivity for months and then released have subsequently been found in their original colonies and foraging areas. For example, radio-tagged lesser short-tailed bats that had been held in captivity for several months on Codfish Island/Whenua Hou found their way back to occupied communal roosts the same night they were released (Sedgeley & Anderson 2000).

Bats born and raised in captivity may not be suitable for release back to the wild for several reasons, e.g. lack of contact with conspecifics, lack of detailed knowledge of any area, inability to forage on wild prey, and inexperience at selecting suitable roost sites. For these reasons it is generally considered survival rates of these bats is low (Racey 2004). However, there are instances of bats raised in captivity habituating to the wild (e.g. Devrient & Wohlgemuth 1997). In 2005, juvenile lesser short-tailed bats that had been raised in captivity were translocated to Kapiti Island to form the basis of a new colony. At the time of writing these bats were still going through a process of 'soft release' and being monitored.

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Banding and marking

This section describes some of the more common techniques used for marking bats and discusses their relative advantages and disadvantages. Some of the important issues that need to be considered before undertaking a marking study are also discussed. Radio-tagging and radio-tracking techniques are dealt with in a separate section (see ‘Attaching radio transmitters’).

There are several effective techniques for temporarily marking bats that are applicable to both extant New Zealand species. These are described below in ‘Short-term marking of long-tailed bats and lesser short-tailed bats’. Long-tailed bats and lesser short-tailed bats differ markedly in their morphology (body size and shape) and behaviours. As a consequence, the suitability of various long-term marking techniques and their effects on the two bat species differ. Not all techniques have been trialled on both species. Long-term techniques for long-tailed bats (trialled and un-trialled) are described in ‘Long-term marking of long-tailed bats using metal bands’ and ‘Long-term marking of long-tailed bats using other methods’, and those for lesser short-tailed bats are described in ‘Long-term marking: lesser short-tailed bats’.

Introduction to marking

Why mark bats?

Recognition of individual animals plays an integral part in ecological research and conservation of bats. Marking can provide information about population size and dynamics, survival, dispersal, migration, social behaviour, feeding ecology, homing behaviour, roost use and almost every facet of bat ecology (Stebbings 2004).

Important considerations before undertaking marking

All marking methods will affect the subject to a greater or lesser extent, at least in the short-term. Marking can affect animals by altering behaviours or interactions with their own or other species; their health and welfare; their capacity to reproduce; population dynamics and other factors. Specifically, marking can affect an animal in three ways:

1. The act of capture and marking can cause pain and stress.
2. The presence of the mark may restrict movement or disrupt breeding or social interactions. Incorrectly placed or poorly fitted tags may lead to injury and loss of function.
3. Observation of marks almost always requires repeated recapture and handling, and so leads to further stress (Beausoleil et al. 2004).

Therefore, workers **must** consider whether it is really *necessary* to mark bats to achieve the proposed research/management objectives. **New Zealand bats are fully protected, and it is illegal to catch, handle, mark or keep them without appropriate permits and ethics approvals.** Marking of bats **must** only be undertaken if a research study or management project requires individual or group recognition. See Mellor et al. (2004) and Beausoleil et al. (2004) for further discussions on ethics and general safeguards for marking wildlife in New Zealand.

Disturbance and other negative effects to bats can be minimised by recognising the advantages and disadvantages of different marks and marking procedures, and by choosing the most affective and humane way of applying the marks (Mellor et al. 2004). Before selecting a particular technique the following issues **must** be considered (adapted from Barclay & Bell 1988; Beausoleil et al. 2004).

- Specific objectives of the study and the nature of the data required.
- Level of recognition required.
- Which species is being studied (not all marks are appropriate).
- Size and conservation status of the population.
- Welfare of individuals and populations. Will the mark affect survival or behaviour?
- Duration of study and length of time marks are required to last.
- Amount of time and resources available to researchers.
- How many individually distinct marks will be required?
- How rapidly will the bats need to be marked?
- How near will an observer need to be to identify the mark?
- Will it be necessary to recapture the bat to read the mark?
- Levels of training and experience required.

There are only two methods currently approved for long-term marking of New Zealand bats; forearm banding for long-tailed bats and passive integrated transponder (PIT) tags for lesser short-tailed bats. Both techniques have strict protocols attached to their use. The Bat Recovery Group will carefully consider any projects involving new long-term marking techniques that are not currently approved. Any new technique which will help improve our understanding of New Zealand bats, and aid in their conservation, **should** be encouraged. All projects **must** incorporate appropriate and adequate protocols for monitoring potential effects of new techniques.

Short-term marking of long-tailed bats and lesser short-tailed bats

Short-term methods are generally far less invasive than long-term marks. They are less likely to cause tissue damage, pain, or stress, and are not likely to restrict movement. Short-term marks, depending on the technique, can last from 1 day to several weeks.

Fur-clipping

Fur-clipping is a useful and harmless technique for identifying individual or groups of bats (Fig. 62). Marks will last for several weeks, but may grow out rapidly (2–3 weeks) when bats are moulting. It is recommended that one or more small patches of fur are cut rather than one large area.

Figure 62. Marking a lesser short-tailed bat with a furclip using fine beard trimmers. Note: A bat weighing bag is being used to cover the bat's head to gently restrain it and prevent it biting. This type of restraint is not necessary for long-tailed bats.

Fur is usually cut from the dorsal surface of the bat in up to four or five locations (e.g. left shoulder, right shoulder, middle, left hind body, right hind body). However, if radio-tracking is also being undertaken in the study area, a furclip in the middle of a bat's back could be confused with a mark left by a radio-transmitter. Up to four clipped patches will result in many unique combinations, particularly if age and sex of the bat are also recorded. The fur is most easily cut with sharp scissors or electric moustache/beard trimmers.

Advantages:

- No known harm is caused to bats.
- Marks are relatively easy to apply, and bat handling time is moderate.
- Several unique combinations possible.
- Marks can last up to several weeks depending on time of year.
- The technique is low in cost.

Disadvantages:

- Marks can grow out with 2–3 weeks during moulting season.
- If a large number of bats require marking, the individual marks get more complex and consequently handling time per individual increases.
- Bats have to be re-caught to read the marks.

Dye-marking

Application of black hair dye has been used successfully on lesser short-tailed bats (B. Lloyd, pers. comm.). There are, however, several disadvantages with this technique. Almost all human hair-dyes contain harsh chemicals such as peroxides, and because the dyes do not 'fix' or 'set' immediately, bat handling time is relatively long. If the dye does not dry and set adequately it will quickly rub off. Quick drying coloured antiseptic skin dyes such as 'gentian violet' or 'magenta' have also been trialled on bats. An individual lesser short-tailed bat was marked with magenta during a captivity trial. The mark faded, but readable at the end of the 26-day trial (J.A. Sedgeley, unpubl. data).

Gentian violet has been used by wildlife carers in Australia to mark bats, but recent trials on lesser short-tailed bats in the Eglinton Valley proved to be unsuccessful. Marked bats held overnight in cloth bags had lost their marks by the next day. Dye marks also disappeared from wild bats that were both fur-clipped and marked with gentian violet when they were recaptured 4 days after being marked (C.F.J. O'Donnell, unpubl. data). Magenta and gentian violet can be obtained from a chemist/pharmacy, but a prescription is necessary for gentian violet.

Small blobs of non-toxic paint could also be considered, but this technique has not been trialled. The use of dyes or similar to mark bats has the same advantages and disadvantages as fur-clipping.

Chemical light-tags

Chemiluminescent tags have been used on bats to obtain information on foraging ranges, hunting patterns, dispersal routes, microhabitat use and flight behaviour (reviewed in Barclay & Bell 1988). They have been used successfully to study habitat use of bats in Australia and micro-habitat use of lesser short-tailed bats in Fiordland, New Zealand (Lumsden et al. 1994; Christie 2003).

The cheapest and easiest method of chemiluminescent tagging is to use small capsules filled with cyalume (Buchler 1976). Cyalume is a phosphor compound and a peroxide-based reactant. The two liquids react when mixed to produce a bright 'cold' light. The brightness and duration depends on the relative proportions of the chemicals. Equal proportions produce a very bright light for about 2 hours (Stebbings 2004). Cyalume can be obtained from emergency light-sticks that are sold in many outdoor and diving/sports shops, and are available in a range of colours (bright-green, white, blue red). Cyalume tags are made by removing liquid from the light-sticks using a hypodermic needle and syringe, and injecting the mixed liquids into small glass spheres, plastic heat-shrink tubing or gelatine pill capsules. If glass spheres or tubing are used, great care is needed to seal the aperture to prevent leakage. There is also some evidence that bats can bite through gelatine capsules and can die from ingesting the contents (LaVal et al. 1977) but other studies found no evidence of this toxicity (Racey & Swift 1985).

A much simpler type of cyalume tag, in the form of a luminescent fishing lure, has been used in Australia (Lumsden et al. 1994) and New Zealand (Christie 2003). Chemiluminescent lures can be obtained from fishing shops and K-Mart. The brands used on lesser short-tailed bats are 2.9 mm wide, 24 mm long, and glow green/yellow. Tags can be glued to the dorsal or ventral surface of the bat's body, depending on the vantage point of the observer and the elevation at which the bat is expected to fly. Tags were attached to un-trimmed fur of New Zealand bats using F2 Contact adhesive (Ados Chemical Company, New Zealand), a non-toxic latex-based glue (surgical appliance adhesive would also be suitable). As with all contact adhesives, a small amount of glue is spread on both the fur of the bat and the surface of the tag, and the tag pressed into place once the glue has dried (Fig. 62). Attachment to un-trimmed fur allows the bat to groom the tag off in a relatively short period of time. There seems little point attaching the tag more firmly if the luminescence wears off after a few hours. If it is necessary to retain the tag on the bat for a longer period, the bats fur can be trimmed before attachment. If the bat's skin is nicked or cut with scissors during trimming, tags **must not** be fitted.

C.F.J. O'Donnell

Figure 62. Long-tailed bat fitted with fishing lure cyalume capsule.

Advantages of fishing lure tags:

- They are relatively quick and easy to apply.
- They are made of a plastic that appears to be thick enough to prevent bats biting through.
- Tags are very light-weight (< 0.8% of lesser short-tailed bat body mass).
- Tags are very cheap.

Disadvantages of cyalume tags in general:

- The toxicity of cyalume liquid to bats is questionable.
- The lifespan of tags is extremely short.
- Tags provide very limited options for individual recognition.
- Cyalume tags can only be observed over a relatively short distance (although this can be improved with binoculars) and are of limited use in dense vegetation or forest interior (Christie 2003).

Other temporary marks

Glue-on tags

Various materials have been glued onto bats, but are mostly groomed off fairly quickly if the bat is active. However, tags may main remain in position for long periods when bats are hibernating (Daan 1969; Stebbings 2004), and can frequently stay on long enough to obtain useful data such as information on roost use. For example, tagged bats (radio-tags, cyalume capsules, plastic disks) can be clearly picked entering and exiting roosts on video-recordings (Sedgeley & Anderson 2000). Coloured reflective tape (either using the sticky back, or adding extra glue) and small disks of plastic have been applied in various positions on bats bodies or heads. In one study, small plastic disks, each with a unique number or letter code, were glued onto bats' heads (Daan 1969). PIT tags (also known as microchips) can also be glued onto bats as a temporary tag, but are more commonly inserted under the skin as a permanent tag (see 'PIT tags (microchips)' below).

Advantages:

- Depending on the type of tag used, this method can be very inexpensive.
- The tags are quick and easy to apply.
- With some tags individual recognition of bats is possible.
- Some tags can be picked up on video cameras enabling bats to be identified without being recaptured.

Disadvantages:

- Generally, glue-on tags are relatively short-lived.
- PIT tags can only be detected at distances of up to 150 mm and require a specialised reader.

Long-term marking of long-tailed bats using metal bands

Forearm banding with 2.9 mm (narrow) flanged metal bat rings from the UK (Porzana Ltd, aluminium alloy split metal bat rings; Porzana@wetlandtrust.org) is the only method currently approved for long-term marking of long-tailed bats.

Metal bands (rings)

Bands (rings) fixed over the forearm are the most widely used and successful long-duration marking method for bats. A variety of metallic bands have been used to individually mark large series of bats with unique number combinations. Large-scale banding of bats with metal bands began in the 1930s in Europe and North-America, and around the 1960s in Australia. Initially bird-leg-bands were used, and later flanged bands were developed specifically for bats (Barclay & Bell 1988; Stebbings 2004).

Several countries including Australia, Great Britain and United States have restricted or prohibited banding in certain species or families of bats where serious injuries or population declines have been attributed to banding (Baker et al. 2001; O'Shea et al. 2004). Banding trials in New Zealand resulted in the approval for metal bands to be used as a long-term marking technique for long-tailed bats (see 'Trials of metal bands on long-tailed bats' below). However, bands caused unacceptable injuries in lesser short-tailed bats, and banding is not currently approved for this species (see 'Long-term marking: lesser short-tailed bats' below).

Advantages:

- After sufficient training, metal bands are relatively easy and quick to apply.
- Numbered bands allow individual recognition of bats.
- Correctly applied bands should last the lifetime of the bat and cause them no harm. Some species of bats have been observed to chew the bands until the numbers become unreadable, but this has not occurred to date in long-tailed bats.

- Banding is probably the most successful technique for providing accurate information on population dynamics, survival, dispersal, migration, social behaviour, feeding ecology, homing behaviour and roost use.

