Best practice manual of conservation techniques for pekapeka/bats in Aotearoa New Zealand

2025

This manual was originally prepared by Jane Sedgeley, Colin O'Donnell, John Lyall, Hannah Edmonds, Warren Simpson, Jo Carpenter, Jo Hoare and Kate McInnes in 2012.

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Contents

1. Introduction

Three species of native pekapeka/bats representing five different taxonomic units are known to occur in Aotearoa New Zealand, all of which are threatened (Molloy 1995; O'Donnell et al. 2010, 2018, 2023). Considerable research effort and active conservation programmes are now 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, and relatively few people currently 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 outlines appropriate research, inventory and monitoring, and management techniques, and provides ethical standards that should be applied when working with bats. This introduction describes the main objectives of the best practice manual, how the manual was developed, accountabilities for maintaining best practices and review procedures.

1.1 Objectives

The objectives of this manual are to:

- provide information and resources on the best techniques currently available to manage and undertake research on New Zealand bats
- assist Department of Conservation Te Papa Atawhai (DOC) staff, external managers and researchers to develop and improve techniques used for the recovery and management of New Zealand bats
- provide guidelines for safe, ethical and responsible practices when handling and studying New Zealand bats
- help formalise the consistent use of best management practices across bat taxa throughout New Zealand
- provide a mechanism for advocating the continuous improvement of bat conservation management and research.

1.2 Development of best practices

The practices described in this manual represent a mixture of techniques that are used globally and techniques that have been adapted or developed for working specifically with New Zealand bats. Most of these techniques have been trialled and used in New Zealand over the last 30 years with approval from appropriate Animal Ethics Committees (AECs). Those techniques that are yet to be used in New Zealand are clearly identified in the text.

1.3 Accountabilities for identifying and maintaining best practices

The following accountabilities are assigned to DOC staff in the context of a best practice manual. However, people outside DOC who can contribute to improving best practices 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 New Zealand bats.

- Operations Managers
- Rangers
- Senior Rangers
- Bat Recovery Group members
- Technical Advisors
- Science Advisors

When a Wildlife Act Authority is provided by the Department, there will be a clause that the Best Practice Manual must be followed. The authority holder then becomes accountable for following this manual.

1.4 Definitions of best practice procedures

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**' or '**do not**'.

Recommended procedures are those that, based on our current knowledge, are considered to be the best methods or procedures but for which there may be reasonable alternatives. These procedures are indicated by the word '**should**'.

1.5 Review process

The Bat Recovery Group is charged with the responsibility of reviewing and revising this manual to ensure that current best practices are promulgated within and outside DOC.

In some sections, alternative approaches may be suggested, but further experience may reveal that one method is superior to the alternatives, resulting in this becoming the single mandatory best practice.

There are also various methods under development that have not yet been written or agreed upon as best practice. Once 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 carefully documented. Adherence to current best practice guidelines should not prevent innovations that may result in improved performance, but it is important that such innovations are monitored and tested fully.

1.6 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 under 'Policy and procedure', some of which can also be found on the DOC website.[1] It is not intended to repeat these guidelines in this best practice manual, as each of these documents is regularly revised and updated and new procedures are regularly added to the sites. Examples include the following documents (see Appendix 1):

- *Wildlife Health Management SOP*
- *Operational Planning for Animal Pest Operations SOP*
- *Animal Pests SOP Definitions and FAQs*
- *Safe Handling of Pesticides SOP*
- *Use of Second-generation Anticoagulants Policy*

[1] www.doc.govt.nz/about-us/our-policies-and-plans/our-procedures-and-sops/managing-animal-pests/standard-operating-procedures/

2. Introduction to New Zealand bats

Note that much of the information in this section came from Lloyd (2001, 2005) and O'Donnell (2001a, 2005).

2.1 Conservation status

Three bat species are known to occur in New Zealand, all of which are endemic: the long-tailed bat (*Chalinolobus tuberculatus*), the lesser short-tailed bat (*Mystacina tuberculata*) and the greater short-tailed bat (*M. robusta*). There have also been cases of vagrants arriving here, although these have mainly been accidental imports with freight (see Biosecurity incursions). The long-tailed bat is a member of the large and widespread family Vespertilionidae, which is represented by seven species in Australasia (Hutson et al. 2001). The long-tailed bat is currently described as a single species, but significant differences in size and genetic diversity have been recorded throughout its geographical range (Winnington 1999; O'Donnell 2001a; Dool et al. 2016). The lesser short-tailed bat and greater short-tailed bat belong to the endemic and monogeneric family Mystacinidae. The lesser short-tailed bat is currently described as three subspecies (Table 1), but significant genetic diversity has been recorded throughout its geographical range. The greater short-tailed bat is generally considered to be extinct because there have been no confirmed sightings since 1967 (Daniel 1990); however, unusual mystacinid-like calls were recorded on Putauhina Island in 1999, leading to speculation that greater short-tailed bats might still be extant (Notes).