Disadvantages:

Bats are generally considered much more sensitive to banding than birds for two main reasons:

1. Bat bands potentially cause more problem injuries because the band is in contact with soft tissues of the forearm and wing-membrane. Bands can cause serious injuries, particularly if the wrong size and design of band is used or if bands are applied incorrectly. Injuries vary according to type of band, species, age of bat, time of year and amount of care taken in band application. Injuries include infections to forearms, wrists and wings and can reduce manoeuvrability.
2. Bats are generally more vulnerable to disturbance than birds. Bats have to be caught to both apply the band and recapture to read the bands. Disturbing and handling bats during an energetically critical period may be stressful enough to reduce survival (Barclay & Bell 1988; Stebbings 2004).

Trials of metal bands on long-tailed bats

The most comprehensive banding trial of long-tailed bats was conducted in the Eglinton Valley, Fiordland National Park. Trials began in the summer of 1993/94 using ‘A’ size metal alloy bird bands. This type of band was chosen because it had been used relatively successfully in Australia on the chocolate wattled bat (*Chalinolobus morio*), a close relative of the long-tailed bat. The ‘A’ bands were first trialled on captive long-tailed bats, and after no ill effects were found, the bands were trialled on 119 free-living bats. Half of these bats were fitted with standard bird bands, and half were fitted with modified bands where the sharp corners were filed down. After 2–4 weeks bats were recaptured and assessed for wing damage and other injuries. Thirty-seven percent of recaptured bats with unmodified bird bands and 77% of bats with modified bands had wing damage. Most injuries were judged to be slight or moderate, but six were considered severe. Wing-abrasion and swelling at the wrist and along the leading edge of the forearm occurred in the severe cases. The bird bands were subsequently removed and this band type is now considered unsuitable for use on long-tailed bats (C.F.J. O’Donnell, unpubl. data).

Flanged bat bands (of different sizes) from Great Britain and Australia were trialled during the following summer season. The British bands were made of a softer metal and had smoother edges than the Australian bands. The Australian bands caused injuries, whereas the British bands caused no short- to medium-term injuries. As a result of this trial, only British bands continued to be used (Fig. 63). Over a 10-year period, 1024 long-tailed bats were banded in the Eglinton Valley, with a total of 5282 recaptures. Less than 10 injuries were recorded. Injuries ranged from very mild to severe swelling of the forearm (C.F.J. O’Donnell, unpubl. data).

C.F.J. O'Donnell

Figure 63. A British 2.9 mm flanged bat band (ring) fitted to a long-tailed bat.

Applying bands

Bands are fitted over the bat's forearm. Some studies elect to use one forearm for females and the other males, although this has limited application. To minimise damage it is very important the band is fitted as loosely as possible so it can freely slide up and down the forearm. However, the gap should be sufficiently small to prevent the band sliding over the wrist or elbow joint, and finger bones becoming trapped. It is important the band is closed evenly, a band that is pinched at one end, and open at the other may cause injury (Barclay & Bell 1988; Stebbings 2004). Correct gap closure for bands fitted to long-tailed bats **should** be c. 0.7–0.8 mm (Fig. 64).

C.F.J. O'Donnell

Figure 64. Correct closure on long-tailed bat band. The closure is small enough to prevent the band from sliding over the wrist or elbow and to prevent finger bones becoming trapped (c. 0.7–0.8 mm). It is important the band is closed evenly.

An effective banding technique is to hold the bat face down in the palm of the hand with the wing partially extended (Fig. 65a). The forearm **must** be supported by the first or index finger (Fig. 65d). The band is applied by the other hand. It is preferable that the bat is banded by one person because it is easier for one person to control the bat and to assess the amount of pressure necessary to apply to the band. See Fig. 65 for more details. Bands that have been previously used on a bat, or new bands that are misshapen **must not** be used because it is difficult to achieve smooth and even closure. **Training is required in fitting the band so it retains correct shape and gap closure.**

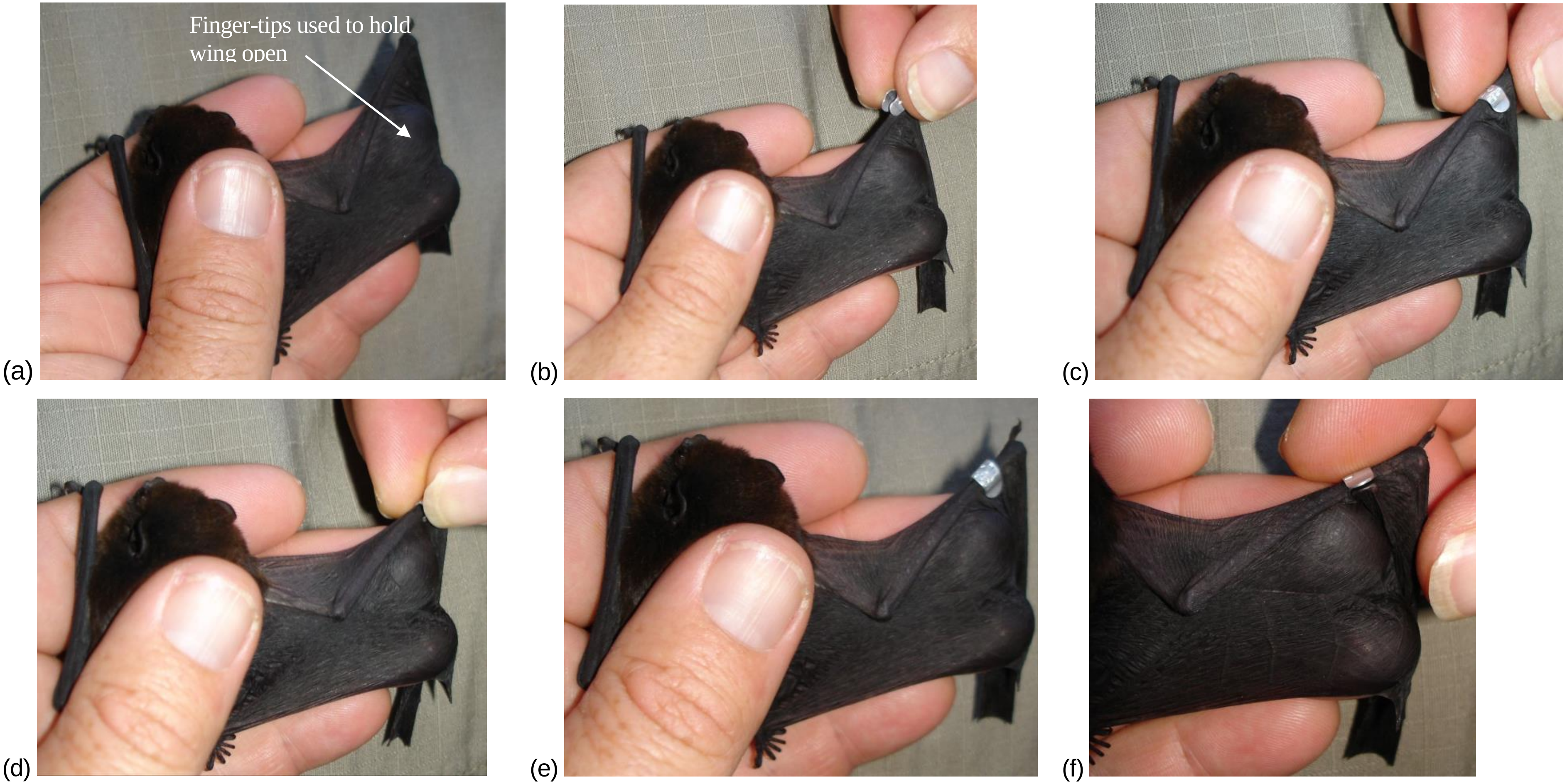

Figure 65. Technique for applying bands to long-tailed bats. (a) Hold the bat face-down in the palm of the hand with the wing partly extended. Fingers positioned beneath the wing membrane between the bat's body and bat's fifth digit can be used to keep the wing open. (b) (c) The new open band is slid into place on the bat's forearm using thumb and forefinger. (d) The band is gently squeezed shut using slow and even pressure from thumb and finger tips. (e) The closed band should not pinch the wing membrane and be free to move up and down. (f) The gap **should** be sufficiently small (c. 0.7–0.8 mm) to prevent the band sliding over the wrist or elbow joint, and finger bones becoming trapped.

Removing bands

If the band is closed too tightly or closed unevenly it should be removed and a new one fitted. If any recaptured bats exhibit injury, or the band appears damaged it **must** be carefully removed using fine cir-clip pliers and a new band fitted to the opposite wing. Old bands **must not** be re-used because it is seldom possible to achieve a smooth even shape.

Bands can be removed reasonably easily using cir-clip pliers; however, the ends of the pliers need to be fine enough to insert inside the band. The tips of the pliers may require filing down if they are not fine enough. If no cir-clip pliers are available it is possible to use fingernails or two loops made out of nylon harp-trap line. One loop is slid under each edge of the band and the two loops are then gently pulled apart. If using either finger nails or loops it is very important to support the bat's forearm. It is necessary to have one person to hold the bat and support the forearm each side of the band, and another person to remove the band. The latter techniques are very fiddly and there is a higher chance of causing injury. It is far preferable to have a fine pair of cir-clip pliers as part of a standard banding kit.

Obtaining bands

The only type of band approved for use in New Zealand is a 2.9 mm (narrow) aluminium alloy flanged bat band manufactured by Porzana Ltd in Great Britain (Porzana@wetlandtrust.org). The design of the band is the result of many years experimentation and is characterised by a lack of sharp edges or burrs. **Bands will only be approved to those who have demonstrated a legitimate need to band, have appropriate permitting and ethical approvals, and can demonstrate they have had sufficient training to band bats, or have arranged for appropriate training**.

Band recording scheme

A full record of all bands applied to bats **must** be maintained, and as a minimum **should** include details of band number, location, date, age, sex and reproductive condition of the bat and name of the person banding. A copy of all banding records **must** be sent to the DOC Bat Recovery Group.

Long-term marking of long-tailed bats using other methods

New research and management projects may require alternative marking techniques for long-term monitoring. There are a variety of techniques that have been used on other bat species, but either they have not been trialled on long-tailed bats or have proven to be ineffective or unsuitable. Some of the more common methods are described below.

Coloured and plastic bands

Split plastic (celluloid) bands developed for individually marking birds have been used on bat species overseas. They are available in a number of colours and sizes and up to three have been applied to one bat. Bands are usually modified by filing the band gap wider and then smoothing and rounding the edges so the band is able to move freely up and down the forearm (Stebbings 2004).

Advantages:

- Bats may not have to be re-caught to identify individuals.

Disadvantages:

- Split plastic bands have caused injuries in some bat species in Britain (Stebbings 2004), and given that split metal bird bands have injured long-tailed bats, this may be an issue in New Zealand as well.
- Bands may only last up to a year, and the plastic can become discoloured or faded.
- Only a limited number of individual combinations can be achieved.

Plastic bands have been not been trialled on long-tailed bats, but given long-tailed bats' reaction to split metal bird bands, **plastic bands are not currently recommended as a long-term mark** for this species. As an alternative it may be possible to order specially coloured flanged metal bands.

Reflective bands

Reflective coloured tape can be applied to metal bands to aid in identification of individuals, or sexes during flight or in the roost (reviewed in Barclay & Bell 1988). Reflective tape greatly enhances the visibility of bands in artificial light, or with camera or image intensifiers. Reflective tape is available in a variety of colours. This technique has not been trialled on New Zealand bats.

Advantages:

- Red, white and yellow tape have the highest reflective properties and are generally easy to distinguish with a headlamp or spotlight and binoculars at ranges of up to 100 m.

Disadvantages:

- Illuminating a bat for a prolonged period either at the roost or during flight is likely to have a disturbing effect on its behaviour.
- Tape is unlikely to remain stuck to the bands for very long.

PIT tags

PIT tags, also known as microchips, have not been trialled on long-tailed bats, but they are being used on lesser short-tailed bats. The technique is therefore described in detail in the lesser short-tailed bat section below. The technique shows potential as an alternative technique for long-term marking long-tailed bats because the PIT tags have been used successfully in similar and smaller sized bat species (e.g. pipistrelles weighing 3–4 g, Bechstein's bats 7–8 g).

Necklaces

Necklaces have been used in some bat species where wing-banding has caused injury and infection, or where there is excessive band chewing. Bats were marked using necklaces made out of bead-clasp keychain or ratchet-style plastic electrical ties. Necklaces **must** not be used on growing juveniles. The ratchet-style electrical ties appeared to cause less abrasion around the bats neck and allowed for finer size adjustment (Barclay & Bell 1988).

Necklaces made of thin ratchet-style plastic electrical ties were trialled on six free-living long-tailed bats in 1993/1994. The ties were made of small knobbly beads and were covered in coloured electrical heat shrink tubing in an attempt to make the necklaces smoother and less likely to irritate the bat. Different colour combinations of heat shrink were used to individually identify each bat. The bats were recaptured at regular intervals over the summer season. By the end of the summer the necklaces had worn away the fur in a band around the necks of the bats. The skin was bare, but intact with no open wounds. All the necklaces were removed because banding was considered to be more acceptable for long-term marking of long-tailed bats (C.F.J. O'Donnell, unpubl. data). **Use of necklaces is not currently recommended as a technique for long-term marking of long-tailed bats.**

Long-term marking: lesser short-tailed bats

PIT-tagging is the only method currently approved for long-term marking of lesser short-tailed bats.