New Zealand bats are protected under the Wildlife Act 1953. The long-tailed bat is listed as Critically Endangered, the lesser short-tailed bat is listed as Vulnerable and the greater short-tailed bat is listed as Critically Endangered, Possibly Extinct under the International Union for Conservation of Nature (IUCN) Red List system (Hutson et al. 2001; O'Donnell 2008, 2021a, 2021b). The New Zealand Threat Classification System was developed to complement the Red List system to reflect New Zealand's unique environments and to account for the country's relatively small size and diversity of ecosystems, as well as the large number of taxa with naturally restricted ranges and/or small population sizes (Molloy et al. 2002). Under this system, DOC currently identifies five endemic bat taxa of concern and one vagrant species (O'Donnell et al. 2018, 2023; Table 1). Only the central lesser short-tailed bat (*M. t. rhyacobia*) can be considered secure on the mainland, with the remaining taxa potentially facing a high risk of extinction on the mainland in the medium term if conservation management is not successful in reversing their declines (Molloy 1995; O'Donnell et al. 2010, 2023). The two single populations of lesser short-tailed bats on Whenua Hou / Codfish Island and Te Hauturu-o-Toi / Little Barrier Island are considered more secure.

Table 1 Conservation status of New Zealand bats based on the New Zealand Threat Classification System (Townsend et al. 2008; O'Donnell et al. 2023)

SCIENTIFIC NAME	COMMON NAME	CONSERVATION STATUS
Chalinolobus tuberculatus	long-tailed bat	Nationally Critical
Mystacina robusta	greater short-tailed bat	Data Deficient
Mystacina tuberculata aupourica	northern lesser short-tailed bat	Nationally Vulnerable
Mystacina tuberculata rhyacobia	central lesser short-tailed bat	Declining

Mystacina tuberculata tuberculata	southern lesser short-tailed bat	Nationally Increasing
Pteropus scapulatus	little red flying fox	Vagrant

2.2 Distributions and populations

2.2.1 Long-tailed bat

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

The size is known for only a few long-tailed bat populations. Banding studies in some of the larger populations suggest that a minimum of 800 bats are present in Grand Canyon Cave near Te Kūiti (O'Donnell 2002b), 150–200 bats are found at Hanging Rock in South Canterbury (Lettink and Armstrong 2003) and > 300 occur in the Eglinton valley in Fiordland (O'Donnell 2000b). Averages of 86 bats have been recorded emerging from roosts in Pukeiti in Hawke's Bay (range = 5–208; Gillingham 1996) and 14 bats have been recorded in the Waitākere Ranges (range = 2–24; Alexander 2001), but these are likely to be underestimates of the total population sizes. Group sizes in plantation forests and urban settings are generally very small (< 10 bats; Dekrout 20.09; Borkin and Parsons 2010).

2.2.2 Lesser short-tailed bat

Lesser short-tailed bats were also once widespread across New Zealand, with fossils having been found in Waikato, Hawke's Bay, the Wairarapa, northwest Nelson, Canterbury and Fiordland. However, there are no current records from much of Northland, the Coromandel, Rotorua, Pirongia, East Cape, western Tararua Forest Park, Nelson Lakes National Park, Mount Richmond, Ōkārito, the Longwood Range and Catlins Forest Park, despite their former presence in all of these areas (Dwyer 1962; Daniel and Williams 1984; Daniel 1990). The species may persist in some of these areas as survey efforts are incomplete.

Recent surveys have shown that lesser short-tailed populations persist in several areas in the North Island and at least three areas in the South Island (Lloyd 2005b). In the North Island, a small population remains in Ōmahuta/Puketī forest in Northland, and lesser short-tailed bats have also been found from north Taranaki across the central volcanic plateau towards East Cape, with significant populations occurring in Waitaanga, Pureora, Rangataua, Kaimanawa, Whirinaki and southeast Urewera forests. These bats also occur in Waioeka Gorge and may still occur in Waitōtara, Kaimai-Mamaku and Ruahine Forest Park. However, recent surveys suggest that the population in the eastern Tararua Range may be extinct.

Only two populations of lesser short-tailed bats have been confirmed in the South Island. These are located in the Murchison Mountains (Fiordland National Park) and the Eglinton valley (Fiordland National Park). A population at Ōpārara has not been recorded since 2001, despite reasonably extensive surveying. Calls suggestive of short-tailed bats have also been recorded in Paparoa National Park and the Dart valley, but their identity has yet to be confirmed. No lesser short-tailed bats have been recorded during preliminary surveys of forests elsewhere on the West Coast, across the top of the South Island, and in parts of Fiordland National Park and northwest Stewart Island/Rakiura, but there are large populations on Te Hauturu-o-Toi / Little Barrier Island and Codfish Island / Whenua Hou (Lloyd 2005b, 2009, 2010, 2011, 2012).

The size is known for only a few lesser short-tailed bat populations. Numbers of bats in Rangataua Forest fluctuated between 5740 and 6977 in early summer from 1995 to 1999, and there have been minimum population estimates of 2700 bats in Waitaanga (B Williams, DOC, pers. comm.) and 1557 bats on Whenua Hou / Codfish Island / (Sedgeley and Anderson 2000), although the latter estimate is probably substantially lower than the actual population size because other roosts were known to be occupied in the study areas that were not counted. Based on these data and unpublished observations, Lloyd (2001, 2005) estimated a total lesser short-tailed bat population size of < 40,000 individuals in the central North Island and < 50,000 individuals across New Zealand.

More recent estimates of minimum population sizes based on roost emergence counts of lesser short-tailed bats include > 2900 adults and juveniles in the Eglinton valley based on counts from a single roost in January 2018 (Thakur et al. 2018); > 5000 adults in Whirinaki based on counts from a single roost in December 2016 (Wills 2018); and > 8000 adults and juveniles in Rangataua based on counts from four roosts on the same night in February 2019 (Beath and Hayward 2019; Notes). At Pureora, the adult (pre-breeding) population size in November 2013 was estimated to be c. 800 bats using mark-recapture and closed population modelling (Dennis 2019). There are not any recent estimates of the whole population.

2.2.3 Greater short-tailed bat

Two subspecies of short-tailed bats were initially described (Dwyer 1962): *Mystacina tuberculata tuberculata*, which has a smaller body size and is found throughout much of New Zealand, and *M. t. robusta*, which has a larger body size and is restricted to the Tītī / Muttonbird Islands, off the southwest coast of Stewart Island/Rakiura. Subsequently, these subspecies were elevated to species status as the lesser short-tailed bat *M. tuberculata* and the greater short-tailed bat *M. robusta* (Hill and 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 once inhabited sites in Waitomo, Hawke's Bay and the Wairarapa in the North Island, and in northwest Nelson, Westland, Canterbury and Central Otago in the South Island. From 1840 until the early 1960s, greater short-tailed bats were only found on rat-free Taukihepa / Big South Cape Island (930 ha) and Rerewhakaupoko / Solomon Island (32 ha) in the Tītī / Muttonbird Islands, 2–10 km off the southwest coast of Stewart Island/Rakiura.

2.3 Basic ecology

2.3.1 Long-tailed bats

Morphology and foraging behaviour

Adult long-tailed bats are relatively small, with a body mass of 8.5–12.3 g and a forearm length of 38.7–40.5 mm (O'Donnell 2001a) (Fig. 2). Their wing characteristics and echolocation calls are typical of moderately fast-flying bats that forage along forest edges and gaps (O'Donnell 2000e, 2001a).

Long-tailed bats are strictly insectivorous. Although a comprehensive study of their diet has yet to be conducted, existing data suggest that they consume a wide variety of aerial aquatic and terrestrial invertebrates (Gillingham 1996; O'Donnell 2005). Gurau (2014) identified Diptera, Lepidoptera and Coleoptera as their primary food sources in both exotic pine plantation and native podocarp-broadleaf forests in the Central North Island.

Breeding and social system

There is little information on the mating system of long-tailed bats. Mating most likely occurs in autumn, but the length of embryonic development and the mechanism for delaying the onset of gestation are unknown (Dekrout 2009). The time of birth varies geographically but occurs sometime between mid-November and mid-December when females congregate in maternity roosts to give birth and raise their young. Each female gives birth to a single pup once per year (O'Donnell 2001a, 2002a).

Long-tailed bats have a complex social system. For instance, the part of the population that is being studied in the Eglinton valley is split into three behaviourally, though not geographically, isolated sub-groups that rarely mix. Bats belonging to each sub-group are spread over many roosts each day, and the composition of bats within these roosts changes 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, farmland and adjacent to urban areas (Daniel and Williams 1984; Dekrout 2009; Borkin and Parsons 2010). These mammals have very large home ranges, with colonies in the Eglinton valley ranging over 11,700 ha. Individual ranges vary greatly depending on the age, sex and time of the breeding season, averaging 237–2006 ha and reaching a maximum of 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 in a range of indigenous and exotic tree species (Daniel 1981; Daniel and Williams 1981, 1983, 1984; Sedgeley and O'Donnell 2004; Dekrout 2009). However, radio-tracking studies have shown that the majority of their roosts are in trees and that maternity roosts are almost exclusively in trees (Gillingham 1996; Griffiths 1996; Sedgeley and O'Donnell 1999a, 1999b, 2004).

Roosting behaviour

Detailed studies of roosting behaviour in Fiordland and South Canterbury have shown that breeding groups of long-tailed bats select specific roost trees and roost cavities that are very distinct from the pool of trees potentially available to them. Bats here were observed roosting inside cavities in the main trunks or limbs of some of the largest and oldest trees available, and these cavities were high off the ground (usually > 15 m), generally had one entrance, had small to medium sized internal dimensions (compared with those of 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 walls, and had a stable internal humidity and temperature compared with ambient conditions (Sedgeley and O'Donnell 1999a, 1999b; Sedgeley 2001a).