PIT tags (microchips)

PIT tags, also known as microchips consist of a small integrated circuit enclosed in a biologically inert glass capsule. PIT tags used in bats are commonly 12 mm long and just under 2 mm in diameter. For use as a long-term mark they are inserted under the skin (subcutaneously) using a 12 gauge needle. A PIT tag contains no power source of its own, but it is powered by a signal emitted from a reader. When the tag is interrogated by a reader placed close by, it responds by transmitting a unique serial number (Stebbings 2004). Handheld readers can detect tags from only 5–8 cm away, but fixed readers have an average detection range of 18 cm. It is also possible to use a loop antenna or a circular reader around a roost entrance or bat roosting box. Readers can detect relatively fast passage of a tagged animal past a reader, but the tag must pass within 7–18 cm of the reader (Beausoleil et al. 2004; Stebbings 2004).

Advances in technology are likely to result in further miniaturisation of transponders and development of sensor transponders that can measure a range of physiological parameters. PIT tags and readers are marketed by several companies, and not all tags and readers are cross compatible. It is advisable to contact the DOC Electronics Workshop to find out what types of PIT tags and readers are currently available and are in use New Zealand, and to find out more about new developments.

PIT tags are being used more widely as long-term mark for bats and in some species as an alternative to banding. Two relatively recent studies have shown the tags to be successful for examining roost use and social dynamics in Bechstein's bat, *Myotis bechsteinii* (Kerth et al. 2002), and for mark-recapture survival estimates in big brown bats, *Eptesicus fuscus* (O'Shea et al. 2004). In the latter study 2073 bats were tagged.

Advantages:

- Each tag has a unique code allowing a virtually unlimited number of bats to be individually marked.

- Tags are long lasting (> 10 years).
- Using automatic readers and dataloggers at roost sites provides opportunity for long-term monitoring without needing to recapture bats, and would be particularly useful for long-term survival studies.

Disadvantages:

- The procedure for inserting PIT tags is highly invasive, and may cause distress and pain.
- It is a difficult procedure to use on a small animal and requires rigorous training. There are currently only three approved trainers: Kate McInnes, Hannah Edmonds and Warren Simpson.
- PIT tags can sometimes migrate underneath the skin and can cause injury and pain, or be expelled.
- Initial outlay for the transponders, and in particular the readers, is relatively expensive.

PIT-tagging trials in lesser short-tailed bats

PIT tags were first trialled in New Zealand on free-living lesser short-tailed bats in Rangataua Forest. The tags were inserted underneath the backs of 100 bats. Their skin was very thin and care was needed to prevent piercing the skin twice. Transponders were found ejected at the base of roost trees, and three or four bats had serious arterial bleeds (B. Lloyd, pers. comm., Bat Recovery Group meeting minutes).

In 2006, 30 lesser short-tailed bats were tagged and held in captivity in a free-flight enclosure in the Eglinton Valley to monitor the short-term effects of PIT-tagging. There were still issues with piercing the skin twice, but this problem was largely solved by trialling different types of insertion gun. The small holes made by the needle healed quickly, and no tags came out of the bats after they were inserted. Three bats were recaptured 10 days and 3 weeks after they were released. Their tags were in place, there was no evidence of scarring, and the bats were healthy. Further details can be found in Sedgeley & O'Donnell (2006).

Further successful trials in 2007 (Sedgeley & O'Donnell 2007) have led to the use of PIT tags to facilitate standard monitoring of the survival of lesser short-tailed bats. Automatic data logging systems have been developed by the DOC Electronics Lab that record the presence of tagged bats at roost sites (see 'Instructions for setting up RFID readers, dataloggers and antennae'—docdm-379889) and survival of tagged bats has been shown to be very high (O'Donnell et al. 2011).

However, the technique is very exacting, and has the potential to cause serious harm. Therefore, anyone wishing to PIT-tag bats will be required to undertake very specific training.

Techniques for marking lesser short-tailed bats using PIT tags

Standard PIT tag for DOC work

DOC uses the Allflex PIT-tagging system (12 mm tags), using a 12-gauge needle on a Henke-jet injector gun for its bat work.

What is required to undertake PIT-tagging in bats?

Firstly the use of PIT tags on bats assumes staff involved in a long-term study on bats will have the appropriate skills for finding, capturing and handling bats as described in this manual.

Appropriate permits and Animal Ethics Committee approval **must** be obtained before starting a PIT-tagging project.

Anyone wanting to PIT-tag bats **must** also receive training from someone already experienced in PIT-tagging bats. At the time of writing only Kate McInnes, Hannah Edmonds and Warren Simpson from DOC can provide training.

We recommend selecting the appropriate person to learn to PIT-tag bats on the following criteria:

- Manager's approval
- They can commit enough time to training, as well as PIT-tagging sessions over several seasons
- Relevant skills (such as experience in PIT-tagging other species, taking blood from animals, vaccinating farm animals)
- Enough experience in handling animals, especially bats

If these last two criteria are not met, we strongly recommend training in needle insertion on animals with an experienced biodiversity ranger, veterinarian or farmer.

Training process

A minimum of three people are **required** to PIT-tag bats, one person to hold the bat (the handler), one to insert the tag (the injector) and the third to record data and prepare needles (the recorder). We recommend a fourth person to handle bat bags when dealing with large numbers of bats. Trainees should become familiar and capable with all aspects of the procedure, although we acknowledge that some people may not wish to inject bats.

This guideline is split into five sections that follow the recommended staged approach.

Assembling PIT-tagging kit

A standardised and well organised kit is essential. We recommend having enough equipment to make up three or four kit bags. Each bag and component items should be clearly labelled. Bags must be checked regularly to replace items which are used.

Before beginning tagging, trainees should familiarise themselves with the equipment and make up clearly labelled equipment bags.

Each kit bag should contain (Fig. 66):

- Two closed cell foam pads or fold-up chairs for PIT-tagger and handler to sit on.
- A dedicated bat bag for handling/restraint on the knee.

- One or two Allflex PIT-tag readers (to check if a bat already has a tag and to check if the new tag is functioning) and spare batteries (9 V).
- We strongly recommend using the Henke-jet steel insertion gun. This gun is safer to use than the disposable blue gun which comes with the PIT tags, which can jam if overused because they are intended to insert a limited amount of PIT tags.
- A large supply of PIT tags. The tags are inside the needles and are sealed inside sterile packets. Do not use a needle if the packet is open.
- Alcohol wipes to clean equipment and hands (between species and between sites).
- Thin gloves for handling bats.
- A 'sharps' container to safely hold used needles. Score a cross shape in the lid so that needles can be pushed into the container and not come out. This container is for use in the field only. Needles should eventually be disposed of in a proper sharps facility at the veterinary surgery or hospital/health centre.
- Rubbish bag (for all the spare sticky labels and used alcohol wipes).
- Recording sheets to record all new PIT-tag numbers and any recaptured PIT tags. See 'Transponder field recording sheet for new tags' (docdm-130625) for new bats, and 'Blank field recording sheet for transponder recapture' (docdm-130631) for recaptured bats.

Additional gear is required to catch bats at their roosts (harp trap, guy ropes, camera to count how many bats are caught) and lots of additional bat bags are required to hold the captured bats prior to tagging. It is useful to hang a rope between two trees and secure bat bags along it.

Figure 66. Some of the main components of a PIT-tagging kit: tag reader, clean bat bag, insertion gun, tags/needles, container to hold used needles, and alcohol wipes. Additional essential items are a spare 9V battery for the reader and recording sheets to record tag and bat details (photo: J. Sedgeley).

Learning how to hold lesser short-tailed bats for PIT-tagging

This section provides step-by-step instructions for holding a lesser short-tailed bat in the recommended position for PIT-tagging.

Holding the bat correctly is crucial to the whole procedure. A competent and confident handler makes the PIT-tagging process faster and more effective. Handlers should initially be adept at handling, ageing and sexing bats.

How to hold the bat in the preferred position for tagging:

1. The handler repositions the bat securely on their knee in the recommended tagging position. The best position for PIT-tagging is for the bat handler and the injector to sit facing each other (Fig. 67). Some PIT-taggers prefer the bat to be held on their own knee.
2. The bat should be held on the top (flat part) of the handler's left knee or thigh to ensure its back is flat for the injection. The posterior of the bat points outwards (Fig. 68). It is best if the handler's knee is positioned slightly higher up than the injector's knees so that the injector is level with the bat. If the injector is left-handed the bat should be held on the handler's right knee. Some injectors prefer the bat to be held on their own knee.

Figure 67. The best position for PIT-tagging is for the bat handler and the injector to sit facing each other. The bat needs to be on the top of the handler's knee or thigh to ensure its back is flat for the injection. The posterior of the bat should point outwards (photo: S. Bernert).

3. It is important to restrain the bat firmly with its head underneath the edge of a cotton bat bag to prevent it struggling or biting (Fig. 68). However, it is also essential to try to limit the holding grip to the bats wrists, elbows and feet and not to pinch any skin, particularly across its back. The skin must be as free and loose as possible and must not be stretched or tensioned.
4. The handler's fingers should be held so they do not interfere with the insertion process. Most handlers prefer to hold the bat's forearms and back legs with their thumbs and forefingers. It is important for the handler to ensure that their fingers holding the rear of the bat are kept as far

down the side of the knee as possible. This is essential to give clear access for the injector to grasp and manipulate the skin and to create enough space for the needle to be inserted.

Holding the bat correctly is crucial to the whole procedure. Do not rush and do not proceed with tagging until both the handler and the injector are happy with the position of the bat.

Figure 68.The best position to hold a lesser short-tailed bat during PIT-tagging. The bat's head is restrained underneath the edge of a cotton bat bag to prevent it struggling or biting. The holding grip should be limited to the bats wrists, elbows and feet to keep the skin as loose as possible. The fingers and hands of the bat handler must be held away from the bat to allow the injector clear access to the injection site (photos: top—J. Sedgeley; middle and bottom—L. McBride).

Learning how to inject PIT tags into bats

This section provides step-by-step instructions for injecting a PIT tag into a bat.

Before starting we recommend trainees should watch a short video clip of a lesser short-tailed bat being tagged by Jane Sedgeley and Kate McInnes (see docdm-131439).

How to inject a PIT tag:

1. Prepare the Henke-jet injector gun ready for tagging:
    a. Wipe the gun with an alcohol wipe.
    b. Setting the gun to ‘S’ for Allflex tags (twist the end of the gun to change settings, Fig. 69) is important, as other settings cause the needle to be pushed further in, resulting in double-punctures.
    c. Put a new needle/tag onto the gun. Needles/tags are supplied in sterile packets. If the packet is open the needle is no longer sterile and it must not be used on a live animal. Remove the needle from its packet and gently twist the needle onto the gun.
    d. The needle should not be uncapped until it is on the gun. It shouldn’t be totally uncapped until just before use, but if necessary the needle can be uncapped and left with the cap loosely on the needle to protect it while preparing the injection site. The cap of the needle should then be eased off slowly and carefully.
    e. The front of the gun **must** be twisted until the needle is aligned correctly so that the bevelled edge faces upwards (Fig. 69).

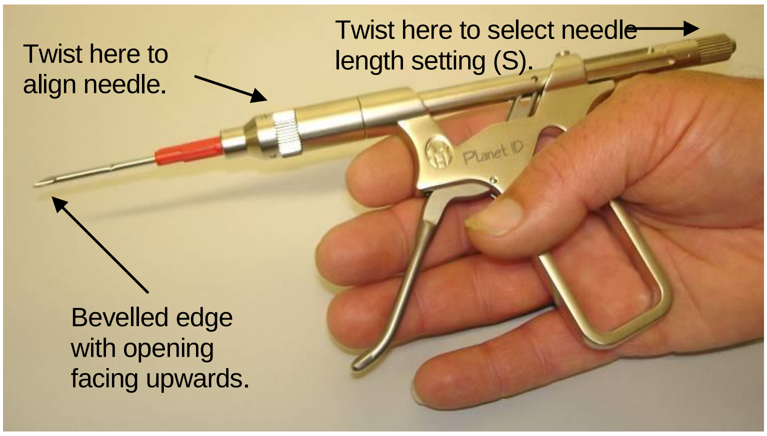

Figure 69**.** Correct positioning of needle in the insertion gun.

2. If the injector is right-handed, the injection gun should be held in the right hand and the skin of the bat manipulated using the left hand (and vice-versa).
3. The needle should be inserted from a caudal to cranial direction, i.e. from the tail towards the head, 2–3 cm along from the shoulder blades (Fig. 70). It is very important not to put the needle in too close to the bat’s head.
4. The needle should be held parallel to the bat’s back. If the needle is angled downwards there is a risk of piercing internal organs. If the needle is angled upwards there is the risk it will go into the skin and then pass out again creating both entrance and an exit holes (double puncture).

Some PIT-taggers rest their gun hand on the handler's, this steadies the hand and makes sure the needle is level.

5. The tag is inserted sub-dermally, i.e. beneath the skin and not into the muscle. Firstly, make sure the handler is holding the bat securely. Grasp or 'scruff' the bat's skin a few times to see what it feels like before attempting to tag it. Roll the bat's skin between their thumb and index finger. This action should loosen the skin and it should be possible to feel the difference between the skin and under-lying muscle. A minimum of skin is held pinched up between the balls of thumb and forefinger to form a tent to inject into. A small tent of skin minimises the risk of double puncturing as the needle punctures the skin almost immediately, whereas a large tent of skin means that the needle only punctures the skin after it is pushed about 5–10 mm under the finger grip. This then means the needle is very close to the back end of the fold and can easily go out the back side when the needle is inserted to the normal depth. If you pick up a small fold and insert the needle at the base, close to the back of the bat, the needle punctures the skin almost immediately, and therefore is nowhere near the back end of the fold when you insert it another 5 mm and squeeze the trigger.
6. The needle should be inserted below the fingers (so that the needle can not be felt by the pinching fingers). Insert the needle until it just penetrates the skin (5 mm) and the bevelled edge is inserted fully. Do not insert the needle as far as the small dimple on the shaft. Once the needle has been inserted correctly, the trigger should be squeezed gently and smoothly.

Figure 70. Close–up of aligning the needle with the back of the bat (photo: S. Bernert).

7. Once the trigger has been pulled, keep the same steady grip on the tent of skin and at the same time gently withdraw the needle. Often you can feel the tag under the skin and some injectors hold the tag in place whilst removing the needle.
8. The aim is to end up with the tag positioned between the bat's shoulder blades. It is difficult to judge the distance exactly when the skin is pinched up. It doesn't matter if the tag ends up a little low because once the needle is withdrawn the tag can be gently squeezed into place using fingers on the outside of the skin. If the tag ends up too high (i.e. too close to the bat's head) it is much harder to manipulate backwards.

9. The needle is then recapped, taken off the gun and pushed into the disposal container.
10. Sometimes tagging will not be successful, usually because either the skin was punctured twice causing the tag to come out, or there was a misfire. A misfire occurs when the needle does not puncture the skin sufficiently or the needle was not pushed far enough under the skin and the tag is not deposited under the skin. In any of these circumstances the bat should not be re-tagged.
11. Limit the time you will work with the bats and the number of chips to insert. Between 20 and 30 tags in a session seem about right before the injector gets tired and may make mistakes (misfires).
12. Compulsory 5-minute breaks with exercise, chocolate and a drink after 1 hour are essential.

PIT-tagging bats in the field

Important considerations

PIT-tagging lesser short-tailed bats is usually undertaken at roost sites at night. A harp trap is suspended outside the roost entrance, and bats fall into the trap as they exit the roost after dark.

The main aim of tagging in the field is to catch and process a large number of bats quickly and efficiently. Since tagging is undertaken in the breeding season it important to return breeding females and young back to the roost as soon as possible. Therefore, it is important to have a team of people dedicated to catching bats in addition to the PIT-tagging team. It is also useful to have several PIT-tagging teams, and for each team to be fully independent and have its own set of equipment.

This section provides:

- Basic guidelines for catching bats.
- A routine for processing large numbers of bats.

Figure 71. Field position for injecting lesser short-tailed bats.

Catching bats

It is recommended that a bat catching team is made up of 3–4 people, one person to haul the trap rope, one or two to control guy ropes, and one person to hold a spotlight. These people are also responsible for removing bats from the harp trap and putting them into bags.

The trap is positioned in front of a roost exit with the catch bag c. 30 cm below the entrance and bats are caught as they emerge on darkness. Once the trap is lowered, bats can be placed directly into cloth holding bags, up to five in each bag, and hung up on a string between two trees to be processed.

There can be up to a thousand bats occupying a lesser short-tailed bat roost tree. It is important that the numbers of bats captured should be limited to a maximum number that is feasible for the PIT-tagging teams to process, as well as appropriate for the size of the capture bag on the harp trap. A small harp-trap bag can hold up to 50 to 60 bats and the larger bags up to 80 to 100 bats. The high level of concentration required to tag bats means the process is very tiring, especially because the process is undertaken at night. It is therefore not practical to work for more than 1.5 hours at a time or expect to tag more than 20 bats in that time. It is important to return breeding females and young back to the roost as soon as possible and they should not be kept for more than 2 hours.

An infrared video camera linked to a screen, secured either to the trap or attached to a tripod or tree, can be used so that bats can be counted as they fall into the trap. Once the target number of bats has been reached the trap is lowered quickly to the ground. If more bats are needed for the sample, the catch bag is taken off and placed on legs near the bat processors. A new empty bag is placed on the trap and hoisted again to the roost.

Bats should be removed from the harp trap bags and placed in small holding bags. The number and noise of bats all running around in the bottom of the harp trap bag can create a sense of urgency, but care must be taken to ensure bats are not damaged in any way in the process of transferring them to smaller bags. This task should be limited to two to four experienced handlers only. There should be no more than five bats per bag. The bags are tied off securely and tied or pegged to a string line between two trees, near to the PIT-tagging teams.

PIT-tagging teamwork

A minimum of three people is required for processing bats in the field, but a larger team of four people is ideal. Teams comprise one person to hold the bat (the handler), one to insert the PIT-tag (the injector), one person to record data and prepare needles for the injector (the recorder), and a fourth person to provide the processing teams with bats, e.g. collect bats from harp-traps and handle multiple bags containing bats (the bat wrangler). Two teams of three people could share a seventh person who would collect bats and take them to the teams for processing.

The procedure for tagging bats in the field is almost identical to that of tagging bats indoors, but the process will only work smoothly if members of a team ensure equipment is well organised and a good routine is developed. It is essential to establish a strict routine in the field so that bats can be handled quickly and efficiently. There will be individual preference about who does each job and the precise order in which each job is carried out. Once a routine has been established (that suits a particular team of people) and becomes familiar it is important to adhere to that routine throughout the tagging session.

Procedure for tagging bats in the field

We recommend a routine similar to this:

1. Find a site away from the roost tree to minimise disturbance. The handler and PIT-tagger of each team should then find their own comfortable spot that is either level for chairs or close-cell foam (Fig. 71).
2. Lay out all equipment onto a piece of close-cell foam (or similar) to ensure equipment stays together and to prevent items getting lost on the forest floor.
3. Catch bats as described above, and hang bags containing bats on a string line between two trees.
4. The recorder takes the sticky label off the PIT tag and needle packet and sticks it on a prepared recording sheet (see 'Transponder field recording sheet for new tags'—docdm-130625 for new bats). Waste packets and spare labels are deposited into the rubbish bag.
5. The recorder uses a handheld reader to check the tag is functioning, and that its identification number matches the number printed on its label.
6. The bat wrangler (fourth team member if you have one) hands over a bag of bats to the bat handler.
7. The bat handler removes a bat from a holding bag, ages and sexes it and assesses reproductive status.
8. Alternatively, the bat injector can take the bat out of the bag; age, sex it, etc.; and then place it on the handler's knee. If time is an issue it is beneficial to use whoever is most experienced and quicker at handling.
9. Once the bat is on the handler's knee the recorder or injector checks the bat to see if it already has a tag using the Allflex handheld reader (also check for Trovan tags if there is a reader available—15 bats have Trovan tags).
10. If a bat already has a tag the handler/or injector examines the bat and the recorder notes the following information onto the recording sheet (see 'Blank field recording sheet for transponder recapture'—docdm-130631):
    a. Age, sex, reproductive status
    b. The tag position (they can move from their original location)
    c. The general health of the bat, any scarring or problems associated with the tag
11. If the bat is new and requires tagging, the recorder hands over a tag and needle to the injector.
12. If the tagging attempt is successful, the recorder uses a handheld reader to check the tag is functioning under the bats skin, and that its identification number matches the number printed on its label.

13. If the tagging attempt is unsuccessful, the recorder puts a line through the entry and records in the comment section what happened (e.g. double puncture or misfire). The bat must be released.
14. For each newly tagged bat the recorder must note all the following information against the correct identification label on the 'Transponder field recording sheet for new tags' (docdm-130625):
    a. Age, sex, and reproductive status
    b. In the notes column record whether the tagging attempt was successful, and if it was not successful note why it failed (e.g. double puncture, misfire)
    c. If the tagging was unsuccessful the label should be crossed out with an X through it
15. The bat is released.

Long-term marking of lesser short-tailed bats using other methods

Several other long-term marking techniques have been trialled on lesser short-tailed bats with little success. These are described below. To be able to develop new and more successful methods for long-term marking of lesser short-tailed bats, the problems associated with techniques previously trialled must be carefully considered.

Banding trials in lesser short-tailed bats

Captive trials

Five band types were trialled on six captive lesser short-tailed bats in the Wellington Zoo. The bands selected for testing were celluloid split bird band sizes C and D, British 2.9 mm flanged bat bands, an Australian 3.25 mm monel flanged bat band, and an Australian 2.8 mm alloy flanged bat band. Initially only one band of each of the five types was tested. The split bird-bands were fitted over the forearm but fed through a small slit made in the wing-membrane using a sterile scalpel. The metal bands were initially fitted using fingers and a pair of needle-nosed pliers. As the trial progressed it became apparent this technique was unsatisfactory, and specialised banding pliers were developed specifically for the task (Lloyd 1995).

The two sizes of split bird bands caused swelling, the C-band moved out of the insertion slit and the insertion slit enlarged to a hole 8 mm × 8 mm around the D-band. The Australian 3.25 mm monel band caused a hole in the wing-membrane and swelling after 16 days; the 2.8 mm band was not on the bat on the first inspection after 7 days. The British band caused no injury after 52 days. The results of this initial trial led to further trials concentrating on the British bat bands with a total of 15 fitted to the six bats over a 5-month period. Eight British bands did not cause noticeable injury; seven caused a variety of injuries including holes in wing membranes, and in one case a wing-membrane caught in the tips of the band so the wing couldn't open. Table 16 summarises the characteristics of bands which did not cause injury compared with those that did cause injury (Lloyd 1995).

Table 16. Characteristics of bands that caused injury to lesser short-tailed bats compared with characteristics of bands that did not cause injury (Lloyd 1995).

	No injury	Injury
Band tips	Flared outwards	Not flared outwards
Band closure gap	0.8–1.2 mm	< 0.7 mm
Even closure	Yes	No
Band fitting method	Bat-banding pliers	Fingers and ordinary pliers

British Mammal Society 2.9 mm bat bands were also fitted to 36 lesser short-tailed bats held in captivity on Codfish Island/Whenua Hou for 46 days during winter of 1996. The bands were fitted using fingers. No injuries were observed during the first 3 weeks in captivity but by the time of release a large proportion of the bats exhibited minor irritation of forearm tissue, and all bands were removed (Sedgeley 1996). In 1998, 385 captive lesser short-tailed bats were fitted with 2.9 mm British bat bands. These were obtained from Lambornes Ltd in Britain and were made of an alloy called 'incoly', a harder alloy than the magnesium-aluminium alloy Mammal Society bat bands previously trialled. All the incoly bands were removed after 1 week because a high proportion of the bats had forearm injuries in the form of abrasions, swelling and weeping (Sedgeley & Anderson 2000).

Banding trials in free-living lesser short-tailed bats

Large numbers of lesser short-tailed bats were banded in the 1980s in Northland, but after abrasions and swellings were seen on the forearms of banded individuals the method was discontinued (Mike Daniels, pers. comm., in Lloyd 1995). In 1996/97, 400 lesser short-tailed bats were banded in Rangataua Forest. Despite catching several hundred bats the recapture rate for forearm banded bats was < 1%, indicating either a very large population, or band-induced mortality (Lloyd & McQueen 1997). Thirteen bats were banded on Codfish Island/Whenua Hou in 1995 using 2.9 mm Mammal Society bands (S. McQueen, pers. comm.). Two of these bats, one male and one female, were recorded emerging from a roost on video (Sedgeley 1996), and one banded female was captured in 1998. The captured female had no observable injuries and the band was in good condition.

Current decision on banding lesser short-tailed bats

Forearm banding of lesser short-tailed bats is not currently an approved technique for long-term marking of lesser short-tailed bats and the Bat Recovery Group recommends banding must not be undertaken in this species until further notice (Bat Recovery Group minutes). Given the range of band types already trialled it seems unlikely an acceptable method of banding lesser short-tailed bats will be found. Some species of bat seem particularly susceptible to band injury which is why in several countries banding is approved for some species, but not others.

A combination of factors may explain why lesser short-tailed bats are particularly sensitive to the shape and closure width of the band, and are more prone to band injury and infection compared with long-tailed bats and other species. For example, lesser short-tailed bats seem have a tendon extending from the wrist and running beneath the forearm which is not usually found in other bats species (Fig. 72). Bands can occasionally become clipped around this tendon (J. A. Sedgeley, pers. obs.). If bands remain stuck in one place rather than freely moving up and down the forearm, there may be a greater risk of injury. Lesser short-tailed bats frequently forage on the ground and amongst leaf litter. These activities may cause bands to become clogged with soil and other debris, which may in turn contribute to increased levels of injury and infection.

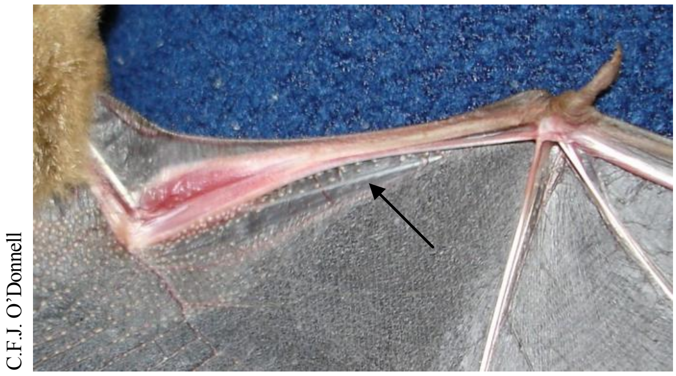

Figure 72. Wing of lesser short-tailed bat showing tendon immediately below forearm. The presence of this tendon can sometimes restrict movement in forearm bands.

Tattooing and freeze branding

Tatooing

Tattooing is potentially one of the most permanent methods for marking wildlife, but will depend on the application quality, and the depth and location of the mark (Beausoleil et al. 2004). Tattoos have been applied to bats by pin-pricking the wing-membrane and rubbing in dye (Barclay & Bell 1988; Heideman & Heaney 1989).

Advantages:

- Pinpricks can be made in different shapes, patterns, numbers or letters to identify individuals.

Disadvantages:

- Wing membrane tattoos seem to only last a few months.