In the Eglinton valley, an average of 14–86 bats have been observed using a roost at any one time, and individuals change roost sites almost every day. Additionally, the bats here seldom use an individual roost more than once during a given summer but will reuse many of these roosts from year to year (O'Donnell and Sedgeley 1999). By contrast, individual roosts are used much more frequently in the modified landscape of south Hamilton where roost sites are very limited (Dekrout 2009).

Fig. 2. Long-tailed bat (*Chalinolobus tuberculatus*) in the hand. *Photo: CFJ O'Donnell*

2.3.2 Lesser short-tailed bats

Morphology and foraging behaviour

Adult lesser short-tailed bats are slightly larger than long-tailed bats, with a body mass of 11.4–22.0 g and a forearm length of 36.9–46.9 mm (O'Donnell et al. 1999; Lloyd 2005b) (Fig. 3). These 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).

The wing shapes of lesser short-tailed bats vary. Data collected in the Eglinton valley demonstrated that they have wings that make them more manoeuvrable than long-tailed bats within vegetation, which is typical of species that are known to forage by gleaning insects (Jones et al. 2003). However, lesser short-tailed bats on Codfish Island / Whenua Hou do not seem to have any particular specialised flight strategy (Webb et al. 1999).

The echolocation calls of lesser short-tailed bats are typical of bats that glean from surfaces, and 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, with a diet consisting of flying and non-flying invertebrates, nectar, pollen, plant material, and fruit (Daniel 1976; Arkins et al 1999; Cummings et al. 2014). Adaptations for nectar feeding include an extensible, papillated tongue and a wide gap between the incisors (reviewed in Arkins et al. 1999).

Fig. 3. Lesser short-tailed bat (*Mystacina tuberculata*) in the hand. *Photo: CFJ O'Donnell*

Breeding

Lesser short-tailed bats have a lek mating system, where males occupy singing or mating roosts that are clustered around communal roosts and call from them at night to attract females. Some singing roosts are shared between multiple males (Toth et al. 2015). 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 unmodified forest (Daniel and Williams 1984). There are no contemporary records of these bats roosting in caves, but there are historical records that show caves were once used, with large concentrations of bat remains having been found in limestone caves (Worthy and Holdaway 1994, 2001) and both lesser and greater short-tailed bats having been recorded roosting in granite sea caves on islands to the southwest of Stewart Island/Rakiura (Stead 1936; Daniel and Williams 1984). There are also several records of lesser short-tailed bats in buildings, but these were all from mountain huts or homes adjacent to large areas of indigenous forest (Daniel and Williams 1984).

Lesser short-tailed bats have very large home ranges, with colonies in the Eglinton valley ranging over 14,710 ha (O'Donnell 2001), and large populations have only been found in extensive (1000-ha) areas of largely unmodified old-growth forest (Lloyd 2001). Individual ranges vary greatly depending on age, sex and time of the breeding season, ranging from 127 to 6223 ha (Christie 2003b).

Roosting

Three types of roost trees have been described for lesser short-tailed bats: 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 that are used by solitary bats and singing/mating bats have much smaller stem diameters and internal cavity dimensions than those used by large groups of bats (O'Donnell et al. 1999; Sedgeley 2003, 2006). The locations of roost cavities that are used by communally roosting lesser short-tailed bats are variable, with roost entrances typically being found on the main trunk and less than 0.3–7 m above the ground in Rangataua Forest and on Codfish Island / Whenua Hou, compared with up to 23 m above ground in the Eglington valley (Sedgeley 2003; Lloyd 2005b). The size of the entrances also varies greatly, from small holes that are only just large enough for bats to enter through to enormous splits several metres long (Sedgeley 2003, 2006; Lloyd 2005b). All of the roosts selected by lesser short-tailed bats 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 two or three communal roosts at any one time (Lloyd 2001; Christie 2003b).

Several features distinguish lesser short-tailed bat roosts from long-tailed bat roosts in the Eglinton valley, with lesser short-tailed bat roosts generally being further inside the forest, often having multiple entrances to the roost cavities, being lower to the ground, and having much larger entrance and internal dimensions than long-tailed bat roosts. Lesser short-tailed bat roosts are also used by far greater numbers of bats (several hundreds), could be occupied for up to several weeks at a time and are re-used on a more regular basis than long-tailed bat roosts (Sedgeley 2003).

2.3.3 Greater short-tailed bats

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

2.4 Major threats to New Zealand bats

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

2.4.1 Predation

Introduced mustelids, rats (*Rattus* spp.), possums (*Trichosurus vulpecula*) and cats (*Felis catus*) all prey on, or attempt to prey on, New Zealand bats. Bats are vulnerable to these predators throughout the year, both 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 and Placido 1975; Hill and Smith 1984), and stoats (*Mustela erminea*) have been seen entering lesser short-tailed bat roosts and preying on these bats in New Zealand (S Wills, pers. obs.), although it remains unclear whether they affect population viability. Rats have a large impact on bat populations, with disappearance of greater short-tailed bats from mainland New Zealand coinciding with the spread of kiore (*R. exulans*) (Worthy 1997), and this species of bat subsequently becoming extinct during the 1960s when ship rats (*R. rattus*) arrived on Taukihepa / Big South Cape Island off Stewart Island/Rakiura, which was its last known refuge (Daniel 1990). Lloyd (2001) also recorded ship rats attempting to enter lesser short-tailed bat roosts during winter, although none were successful at capturing the bats, and 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).