Freeze branding

Freeze branding selectively destroys pigment producing cells in the hair follicles, resulting in the production of white hair or de-pigmented skin. Freeze branding has been used successfully to mark bats in the USA (e.g. O'Shea et al. 2004).

Advantages:

- If properly applied, freeze branding produces long-lasting clear and highly visible marks (Beausoleil et al. 2004).

Disadvantages:

- Freeze branding must be carefully tested in each species to determine optimum application time. Applying the brand for too long will produce extensive skin sloughing and scarring. Applying for too little time may result in too few white hairs being produced.

- Equipment and refrigerant materials can be dangerous to handle and impractical to use in the field (Beausoleil et al. 2004)

Tattooing and freeze branding trials in lesser short-tailed bats

Both of these techniques were trialled on lesser short-tailed bats held in captivity at Wellington Zoo. Both techniques were considered to be unsuccessful. Contact Brian Lloyd at DOC, Wellington for more details.

Necklaces

Necklaces have not been trialled on lesser short-tailed bats, but they are unlikely to be suitable for use in a species with terrestrial habits. The necklaces could get caught on vegetation, and at the very least get clogged up with dirt. **The use of necklaces in lesser short-tailed bats is not recommended.**

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Attaching radio transmitters

Radio-tracking is a standard technique for studying bats and a necessary precursor for inventory and monitoring and measuring the outcome of management. In this section we describe the reasons for radio tracking, the standards for attaching transmitters, how to track bats and briefly alert readers to analysis techniques. Radio-tracking is a specific technical skill; however, radio-trackers will also need to be skilled in identifying areas of bat activity to find locations in which to set traps, set up harp traps or construct mist net rigs; and handle bats competently. Training may be needed to learn how and where to place traps to optimise capture rates.

Reasons for radio tracking

Radio-tracking is a successful way of:

- Locating bat roosts so that bat workers can:
    - Identify important areas for management (such as pest control)
    - Count the numbers of bats using the site
    - Catch bats for marking or other purposes
- Determining home range size and designing adequate reserves
- Identifying important habitat types
- Identifying if development proposals might impact on bats (e.g. dams or wind farms)
- Measuring short-term survival (in relation to management; O'Donnell et al. 1999)
- Studying behaviour

Disadvantages and problems

- In bats, the biggest problem is keeping transmitters attached for long enough to last through the desired monitoring operation. Although battery life of transmitters potentially lasts for 4 weeks, in reality transmitters rarely stay attached for longer than 2 weeks.
- Sample size (e.g. the number of bats tracked or roosts found) is generally small because of the cost of buying transmitters and then following the animals—thus limiting the potential inference of studies.
- Individuals or age and sex classes selected for radio-tracking may not behave typically compared with all bats (e.g. an individual may not regularly forage and roost in the area where poison bait has been laid).
- Tracking is usually limited to summer when bats are easier to catch.

Radio transmitters

There are a range of small transmitters available that suit bats. The basic rule is that they should be as small as possible and not weigh more that 5–10% of the bat's weight so that it does not have to carry a heavy load.

Most people in New Zealand use transmitters from Holohil Systems[10], Carp, Ontario, Canada, because of their proven reliability but there are other brands available.

The standard Holohil transmitters used are 0.7 g transmitters (model BD2A with aerial length 160 mm). These transmitters have a battery life of about 28 days. When ordering ask for:

- Flat package (so it is does not sit too high on the bat's back)
- Deactivation by magnet (very easy to start)
- Reinforced base of aerial with c. 1 cm heat shrink to strengthen it (otherwise these bats will bite off the antenna)
- Receiver frequency range 160.121–161.110 MHz for New Zealand conditions. It is best to ask for 2-channel separation so that the frequency of one bat does not overlap the signals of others that are being tracked.

Although Holohil provide even smaller transmitters (0.35 g) they only last 7 days because of their small battery.

Before ordering transmitters check to see if there are other radio tracking studies in your study area. If there are, it will be important that you order transmitters with other frequencies than those already in use—it can be very frustrating if you end up tracking the wrong animal.

Attaching transmitters

Transmitters are attached between the scapulae (shoulder blades) on the upper back of the bats (Fig. 73).

They are glued on after the fur has been trimmed to make it shorter over an area the size of the transmitter. Gluing ensures that the bats don't have to carry bulky harnesses and ensures that the transmitter falls off. It also means that the transmitter may be recovered – and can be reused once a new battery is put in.

Transmitters are usually attached using a latex-based contact adhesive glue (F2®, Ados Chemical Co, Auckland, New Zealand), which is readily available at hardware shops. Other glues that are suitable are 'Skin Bond' or 'Vet Bond'. Do not use 'Super Glue' because it is carcinogenic.

The procedure for trimming the fur involves:

- Sitting the bat on a flat surface and holding its forearms firmly to immobilise it (Fig. 73). Short-tailed bats wriggle a lot, so make sure the grip is tight without squeezing it too hard. It helps to place the bat flat on a holding bag and fold one end over the head of the bat to calm it down.
- Trim the fur with very sharp scissors to ensure the transmitter grips, cutting an area no larger than the area of the transmitter. The fur should be a couple of millimetres long (Fig. 74).
- Use a cotton bud to make sure the surrounding fur does not overlap the area cut. Then apply a thin coat of glue (with the cotton bud) to the area of the bat's back as well as the underside of the transmitter.

[10] http://www.holohil.com/transmitters.htm

- Wait 4 minutes, then carefully place the transmitter on the back of the bat. It will grip straight away and the bat is ready for release.

Transmitters usually fall off after 2–3 weeks (maximum recorded = 28 days), although if the animals are moulting they may fall off earlier.

Figure 73. Illustration of lesser short-tailed bat with fur trimmed to the correct length (photo: J. Sedgeley).

Figure 74. The procedure for attaching a radio transmitter to a long-tailed bat (photos: top left—S. Bernert; remainder— J. Sedgeley).

Recording locations of bats

The signals of bats are picked up using a radio receiver attached to antennae tuned to 160 MHz. The most commonly used receivers are handheld TR4 receivers (Telonics, Mesa, Arizona, USA) with handheld, 3-element yagi aerials (e.g. Sirtrack, Havelock North, New Zealand; Fig. 75).

Bats can also be tracked by vehicle using a TR4 or a scanner-receiver (e.g. Advanced Telemetry Systems ATS, Isanti, MN, USA). Vehicle-mounted omnidirectional aerials can also be used (e.g. Tait Electronics, Christchurch, New Zealand).

Three-element yagi aerials are directional, so following the direction of the loudest signal will get you moving towards the bat. This can be a bit of an 'art form' where you learn many tricks for interpreting signal strength and direction. Learning how to radio track is something that has to be done in the field—there is no substitute for getting training in a real situation.

Finding bat roosts is best done by tracking with the aid of a bat detector at dawn when the bats often swarm around the roost cavity if it is warm. If you get to the roost in time you will see bats entering the roost cavity and this will save time later when you go to set up a bat trap or to do an emergence count Otherwise, you may need to climb the tree with the radio-receiver to try and locate the roost (following DOC's 'Roped tree work SOP'—docdm-159363).

The BD2A transmitter has a range of c. 400 m to several kilometres depending on whether the signal is impeded by surrounding landforms. We have recorded one bat nearly 5 km away when it was roosting near the bush line, but this is unusual. If a bat is high in a roost tree on slight or moderately sloping terrain then typically it can be heard 1–1.5 km away.

Figure 75. Tracking a bat with a 3-element yagi antennae (photo: S. Bernert).

Home range analysis

There are many analysis options for home range data (the point location and habitat data you may collect). Good general references include White & Garrott (1990), Kenward (1987) and Amelon et al. (2009). Examples of home range analysis for long-tailed bats can be found in O'Donnell (2002) and for lesser short-tailed bats in O'Donnell et al. (1999) and Christie (2003).

One powerful computer program for home range analysis is the Ranges analysis system (Kenward & Hodder 1996), which uses non-parametric techniques to describe range statistics. These techniques make no assumptions about the underlying distribution of fixes, and hence overcome potential problems of autocorrelation (Swihart & Slade 1985). Fully revealed home ranges (where plots of size of foraging area versus the number of fixes had reached an asymptote) can be expressed as Minimum Convex Polygons (MCPs) to facilitate comparison with other studies (c.f. Harris et al. 1990). The proportion of the range that could be defined as a core area (Harris et al. 1990) can be determined by plotting fixes against range size on a utilisation plot (Ford & Krumme 1979) using cluster analysis and determining the inflexion point of the curve.

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Taking tissue samples for genetic purposes

Tissue sampling for bats is covered by an approved standard operating procedure (see 'Tissue sampling for bats SOP'—docdm-929116).

The purpose of this section is to describe the tissue sampling SOP. The SOP has the following aims:

- To provide guidance to DOC staff or independent science providers commissioned by DOC who do not work for an institution that has an Animal Ethics Committee (AEC) and are planning to collect scientific tissue samples from native bats (long-tailed bat and lesser short-tailed bat) in the field.
- To provide a formal mechanism for ensuring that any impacts on manipulated animals are minimised during tissue sampling.
- To enable DOC staff to meet statutory species management requirements involving a significant amount of routine interaction with live animals without the requirement for DOC AEC approval.
- The SOP **must** be read in conjunction with the section 'Handling, examining, measuring and releasing bats' in this best practice manual.

Operators must seek separate AEC approval for any project involving manipulation of animals that does not constitute routine species management.

Reasons for tissue sampling

Tissue samples are taken for genetic analysis of individuals and populations. Results can help solve conservation problems and guide species management programmes by ascertaining:

- Genetic relationships within and between populations
- Genetic effects of population bottlenecks
- Taxonomic status and relationships, and evolutionary ecology
- Parentage
- Hybridisation
- Sex assignment of individuals, e.g. juveniles for translocation, adults lacking sexual dimorphism
- Metapopulation dynamics

When should a tissue sampling project proceed?

A tissue sampling project can only proceed if:

- There is clear benefit to the conservation, health, or welfare of the species. This is particularly important with:
  - Threatened and endangered species
  - Species inhabiting sites of significant conservation value
- There are no other means of obtaining the same information (e.g. work undertaken elsewhere).

- There is no useable tissue from the same individual or species currently in storage.
- Any potential negative effects on the conservation of the species are avoided, remedied or mitigated.
- Relevant parties are consulted where appropriate (e.g. Bat Recovery Group, DOC Wildlife Health Coordinator, iwi/rūnanga).

The project must aim to sample the smallest number of individuals required for the appropriate analysis.

Process

The SOP contains key information to assist project managers and *trained* operators to:

- Identify need for tissue sampling
- Seek separate AEC approval where appropriate
- Plan the project safely and appropriately
- Ensure relevant experience is accredited to operator
- Collect tissue samples from native bats with animal welfare being paramount
- Store and transport samples safely

Users of the SOP **must** note:

- **DOC staff:** Capture and holding of protected species by DOC staff as part of their normal, routine work duties does not require a permit under the Wildlife Act.
- **Non-DOC staff:** Other users of this document will need to ensure they obtain appropriate permits from DOC for the capture and holding of protected species and also obtain AEC approval for any research projects involving manipulations of these animals.

When to take tissue samples

Bat workers should avoid handling bats at inappropriate times of the year where potentially negative impacts have been identified. Impacts of handling and sampling can include:

- Compromising condition of a bat during winter (e.g. bats may not be able to afford the time it takes to recover from tissue sampling due to torpor)
- Compromising the condition of heavily pregnant females

Due to torpor during winter, it is necessary to sample bats during the breeding season. Care should be taken to ensure bats are processed as quickly as possible so lactating females can resume parental care.

Bats should generally not be handled in wet weather (due to them becoming cold, going into torpor and not being able to be released immediately at the site of capture). If a sample has to be collected under damp conditions, it would be considered an exceptional circumstance and should only proceed under artificial cover from rain and wind (e.g. tent-fly, tarpaulin).

Handling time

For each species, it is important for operators to:

- Minimise individual handling time
- Know how individuals are going to respond to prolonged handling and manipulation

Wherever possible, operators must ensure that multiple captures are housed appropriately while separate individuals are being sampled. Bats can be held in cloth holding bags, hung securely at a low height. To prevent overcrowding and aggression, the recommended maximum number of bats in a bag is ten for long-tailed bats and five for short-tailed bats.

If possible, attempt to sample bats shortly after dusk so they have adequate time to feed and recover following the procedure.

Training

The Bat Recovery Group leader must approve all trainers. Refer to the section on 'Permitting, ethics approval and training'.

Specialised training must be undertaken in order to minimise stress and avoid injury or death of bats during the sampling procedure. There is no prescription for attaining minimum standards; for example, a new bat handler will not be automatically approved to take tissue samples after they have taken samples on 10 occasions. New bat handlers should be able to demonstrate a minimum level of competency in the required technique to the trainer's satisfaction, and the amount of training required to reach this level will vary according to the skills and experience of individual trainees.

OSH requirements

All handlers must read and follow DOC's risk management and safety planning procedures. Handlers must also ensure they read the 'Health and safety' section for more details about the potential health risks of handling bats, particularly in relation to lyssavirus.

Tissue sampling protocol

Before commencing sampling:

- Ensure all equipment is laid out ready and close to hand.
- Pre-fill vials with 70% ethanol and keep several close to the sampling area in a secure polystyrene holder.
- Clearly designate and label a rubbish bag for swab, cotton bud and biopsy punch disposal.
- Make sure the hard surface that the sampling will take place on (Fig. 76) has been sterilised with 70% ethanol, and is relatively smooth and undented (e.g. an ice cream container lid).
- Remove a brand new biopsy punch from its packet and position as close to the workspace as possible. One biopsy punch will last for an average of 20, and up to 30, punches before it

becomes blunt. Once a biopsy punch has been used for 30 punches it **must** be changed for a new one.