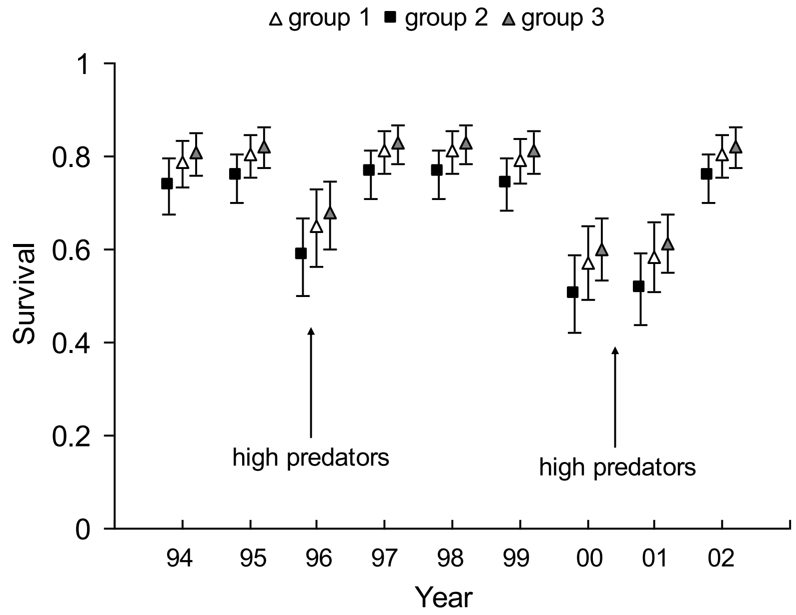


Fig. 1. Annual overwinter survival (mean ± SE) of adult female long-tailed bats (*Chalinolobus tuberculatus*) in three social groups in the Eglinton valley from 1993 to 2003. Survival was lower in years when there were high numbers of introduced predators (1996, 2000, 2001). Taken from Pryde et al. (2005).

Feral cats also appear to be common predators of bats (Dwyer 1962; Daniel and Williams 1984; O'Donnell 2000a), accounting for 28% of reported long-tailed bat deaths and 26% of lesser short-tailed bat deaths (Daniel and Williams 1984), including juveniles (Daniel and Williams 1981). In Rangataua Forest, at least 102 lesser short-tailed bats were killed by a wild cat at two colonial roost trees over a 7-day period in 2010 (Scrimgeour et al. 2012). Similarly, Dowling et al. (1994) reported that cats accounted for 45% of injured *Chalinolobus* bats handed in for care in Victoria, Australia. Additionally, possums have been recorded attempting to reach into cavities containing young long-tailed bats in South Canterbury on 50% of nights when roosts were monitored using video cameras (O'Donnell 2000d).

Ruru/morepork (*Ninox novaeseelandiae*) are native predators of bats (Stead 1936; Dwyer 1960; Daniel and Williams 1984), and Griffiths (1996) also observed introduced ruru nohinohi / little owls (*Athene noctua*) attempting (unsuccessfully) to catch long-tailed bats near Geraldine in South Canterbury. Little owls have also been reported taking bats in Britain (Speakman 1991).

In the Eglinton valley, Fiordland, introduced starlings (*Sturnus vulugaris*) and ship rats have been reported making nests in long-tailed bat roosts (Sedgeley and O'Donnell 1999b), although it is not known if these animals actually preyed upon or displaced bats from these roosting sites.

2.4.2 Competition

Competition for roosting sites by exotic species is also an issue for New Zealand bats as it may limit the availability of roosts. Griffiths (1996) found that starlings, house sparrows (*Passer domesticus*), feral pigeons (*Columba livia*) 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 also been suggested in Europe (Mason et al. 1972; Rieger 1996).

There has also been speculation that both exotic and native species may compete with lesser short-tailed bats for food sources, as many of the types of fruits eaten by lesser short-tailed bats are also eaten by many bird species, rodents and possums (Daniel 1976). Ecroyd (1995) demonstrated using infrared video surveillance that ship rats and possums feed on significant amounts of nectar from the flowers of pua o Te Rēinga / wood rose (*Dactylanthus taylorii*), making this resource unavailable to lesser short-tailed bats, and Molloy (1995) also suggested that introduced wasps, which feed on nectar, fruits and insects, may compete with lesser short-tailed bats. Possums cause serious damage to native ecosystems through their selective browsing on the buds, flowers and fruits of a wide variety of native trees and shrubs, and have also caused mortality of native trees (Cowan and Waddington 1990), leading to the modification of bat habitat and possibly competition with lesser short-tailed bats for some foods.