Keep surplus cotton buds, biopsy punches and sterile tweezers somewhere with easy access.

Prevent further captures

Closing or removing trapping devices (e.g. mist net, harp trap) after capture is essential for the following reasons:

- To prevent captures of excessive numbers of bats if not enough personnel are present to attend to them.
- Failure to do so could severely compromise the safety of any excess bats trapped, and of the bats being sampled under time pressure. This is particularly true for potentially large roost captures of lesser short-tailed bats, which require faster trap management than long tailed bats.

Identifying suitability of bats for tissue condition

Before commencing sampling:

- Identify bat (check band number or PIT tag) and check that the individual is required for sampling (e.g. they have not been sampled before—previous punches heal in a short period of time and are hard to recognise).
- Assess condition. Bats that are in poor condition (e.g. less than 85% of mean weight for species), mothers with young attached to the nipple, heavily pregnant females, and juveniles in their first 4 weeks of flight should not be tissue sampled.
- Perform any other necessary manipulations (e.g. morphometrics).

Figure 76. Taking a tissue sample from a long-tailed bat.

Assistant handler(s)

At least one experienced handler must be employed, in most cases, to:

- Restrain the bat firmly and confidently to prevent any movement during tissue collection (Fig. 76).
- Stretch out the wing to its full capacity to provide a taut surface for the biopsy punch.

There are no circumstances when operators can work alone or without assistance as it is not an approved or safe practice.

Minimising stress during restraint

Handlers can make restraint during tissue sampling easier and minimise stress to the bat in the following ways:

- Hold the bat steady with its back against the prepared hard surface.
- Inform the operator if the bat is about to struggle.
- Process the bat as quickly and efficiently as possible.
- For lesser short-tailed bats, the handler should wear a polypropylene glove on the non-dominant hand to prevent being bitten. If the bat does happen to bite the handler, the handler must ensure they do not withdraw their hand as this can cause injury to the bat's teeth. Blowing on the bat will encourage it to stop biting.

Lesser short-tailed bats appear to be more sensitive to prolonged and insensitive handling than long-tailed bats. Very occasionally lesser short-tailed bats will convulse during a lengthy handling period. If this happens, the handler **must** immediately cease examining or sampling the bat, place it somewhere quiet to recover (e.g. in a cloth holding bag by itself) and then release it as soon as possible.

Wing biopsy procedure

Handler: Hold the bat face up against the sterilised hard surface with non-dominant hand (Fig. 76). The dominant hand stretches the wing out to its full capacity, parallel and as close as possible to the hard surface.

Operator: Remove protective cap from the biopsy punch and unscrew lid from a prepared ethanol vial:

1. Press the biopsy punch firmly down on the main part of the wing, between the fifth finger and the body and avoiding major blood vessels. The wing should be completely flat, and the punch completely vertical.
2. Twist the punch gently 360 degrees both left and right, ensuring the blade has completely punctured the wing membrane. If necessary, the handler can lift the bat's wing very slightly from the cutting board to check that the punch is all the way through the wing membrane.
3. Carefully remove the biopsy punch from the wing. The sample will either be lodged in the punch or stuck on the cutting board. If it is the latter, the handler must be very careful to avoid moving the sample as they remove the bat from the sampling area.

If sample remains in biopsy punch
Place the punch in one of the prepared ethanol vials and shake the punch gently to dislodge the tissue. If the tissue is wedged then a pipette can be used to flush 70% ethanol through the punch, dislodging the sample. Once the sample is in the vial, sterilise the punch by shaking it again in the ethanol vial.

If sample remains on cutting board
Remove the sample with sterile tweezers and place it in an ethanol vial. Sterilise the tweezers by either shaking them in the ethanol vial or wiping them down with a cotton bud moistened with 70% ethanol.

Sample each wing
It is recommended that operators take a sample from each wing and label the separate vials appropriately (e.g. Left and Right). This provides a backup should one sample be misplaced. Some operators may also choose to swab the wing membrane with 70% ethanol prior to taking the sample, although this is not a necessity.

Hygiene

Basic hygiene measures to prevent the spread of infectious diseases and parasites between individuals and populations include:

- Washing hands, or using medical hand wipes (if no water available) between bats handled/sampled
- Discarding and replacing holding bags as soon as soiled (e.g. faeces, blood) during the sampling operation

The cutting board **must** be thoroughly swabbed down with 70% ethanol between bats, and the tweezers and biopsy punch sterilised also.

After sampling, bats are occasionally too cold or stressed to be released immediately. Operators should consider steps they will take in these situations, such as:

- Placing cold or torpid bats in a cloth handling bag and keeping somewhere warm (e.g. inside the handler's jacket) until they have sufficiently recovered
- Stroking the tail of reluctant bats to encourage them to fly
- Keeping a spotlight trained on all releases or a bat detector switched on to ensure they fly correctly and do not fall to the ground

Bats should preferably be released from the capture site, while being held up high in an area with few obstructions.

Healing times

Wing holes are typically fully healed within 3–4 weeks, without impairment to flight or reproductive success, and can only be identified by a small pale-coloured patch on the wing. Juvenile bats heal particularly quickly and may even heal without any discolouration.

Labelling samples

It is vital that all tissue samples are labelled with the following information:

- Individual ID (e.g. PIT tag, band number)
- Date of collection

This is to ensure the label contains information that relates back to the electronic database. Other helpful labels can include:

- Colony group
- Left or right wing
- Location

Vials should be labelled with sticky labels that wrap around the entire vial, to ensure they do not fall off. A fine-tipped indelible marker or sharp pencil can be used to write on the label. Note that some marker pens can run and become illegible in contact with alcohol.

Storage of samples

Samples should be kept in 70% ethanol and refrigerated. If samples are sent overseas, the International Air Transport Association provides guidelines for shipping dangerous substances, including alcohol (classified as a Class 3 flammable liquid). General requirements dictate that samples are triple packaged, with one layer containing enough absorbent material to absorb the total quantity of ethanol.

A customs declaration is also required, containing the following information:

- Species
- Number of samples
- Quantity and strength of ethanol
- Commercial value of samples

A Wildlife Act Permit for shipping overseas may also be necessary.

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Guidelines for temporarily keeping bats in captivity for research purposes

This section was originally developed for DOC Te Anau Area Office to provide guidelines for researchers seeking permits to hold lesser short-tailed bats in short-term captivity.

Permitting requirements

In addition to normal permitting requirements:

- Applicants must demonstrate willingness and ability to keep bats in captivity using the captivity guidelines below.
- A full report of captivity procedures including number of bats kept, numbers released, husbandry techniques (amount of food, housing) health, any deaths and causes must be provided at the end of the work.
- A report summarising preliminary research findings should be provided at the end of field work. Copies of final research findings and any published results should be provided at a later date.

Important considerations

All bats **must** be aged, sexed and their reproductive condition assessed before they are taken into captivity. Pregnant, lactating bats and dependant young **must not** be taken into captivity during the breeding season *except under exceptional circumstances*. For example, pre-approved and permitted captive breeding projects or transfers.

Requirements for keeping bats in short-term captivity

When bats must be kept in captivity for research purposes it is acceptable to hold them in cloth bags over 1 night, but they must be housed in more spacious holding boxes and exercised if they are to be kept for longer. If bats need to be kept for more than 3 days bats must be held in a free-flight enclosure.

1. Keeping bats in cloth bags

Aim to keep bats in bags for as short a time as possible.

Unless bags are brand new, they must always be thoroughly washed and disinfected between different study areas and if they have been used previously to hold other species. Bags should be washed using a disinfectant detergent (e.g. TriGene), and rinsed thoroughly. This can be done in a washing machine.

A bat may be kept in a bag overnight (which almost always means the next day as well) providing it is to be released the following night. Bags should be kept somewhere quiet with low light. During warmer months, care must be taken to ensure bats do not become too hot or dehydrated. The bottom of the bag should be moistened to provide humidity. The bat must be offered food and water during this period (see feeding methods for both species below).

It is preferable to keep only one bat in each bag to ensure it gets enough food and to avoid any aggression. Two bats could be kept together in larger bags.

It is fine to have several bats in a bag just prior to release. In fact it often makes release easier because the bats will warm each other up and will be more likely to be active and ready to fly.

2. Keeping bats in wooden cages

If a bat is to be held for more than 1 night, it must be kept in specially designed holding box. Boxes must be well ventilated, dark and either have grooves in the walls or a suitable material such as shade cloth fixed to the walls to allow bats to grip while roosting. Soft material can be attached to the wall so the bats can choose to roost under the cloth.

Store the box in a quiet location with appropriate ambient temperatures. Generally, for most research purposes it is easier to keep bats warm and active. For bats to remain active they should probably be kept at temperatures ranging from 25–28°C. In very hot conditions it may be necessary to provide extra humidity by placing a dampened cloth or paper on the floor on the box.

DOC Te Anau Area Office has some wooden bird transfer boxes that can be adapted for use with bats by stapling cloth or mesh on at least one inside wall of the box (Fig. 77). These boxes are 40 cm long, 28 cm wide and 20 cm high and are divided into two compartments with a removable partition (i.e. each compartment is 20 cm × 28 cm × 20 cm).

Holding boxes must also be carefully disinfected between use by birds and bats. Fig. 78 provides an example of a design commonly used in the UK and recommended in the UK *Bat Workers' Manual* (2004).

Figure 77. Bird transfer box adapted for use by bats (each compartment contains a small bowl of water and a separate bowl with meal worms).

We have kept up to five bats in each section of the bird transfer boxes shown in Figure 77, but only held that many bats for 1 night. We do not recommend holding more than three bats per side for longer periods. We have kept same sex bats together and also mixed males and females together. However, for mixed sexes we would try to keep the sexes balanced, or more females than males.

It important to note that whilst we had no problems keeping bats under the conditions described above, it is likely because we only kept them for between 1 and 3 nights and provided them with food and water *ad libitum,* i.e. there was little competition for food.

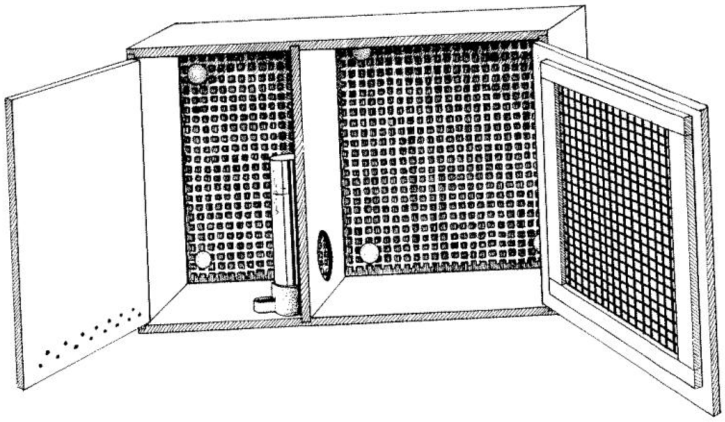


Illustration ©T.P. McOwat, the JNCC *Bat Workers' Manual* (2004)

Figure 78. Design of bat holding box commonly used in the UK. Note that water is provided using a water-feeder that is used for feeding captive birds.

3. Feeding and providing water to captive bats

Captive bats must be provided with food and water every day, even if they are held for only 1 day. If bats are kept in holding boxes or a free-flight enclosure, food and water can be provided in separate dishes. If bats are kept in bags, mealworms can be dropped directly into the bottom of the bag, but water will need to be given by hand. Water can be offered from a damp cotton wool bud or using a pipette/eye dropper. When offering water by hand, ensure the bat is held with its feet higher than its head to avoid the bat accidentally ingesting water into its lungs. Long-tailed bats will usually need to be fed on mealworms by hand.

Bats will only eat if they are active. For bats to remain active they should probably be kept at temperatures ranging from 25–28°C. If they are kept in cooler conditions they may enter torpor and will need to be warmed up before they can eat.

Short-tailed bats do not need to be taught to recognise mealworms because beetle larvae form a natural part of their diet in the wild. Therefore, they do not usually require hand-feeding and will generally take mealworms direct from feeding dishes or the bottom of a bat bag. They will also take water directly from a dish. If the bat is held inside a bag it will have to be offered water by hand using a damp cotton wool bud, a small spoon, or by using a pipette/eye dropper.

Long-tailed bats are aerial insectivores feeding primarily on moths and other flying insects. They do not recognise mealworms as natural food and will therefore require hand-feeding. It is possible to train bats to recognise mealworms as natural food and to feed themselves, but this may not be possible in the relatively short time period of temporary captivity. Guidelines for feeding and providing water to a long-tailed bat are as follows:

- Take the bat out of its bag or holding box and if necessary gently warm it up by holding it in cupped hands. It will not eat or drink until it becomes sufficiently active.
- Once the bat is active hold it in one hand and offer it food and water. Ensure the bat is held with its feet higher than its head to avoid the bat accidentally ingesting water or fluids into its lungs.
- Water can be offered from a damp cotton wool bud, small spoon, or by using a pipette/eye dropper.
- When offering a mealworm, firstly cut its head off and hold the cut end of the body to the bat's mouth using tweezers. The bat should lick the soft insides of the mealworm and might eventually grab it out of the tweezers and chew it up.
- Mealworms should be offered one after the other until the bat refuses to eat any more.
- The bat should then be replaced inside its bag or holding box and the container put in a warm place (so the bat stays active to digest food).
- Live mealworms should be placed inside the container used to house bats, either directly into the bag, or inside a dish for bats held in boxes or in an enclosure. A note should be made of the number offered so it is possible to see if the bats have learnt to feed themselves.