2.4.3 Habitat degradation

Before humans arrived in New Zealand, indigenous forest covered 85–90% of the country (McGlone 1989). However, this forest has now been reduced to c. 14% of its original area (Stevens et al. 1988). Dwyer (1960, 1962) concluded that the decrease in the distributional area of bat populations was correlated with the removal of indigenous forest during the last century and the failure of either bat species to survive well in open country or urban areas. The 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 and Drummond 1904; Dwyer 1960; Barrie 1995), with early records noting that the burning and felling of trees for timber and the clearance of land for agriculture or mining by European colonists disturbed large colonies of long-tailed bats (Buller 1892; Cheeseman 1893). Similarly, the majority of bat deaths recorded by Daniel and Williams (1984) occurred when the bats' roost trees were cut down, and there are still instances of bat roosts being felled for firewood and timber in Nelson, Buller and Canterbury in the South Island and in the King Country in the North Island (O'Donnell 2000a, 2000d), as well as during the removal of old trees near housing in Waikato (K Borkin, pers. obs.).

Today, habitat degradation usually relates to the loss of old-aged preferred roost trees in areas where bat colonies are found or a decline in the quality of important foraging habitat (e.g. Sedgeley and O'Donnell 1999a, 1999b, 2004; Sedgeley 2003). Major threats include the 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.

Habitat can also be degraded by anthropogenic effects, including increasing traffic volumes, lighting and noise. Trials have shown that adding light to short-tailed bat habitat reduces their activity (K Borkin, pers. obs.), while studies in the Hamilton area have indicated that long-tailed bat activity is highest in areas where roads, housing, streetlight density and light itself are lowest (Le Roux and Le Roux 2012; Dekrout et al. 2014; Borkin and Smith 2019; Notes). The activity of long-tailed bats also appears to decline when overnight traffic volumes increase (Borkin et al. 2019), and a playback experiment found that there was less long-tailed bat activity when aircraft noise was played back in areas where bats were not usually exposed to this noise (Le Roux and Waas 2012).

Long-tailed bats have been recorded in commercial plantation forests, but roost trees in these areas are regularly felled as part of normal logging operations (Borkin et al. 2011). Additionally, habitat that has recently been subjected to extensive logging has been converted to pasture (Borkin and Parsons 2010). Primary feeding habitats that occur along waterways can also be affected by a variety of river control works, including water abstraction from foraging sites, the construction of dams that may drown foraging sites and changes in water quality that lead to reductions in invertebrate prey numbers (O'Donnell 2000a, 2000d).

2.4.4 Disturbance

Disturbance by rock climbers, particularly in winter when bats are using the same limestone crevices as climbers use, was identified as a potential risk to long-tailed bats at Hanging Rock escarpment in South Canterbury (Griffiths 1996). Disturbance of cave-roosting bats also remains a concern at Grand Canyon Cave (D Smith, DOC, pers. comm.), with recorded instances of all bats in this cave taking flight while people have been watching them (N Miller, DOC, pers. comm.). Bats have also been observed to vacate tree roosts and artificial bat roosts during the day as a result of human disturbance (CFJ O'Donnell and K Borkin, DOC, pers. obs.).

3. Conservation management techniques for New Zealand bats

The purpose of this section is to identify conservation management techniques that aim to sustain or improve the status of bat populations in New Zealand.

3.1 Desired outcome of bat management projects

The ultimate outcome of any bat management project should be to maintain the long-term security of the bat populations in the area where work is being proposed and/or to restore environments within an area where bat numbers have declined. Seven general management techniques are available.

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

Aspects of each of these can be customised for local bat conservation projects. The Bat Recovery Group can advise on the type and level of management required to recover bat populations. Additionally, inventory and monitoring programmes can measure where and when outcomes are being achieved.

3.2 Recovery potential

Bats are very long-lived and attempt to breed each year, so bat populations have good recovery potential if threats are removed. Potential habitats where populations could be restored are extensive, and there is a strong interest in the conservation of bats across New Zealand, indicating a strong potential for developing cooperative conservation projects. However, because female bats only give birth to a single pup once per year, recovery will be slow and difficult to detect in the short term.

3.3 Management techniques

Management techniques for general restoration work include the following components.

3.3.1 Establishing the presence and significance of bats in an area and determining at what point to initiate management

Inventory aimed at establishing the presence or relative numbers of bats in a particular area is an important task in those areas where there is uncertainty about the status of the bat populations.

This may result in the discovery of significant populations that were previously unrecognised and provide additional knowledge about the status of populations to inform management.

3.3.2 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 that 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 negatively affect bats is also 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 proposals for:

- water abstraction that would reduce the availability of aquatic foraging habitat
- building or modifying structures (e.g. roads, canals) where this might affect roosting or foraging habitats
- removing trees that may provide roosting habitat from roadsides, reserves, campgrounds, tracks, etc.
- logging forest on private land
- poisoning operations that would use new, non-approved baits and lures that would potentially attract bats
- gravel extraction on riverbeds where significant bat foraging habitat occurs
- damming rivers in areas where significant bat foraging habitat occurs.