We found the best approach was to provide food *ad libitum* so bats could regulate when, how often and how much they wanted to eat. In the boxes we provided large quantities of mealworms spread among two or three bowls and a constant supply of plain water in a separate bowl.

As a general guide a lesser short-tailed bat can be expected to easily eat between 10–20 large mealworms per day, and will often eat many more. They will eat fewer in colder conditions and more in warm conditions and during the breeding season. The mealworms offered to the bats should be counted before and after feeding to determine how many are being eaten per session.

If kept for no longer than 7 days, bats can be fed on ordinary commercial mealworms. However, if they are to be kept for longer, bats need to be given vitamin and mineral supplements. This is most easily achieved by feeding bats with nutrient enriched mealworms. Mealworms should be taken from their bran medium and transferred into a container of Wombaroo® insectivore mix. The mealworms should be held in this mix for a day, and then taken out and fed to the bats.

4. Exercise

Bats can rapidly lose condition if kept in captivity with no exercise. If bats are to be kept for longer than three days they should be allowed free flight daily where possible. Bats could be exercised in a room and then recaptured to return them to their holding boxes, but it is likely the bats will get stressed. It is preferable to keep bats inside a free-flight enclosure (that includes roost boxes) so they can exercise at night under more natural conditions.

5. Longer-term temporary captivity

It is important to note that most of the information on keeping bats in longer-term captivity comes from experiences with keeping lesser short-tailed bats. We have very little experience to date in holding long-tailed bats in flight cages.

If bats are to be kept in captivity for longer than 3 days it is preferable to keep them in the free-flight enclosure at Knobs Flat (Fig. 78). Roost boxes are available, but food and water dishes will need to be provided (large Pyrex® or glass baking dishes work well). The enclosure can comfortably hold 30 to 40 bats at a time. To avoid competition this many bats should be provided with at least two or three roost boxes, and several food and water dishes.

Figure 78. The free-flight enclosure is located in the forest behind Knobs Flat Field Station. It is large enough for the bats to fly around inside, it is approximately 10 m long × 5 m wide × 2 m high. The enclosure contains vegetation to provide bats with a natural environment and a choice of several roost boxes, feeding and water dishes.

6. Monitoring weight and health

It is recommended to have an experienced bat keeper or vet present if bats are to be kept for longer than 3 days.

Bats held in short-term captivity must be weighed daily, and those in longer-term captivity must be weighed weekly. The bats should also be checked for signs of ill health (e.g. lethargic, dull sunken eyes) or injuries (e.g. bites, wing tears).

Weights should be carefully monitored and food intake regulated if necessary. If bats gradually lose weight they should be offered more food. If a bat does not eat while in captivity, or eats but continues to lose weight over 2 days, it should be released. Water should be offered immediately before release using a cotton bud or pipette/eye dropper.

Sometimes an individual bat will lose weight, or not put on weight at the same rate as other bats held with it. It may be necessary to isolate this bat from others to ensure it gets enough food.

For temporary captivity it generally seems best to keep bats warm and active and make food freely available. However, in some cases bats will dramatically put on weight, particularly if they have little exercise. In these cases it may be necessary to cut back food. This has not generally been a problem for lesser short-tailed bats held in bags or boxes for a few days or free-flight enclosures for up to a month.

7. Limiting research procedures

To reduce stress on the bats, restrict the number of research procedures to only one per day.

8. Releasing

Bats should be released as close to point of capture as possible. Unless there are unavoidable circumstances, bats should always be released either at dusk or at night. If it is not possible to release a bat at these times it is preferable for the bat to be placed in an artificial roost box, or in a crevice or tree hole so it can emerge by itself when it gets dark.

Bats need to be warm and active in order to fly. If a bat becomes torpid and is unwilling to fly, it should be warmed for a few minutes before release. Bats can be warmed up by holding the bat inside loosely cupped hands, or popping the bat inside a bag and placing it under clothing and next to warm skin.

9. Feedback and reporting

This document provides general guidelines for keeping bats in captivity. Feeding and exercise requirements will vary among individuals and species and at different times of year.

It is important, therefore, to carefully document the exact procedures used for captivity including number of bats kept, how they were housed, amount of food and exercise and any health issues or deaths. Ongoing feedback is important for assessing how well the captivity protocols are working and if any adjustments are necessary.

10. Useful documents and links

- Useful information on first aid care: http://britishwildlifehelpline.com/index2.html
- UK *Bat Workers' Manual*: http://www.jncc.gov.uk/page-2861
- DOC's 'Wildlife health SOP' (olddm-766252)

Permitting, ethics approval and training

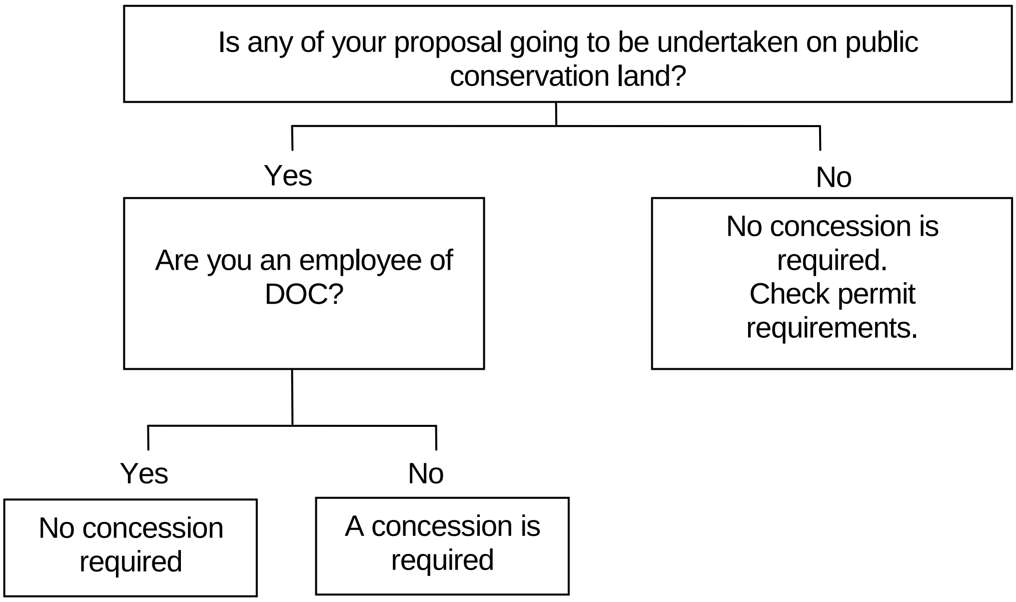
Permitting

Do you require a permit?

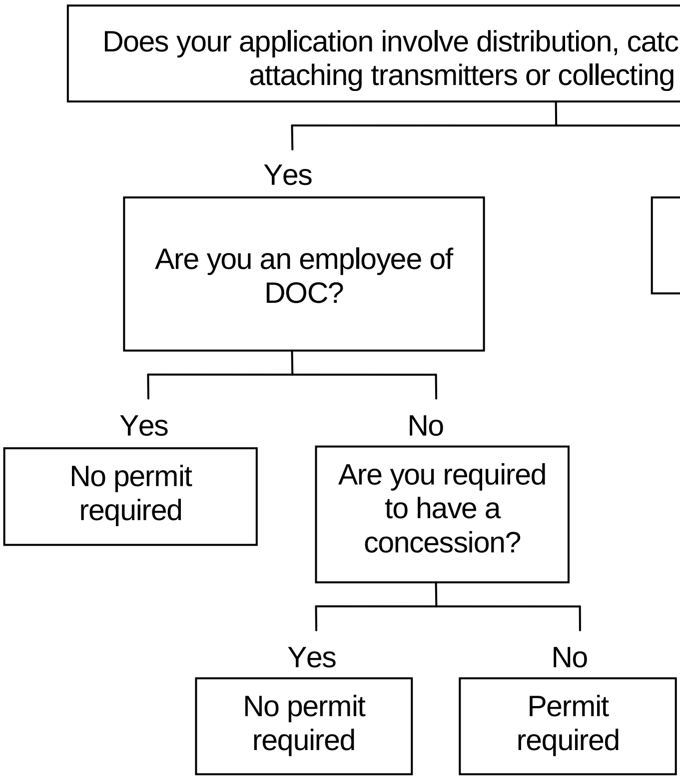
All New Zealand bats are protected under the Wildlife Act 1953 and authorisation (by way of permit or concession) from DOC is required if your proposal involves disturbing, catching, and handling, etc. If you are an employee of DOC then this authorisation is not formally required as you are ‘acting in good faith in pursuance of your duty’. **DOC employees do not require permits.** The only possible exception to this would be if a banding scheme similar to that used for birds was developed, but no scheme currently exists.

The schematics below are to assist with determining if you need a concession or a permit.

How to determine if you require a concession



How to determine if you require a permit



Role of the Bat Recovery Group in permitting

The Bat Recovery Group is an advisory group and is not a permitting authority, and if required can provide advice to conservancies on applications to manipulate bats. Generally the Bat Recovery Group will support proposals if the proposed study/project:

- Enhances knowledge of bats
- Benefits bat conservation
- Does not unduly impact on bats, or impacts are low
- Uses only approved techniques (animal ethics committee, and this manual)

Animal ethics approvals

DOC's obligations under the Animal Welfare Act

The DOC Animal Ethics Committee (AEC) meets DOC's obligations under Part 6 of the Animal Welfare Act. When animals are manipulated as part of an approved research, testing or teaching project, Part 6 of the Animal Welfare Act applies. This recognises that compromised care and some pain or distress to a small number of animals may result in significant benefits to people, other animals or the environment. However, such use carries with it significant responsibilities and strict legislative obligations. Every proposed project is subject to scrutiny and approval of the AEC.

The functions of the AEC are:

- To consider and determine on behalf of the Director General of DOC (referred to in the legislation as the 'code holder') applications for the approval of projects
- To consider and determine, under section 84(1)(a), applications for the approval of projects
- To set, vary, and revoke conditions of project approvals
- To monitor compliance with conditions of project approvals
- To monitor animal management practices and facilities to ensure compliance with the terms of the Code of Ethical Conduct
- To consider and determine applications for the renewal of project approvals
- To suspend or revoke, where necessary, project approvals
- To recommend to the Director General amendments to the Code of Ethical Conduct

Section 80, which sets out the purposes of Part 6, should be read carefully by all those involved in the use of animals in research, testing and teaching. The section provides guidance on the circumstances under which animals can be manipulated. Such guidance is particularly important for the AEC when they are considering project proposals. Principles include:

1. The findings of the research, or testing or the results of the teaching will enhance understanding of humans, animals, or the natural or productive environment.
2. The anticipated benefits of the research, testing, or teaching outweigh the likely harm to the animals.
3. Any research, testing or teaching involving the use of a non-human hominid is in the best interests of the animal, or is in the interests of the species to which the animal belongs and the benefits outweigh the harm to the animal.
4. Where animals are ill or injured they must receive, where practicable, treatment to alleviate unreasonable or unnecessary pain and distress caused by illness and injury, except where this is not possible because of the nature of the work, in which case any pain or distress must be reduced to the minimum possible in the circumstances.

Applying for Animal Ethics Approvals for bat work

Operators must seek separate AEC approval for any project involving manipulation of animals that does not constitute routine species management.

Bat workers within DOC **must** follow DOC's procedures for applying for Animal Ethics Committee approval.

Where external bat researchers are applying for permits under the Wildlife Act/Conservation Act, DOC **must** ensure they have appropriate ethics approvals if necessary.

Recent examples include approvals for banding bats, inserting transponders and tissue sampling.

Do I need AEC approval?

DOC has a flow chart to assess whether Animal Ethics Committee approval is required (see ‘Application flow chart’—docdm-232655). However, if bat workers are uncertain whether they need to apply, they should approach the current chair of the AEC via DOC’s National Office.

How to apply to the AEC

Proposals require identification of the type and number of animals to be used, the nature of the manipulations, justification for the use of animals and the species and numbers involved, provisions for the care of animals and steps to be taken to minimise any distress or suffering.

Applications must be written in non-technical language. The application **must** be easily understood by an intelligent lay person.

AEC application deadlines

Applications for consideration by the AEC must be submitted by the 25th of any month—except December. There will not be an AEC meeting in January.

Application form

The DOC Animal Ethics Application form to manipulate live animals is available—see ‘AEC application’ (docdm-737250).

Training and competencies

Acquiring training and demonstrating competences

Before approval is granted by the relevant conservancy it will be a condition that all applicants will have met minimum levels of competency in approved bat conservation research techniques relevant for the proposed project. DOC employees will also be required to meet the same level of competency.

Minimum competencies may be obtained in two ways, either on an existing bat conservation programme/project, or on the applicant’s own project. Gaining competences on an existing project will be reliant on whether an appropriate project is running at the time of application. Therefore, it is recommended that the applicant budget for an approved operator to spend time training the applicant on the applicant’s project. This approach may better allow the necessary skills to be targeted.

The applicant will be required to demonstrate that training has been either been undergone, or has been arranged (e.g. a letter of recommendation from an approved trainer) or be able to demonstrate prior learning/experience. It is important to note that many techniques used on bats are specialised and it does not automatically follow that techniques used on birds are applicable to bats. For example, the technique for banding bats is very different to that of banding birds, so even if a person is an experienced bird-bander they will still require specialised training to band bats.

The minimum levels of competency are only required where the operation involves catching, handling disturbance, etc. It is not mandatory to demonstrate competencies where disturbance or handling is not required (e.g. survey, as there is no risk to bats). However, it is advisable to read relevant sections of this manual and where possible have the techniques demonstrated to ensure that you have the greatest chance of success with your proposal.

List of potential trainers

This list provides names and contact details of people with skills necessary for training in certain techniques. It should not be assumed these people will always be willing or available to offer training.

Name	Skills	Contact details
Colin O'Donnell	Most techniques	DOC RD&I, Christchurch codonnell@doc.govt.nz
Kate McInnes	Transponders	DOC RD&I, Wellington kmcinnes@doc.govt.nz

Standards of competency

Listed below are examples of techniques commonly used in bat research that involve handling and disturbance of bats and as such require permitting. Specialised training **must** be undertaken in order to minimise stress and avoid injury or death of bats. There is no prescription for attaining minimum standards, i.e. a new bat worker will not be automatically approved to mist net after they have extracted a bat from a net on 10 occasions. New bat workers **must** be able demonstrate a minimum level of competency in the required technique to the trainer's satisfaction, and the amount of training required to reach this level will vary according to the skills and experience of individual trainees.

Examples of techniques requiring training:

- Extracting bats from mist nets
- Use of harp traps at roost sites
- Handling bats
- Take morphological measurements, age, sex reproductive status bats
- Forearm band (long-tailed bat only)
- Temporary marks (fur-clipping)
- Wing biopsies for genetic sampling
- Attaching transmitters
- Inserting transponder tags (also known as microchips or PIT tags)
- Release techniques

Additionally, we recommend a working knowledge of the following:

- Capture techniques (setting mist nets, positioning harp traps)
- Acoustic survey techniques, ability to distinguish between echolocation calls

- Radio-tracking

Health and safety

Requirements for health and safety plans

DOC staff working on bats must ensure that they operate under approved safety plans that are produced through the Risk Manager system.

Risk Manager[11] is DOC's primary health and safety management tool. The 'Risk Manager user manual' (docdm- 611915) provides detailed instruction on how to use the web-based system, module-by-module, and starts with a basic introduction to navigating around the system.

The use of Risk Manager is mandatory to manage hazards, develop safety plans, and record and investigate incidents.

DOC safety plans are generally approved by the Area Manager in the area that the work is being undertaken. External researchers working under other auspices must have an approved safety plan from their parent institution. However, Area Managers should assess these when processing applications to undertake research on DOC-administered land or protected species.

It is important that hazards relating to bat monitoring are properly identified, assessed and controlled and linked to safety plans. These hazards should be reviewed following any incident.

Some Australian bat workers have formulated useful risk management protocols for working in confined spaces, which is worth reading (see Armstrong & Higgs 2002).

General hazards relating to working in the field almost always apply to working on bats. However, several hazards specific to bat-related work are emphasised in Table 17.

[11] www.riskmanager.co.nz

Table 17. Important hazards relating to working on bat projects.

Hazard	Description
Visiting bat roosts and catching bats at night in the forest and/or in caves	People working at night should generally work in pairs. The only exception is when undertaking bat roost counts < 200 metres from the road and if the roosts are on marked tracks. If undertaking a roost count at night, never deviate from the agreed-upon track, fill in the intentions board, and carry a Personal Locator Beacon (PLB), handheld radio, first aid kit and appropriate clothing. Individuals should always carry at least two torches and a spare battery in case of failure. No river crossings should be undertaken alone.
Climbing trees and/or in caves or limestone cliffs to find bat roosts and/or set up harp traps	Climb according to DOC Standard of Practice. New staff must be trained to a standard of competency and by an authorised DOC instructor (as per SOP). Climbing equipment upgraded according to SOP and to be of UIAA standard. One person to be climbing at a time. At least one more person on ground, trained in rescue techniques.
Radio-tracking bats at night from a vehicle	Maintain roster of night workers to avoid over-tiredness. Shifts should be no longer than 3 hr. Current Warrant of Fitness, registration, appropriate driver licenses; regular vehicle servicing; first aid kit carried.
Rigging trees with sling shots and ropes or strings for mist nets, climbing and bat traps, which may result in falling objects	Approved NZSS/UIAA standard safety helmets to be worn on ground at all times. Correct knots and pulley systems to be used and security tested by designated safety supervisor. All equipment to be of high quality. Safety visors for people using slingshots.
Bites received from handling bats resulting in possible infection including very remote chance of rabies-related lyssavirus	Follow Bat Recovery Group guidelines. Restrict number of personnel handling bats. Gloves to be worn, except when necessary to have bare hands (attaching transmitter or extracting from mist net). First aid kit present. Rabies injections for staff handling bats frequently.

Disease risk to human health

Lyssavirus

A rabies-related lyssavirus, which can be fatal to humans, has recently been found in several species of Australian bat. It is not known how long the virus has been in Australia, or how widespread it is. There is a remote chance that this virus is present in New Zealand bats.

What is lyssavirus?

The term ‘lyssavirus’ comprises a group of viruses which fall into seven related gene groups, one of which is common rabies. Several of the lyssaviruses give rise to clinical disease in bats and many have been shown to affect humans and/or domestic animals. Scientists at the Australian Animal Health Laboratory (AAHL) have found that the cases in Australia belong to a new lyssavirus gene group—gene group seven. Although this particular bat lyssavirus has only been isolated in

Australia, it is possible that it will be present in other countries in the region where bat species are found, including New Zealand. This is a relatively new virus, so the information below is based on the close relative of the common rabies.

Symptoms

Similar to rabies (see 'Rabies'—olddm-544362): illness with numbness and weakness in limbs, progressing to coma and death from encephalitis.

Contracting lyssavirus

Lyssavirus could be contracted from a bite from an infected bat.

Treatment

Immunity to rabies when administered after an exposure (post-exposure prophylaxis) or for protection before an exposure occurs (pre-exposure prophylaxis). However, the effectiveness of this inoculation against the different strains of lyssavirus is uncertain. Once the disease has developed there is no cure.

The best immediate treatment after potential exposure is to wash the wound thoroughly with soap and water, and seek medical attention immediately. Avoidance of handling affected or sick bats and wearing protection such as gloves are the best precautions for avoiding risk of exposure.

Recommendations from the Bat Recovery Group

New Zealand bat workers should take precautions when handling bats, as is currently the case in Europe, America and now Australia:

- All bats which appear to be sick should be handled with gloves. If gloves are not available then bats should not be handled.
- Bodies of sick or dead bats should be necropsied, and results put on the national database, as per the DOC 'Wildlife health SOP' (olddm-766252).
- Numbers of workers handling bats on particular projects should be minimised.
- Workers handling short-tailed bats (which bite often) should wear gloves whenever possible, until the risk is more fully understood.
- Workers handling short-tailed bats without gloves could take a precautionary approach and obtain rabies vaccinations.
- Any bat bites should be washed thoroughly with soap and water, and medical attention should be sought immediately if there is concern about the seriousness of the wound.

Further information:

- DOC 'Infectious diseases information system' (docdm-383258).
- Information on the close relative of the common rabies (see 'Rabies'—olddm-544362).

Other web-based fact sheets:

- Bat Conservation International: http://www.batcon.org/

Disease risk to bats

There has been limited work on diseases in New Zealand bats. This has been summarised by Duignan et al. (2003) who did not identify any significant disease issues to date. However, absence of records to date does not mean that there is no risk of transmitting diseases among populations of bats. Therefore, a precautionary approach should be taken. DOC's requirements relating to wildlife health are laid out in the 'Wildlife health SOP' (olddm-766252; 'Wildlife health SOP index page'—olddm-757175).

Additional related documents

- Safe handling of pesticides (docdm-22730)
- Helicopter safety—technical document (docdm-208219)
- All terrain vehicles and all terrain vehicle utility—technical document (docdm-425085)

References

Armstrong, K.N.; Higgs, P. 2002: Draft protocol for working safely in confined spaces. *Australasian Bat Society Newsletter 19*: 20–28.

Duignan, P.; Horner, G.; O'Keefe, J. 2003: Infectious and emerging diseases of bats, and health status of bats in New Zealand. *Surveillance 30(1)*: 12–15.

Biosecurity incursions

Biosecurity is the exclusion, eradication or effective management of risks posed by pests and diseases to the economy, environment and human health.

Accidental importations of bats

Six exotic species of bats from three Microchiropteran families have arrived dead in New Zealand as stowaways in cargo. Three were vespertilionids: a Japanese pipistrelle *(Pipistrellus javanicus abramus)* arrived in a cargo of car parts (Daniel & Yoshiyuki 1982), an Australian lesser long-eared bat *(Nyctophilus geoffroyi)* in a cargo of timber (Daniel & Williams 1984), and an Australian little forest bat (*Vespadelus vulturnus*) in a crate of aircraft parts (O'Donnell 1998). A small unidentified bat belonging to the Molossidae arrived amongst a shipment of bananas from Ecuador in 2002 and was found in a Queenstown shop, and a dog-faced fruit bat, *Cynopterus brachyotis* (Family Pteropidae), a relatively common bat in Malaysia and one of the smallest of the flying foxes was found in a shipment of cement arriving in Dunedin in October 2004. Another Molossid, *Tadarida plicata*, was found dead in a vehicle at Papakura after being imported from Thailand in June 2005 (C. O'Donnell, pers. comm).

Risks

Exotic bats may pose potential threats to New Zealand fauna and humans, particularly through disease risk. For example, Pteropidae are known to carry the rabies-related lyssavirus, which can be fatal to humans and other bats. The species of *Cynopterus* found recently raised antibodies to Nipah virus (family Paramyxoviridae) in a recent overseas study. Nipah virus caused disease in pigs and humans in peninsular Malaysia in 1998–99.[12]

So far, all interceptions have been of dead bats. There is only a slight risk of disease transmission to New Zealand from cases like this. In addition, most bats have been tropical species that are unlikely to become established here given the current climate. However, all interceptions demonstrate the need for vigilance at the border.

Pre-border interception

Where a bat has been identified before biosecurity clearance or while it is still under the jurisdiction of the Ministry for Primary Industries (MPI), the incursion is dealt with by MPI (see box below). This is because it is not an incursion *per se*, i.e. it has not been released into the environment. DOC is alerted in due course in these situations. In some situations DOC staff have been asked to assist with identification of bats.

Post-border interception

The interception of exotic bats within New Zealand becomes an issue for DOC because of the risk of becoming established in the wild.

[12] see http://www.cdc.gov/ncidod/eid/vol7no3/yob.htm

For advice on procedures to follow in this situation see ‘Reporting procedure for suspected new organisms’ (docdm-290718)

If you think you’ve seen a potential new bat species or disease or pest symptoms:

Call the Ministry for Primary Industries Hotline

0800 80 99 66

http://www.mpi.govt.nz/biosecurity-animal-welfare/pests-diseases

Note: This phone number is not the equivalent of a 111 emergency number. It is for the public (including DOC staff) to report anything unusual—either sign (such as disease symptoms that you may not have seen before), or an organism that might be new to New Zealand. Most people are unaware that under the Biosecurity Act, every person has a legal obligation to report any suspected new organism as soon as practicable.

MPI has access to experts at diagnosing new species—often they can even do this over the phone with the information that you’ve given them.

References

Daniel, M.J.; Williams, G.R. 1984: A survey of the distribution, seasonal activity and roost sites of New Zealand bats. *New Zealand Journal of Ecology 7*: 9–25.

Daniel, M.J.; Yoshiyuki, M. 1982: Accidental importation of a Japanese bat into New Zealand. *New Zealand Journal of Zoology 9*: 461–462.

O’Donnell, C.F.J. 1998: Accidental importation of an Australian bat (Mammalia: Chiroptera: *Vespadelus vulturnus* into New Zealand. *New Zealand Journal of Zoology 25*: 455–456.

Appendix A

The following Department of Conservation documents are referred to in this method:

olddm-759839	ABM instructions
docdm-737250	AEC application
docdm-425085	All terrain vehicles and all terrain vehicle utility—technical document
docdm-51708	Animal pests SOP definitions and FAQs
docdm-232655	Application flow chart
olddm-723120	Bat Recovery Group recommendations 2001–2004
olddm-715142	Bat Recovery Group recommendations 2004/2005
docdm-590733	Bats: counting away from roosts—automatic bat detectors
docdm-590701	Bats: counting away from roosts—bat detectors on line transects
docdm-130631	Blank field recording sheet for transponder recapture
docdm-266180	Captive management SOP
olddm-766783	Code of ethical conduct for the manipulation of live animals
olddm-718668	Collection, storage and transport of diagnostic samples from birds and reptiles
docdm-22907	Extracting a long-tailed bat from a mist net
docdm-208219	Helicopter safety—technical document
docdm-383258	Infectious diseases information system
docdm-379889	Instructions for setting up RFID readers, dataloggers and antennae
docdm-590958	Introduction to bat monitoring
olddm-921158	Massey University IVABS pathology sample submission instructions
docdm-95676	Preparing an Assessment of Environmental Effects
olddm-32604	Psittacine pox internal prevention and emergency response plan for DOC
olddm-544362	Rabies
docdm-290718	Reporting procedure for suspected new organisms
docdm-611915	Risk Manager user manual
docdm-159363	Roped tree work SOP
docdm-22730	Safe handling of pesticides
olddm-574301	Sequence of lesser short-tailed bat calls
olddm-574297	Sequence of long-tailed bat calls
olddm-757423	Suspected incursion response procedure
docdm-929116	Tissue sampling for bats SOP
docdm-1089378	Translocation SOP
docdm-130625	Transponder field recording sheet for new tags
docdm-97398	Use of second generation anticoagulants on public conservation lands
docdm-131439	Video clip of a lesser short-tailed bat being tagged
olddm-766252	Wildlife health SOP
olddm-757175	Wildlife health SOP index page
olddm-677719	Wildlife submission form