Another important area of advocacy relates to making submissions on sustainable forestry management plans on private land under the requirements of the Forests Amendment Act 1993. Logging generally targets a significant proportion of trees that are preferred roosting sites of bats, so there is a high risk that localised tree selection would wipe out a population unless roosting patches are identified and protected. Bat detectors can be used to determine the presence of bats, and any management systems proposed for use in the future need to leave sufficient trees to ensure that bat populations survive.

DOC employees with responsibilities for bat conservation should use data on the types and sizes of bat roosts to argue for rules to prevent the impacts of logging and forest clearance on bats through their statutory planning procedures and input into management plans. Management plans need to demonstrate that safeguards are in place to prevent bat populations from being threatened.

The roost tree preferences of New Zealand bats can be found in Sedgeley and O'Donnell (1999a, 1999b, 2004) and Sedgeley (2003) which is summarised below.

Roost tree preferences of long-tailed bats

Long-tailed bats roost in trees with a large diameter at breast height (DBH). Roost trees are mostly greater than 80 cm DBH and can reach up to 250 cm DBH (Fig. 4) and are usually 200–650 years old. Long-tailed bats roost in small- to medium-sized cavities that are usually high up on trees (15–20 m from the ground). They move to a new roost tree virtually every day, and one group can use over 100 different roosting trees. In Fiordland, 74% of long-tailed bats roost in red beech (*Fuscospora fusca*) trees, 21% in standing dead trees, 4% in silver beech (*Lophozonia menziesii*) trees and 1% in mountain beech (*F. cliffortioides*) trees, while in Northland, they roost in the large kauri (*Agathis australis*) trees. In podocarp-hardwood forests, long-tailed bat roosts have been found in rimu (*Dacrydium cupressinum*), miro (*Pectinopitys ferruginea*), kahikatea (*Dacrycarpus dacrydioides*), mataī (*Prumnopitys taxifolia*) and tōtara (*Podocarpus totara*) trees ranging from 50 to 180 cm DHB.

Roost tree preferences of lesser short-tailed bats

Lesser short-tailed bats tend to use larger roost cavities than long-tailed bats. In mixed beech forest, they roost in splits and hollows mainly in large red beech trees that are 40–160 cm DBH (Fig. 5), while in podocarp-hardwood forest, they are dependent on large-diameter Hall's tōtara (*Podocarpus laetus*), rimu, southern rātā (*Metrosideros umbellata*), mataī and miro trees. Most roosts occur in trees greater than 80 cm DBH, but some are also found in smaller trees, most of which are used by solitary bats, groups that require smaller cavities for hibernation or for mating/singing. Lesser short-tailed bats may use a particular roost for just 1 day or continuously for up to 6 weeks before moving to another tree roost.

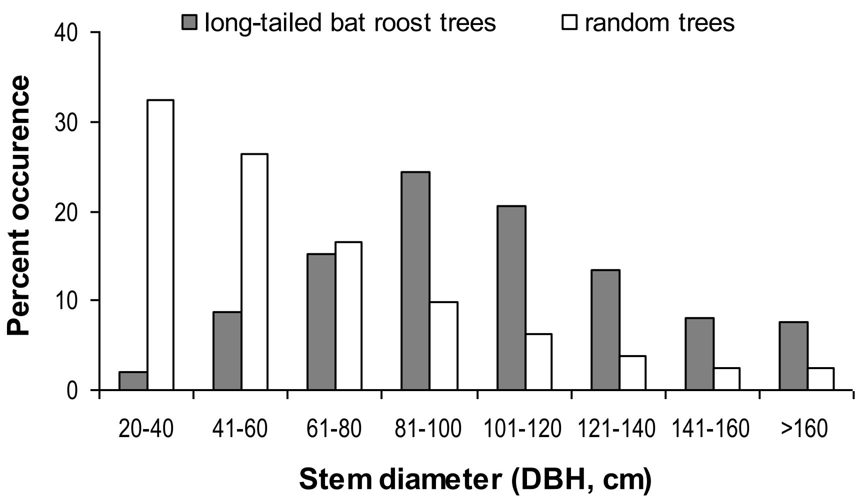


Fig. 4. Stem diameter at breast height (DBH) of beech trees used by long-tailed bats (*Chalinolobus tuberculatus*) in the Eglinton valley

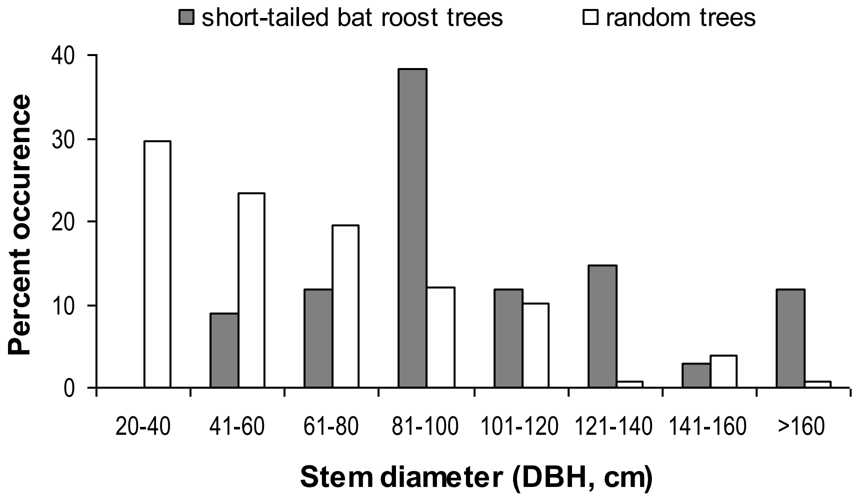


Fig. 5. Stem diameter at breast height (DBH) of beech trees used by lesser short-tailed bats (*Mystacina tuberculata*) in the Eglinton valley

3.3.3 Non-statutory advocacy and education

Non-statutory advocacy 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 could include giving public talks, organising field trips, involving the public or specific interest groups in conservation, writing factsheets, and circulating relevant information and factsheets.

Non-statutory advocacy and education are powerful tools for:

- increasing landowner awareness of preferred maternity roosts to prevent them from being felled
- increasing the security of existing known roost trees
- encouraging the maintenance of existing habitat surrounding roosts
- encouraging the enhancement and restoration of sites through replacement plantings
- undertaking relevant statutory advocacy
- facilitating the establishment of a local landcare group for bat conservation.

For example, DOC staff in South Canterbury work to stop or reduce the loss of maternity roosts to wood cutting, clearance and senescence by working with local landowners, district and regional council staff, and other interested parties.

Important messages to get across to the public include:

- the fact that there are native bats in New Zealand (promotes values/interest)
- where bats are found in New Zealand (feedback from the public/DOC on sightings)
- the importance of conserving mature native and exotic trees, including in rural/peri-urban areas (major roosting sites are still threatened by logging and tree felling)
- the need to monitor population trends (advocacy to help this happen)
- the importance of pest control for protecting bat populations

- the importance of maintaining/restoring landscape connectivity in fragmented landscapes.

Note that the messaging may need to have a different emphasis depending on the audience, with target groups including:

- landowners / farmers / forestry companies
- DOC managers and regional office staff
- territorial authorities / councils
- schools
- iwi
- Interest groups (e.g. WWF New Zealand, tramping groups, Forest & Bird)
- sponsors
- service groups
- politicians
- journalists
- research groups.

Important resources include:

- DOC website
- other web-based information on bats
- zoo exhibits (flying foxes)
- DOC visitor centres
- talks by bat workers
- newspaper, magazine, radio and television articles
- summer programmes / visitor programmes (e.g. DOC Southland's walks with the bats)
- *Dactylanthus* advocacy
- community bat conservation groups (e.g. Project Echo, Pelorus Bat Recovery Project, Catlins Bats on the Map).

3.3.4 Pest control to enhance habitat quality and reduce the risk of predation

Introduced mammalian pest species are considered a major threat to the continued viability of bat populations in New Zealand as a result of not only predation but also food competition, as possums, rodents and stoats consume large numbers of insects, and possums and rodents also reduce the availability of fruit and nectar. Therefore, integrated and effective pest control programmes that target possums, rodents, stoats and/or feral cats are likely to benefit bat populations if effort is sufficiently intensive by:

- reducing the threats of direct predation on bats
- increasing the availability of food resources for bats.

Both rats and stoats are killed during 1080 possum control programmes, although the kill rate is not always consistent, and field studies have highlighted the benefits of pest control operations to bats. For instance, Edmonds et al. (2017) concluded that the survival of the short-tailed bat population in the Eglinton valley was enhanced by a large-scale aerial 1080 operation, and O'Donnell et al. (2011) showed that the survival of tagged lesser short-tailed bats appeared to be enhanced significantly by an operation in the Eglinton valley that used pindone in bait stations. 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 (O'Donnell et al. 2017).

Biosecurity and 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 Codfish Island / Whenua Hou, Te Hauturu-o-Toi / Little Barrier Island and Kapiti Island.

Defining the management area

It is important that management areas focus on the roosting ranges of bat colonies, which requires some knowledge of where their core roosting areas are. There are three main ways of achieving this.

1. Radio tracking – The primary way of identifying roosting areas is by radio tracking a sample of bats during the breeding season and mapping their roost locations. However, radio tracking is labour intensive and a large sample of tracked bats is required to fully reveal a colony's roosting range because not all bats in the colony occupy the same roosts each day, instead moving among a network of roosts daily.
2. Large-scale management – If radio tracking is not an option, an entire forest block in which bats occur can be managed, automatically protecting the roosts within it.
3. Observations – Observers in the field can undertake watches along forest margins at dusk to identify the flyways used by bats. Automatic bat detector units can also be used to identify areas of high activity at dawn and dusk to help refine the management area within a forest. However, this technique needs to be used with caution because bats are fast flying and highly mobile, so high-use areas are not always near roosts.

Predator control operations will need to cover a large area to benefit New Zealand bats due to the large home range requirements of both species and their need for large numbers of roost trees. Large management areas are also needed to maximise the chance of lowering the densities of predators in core areas and to minimise reinvasion rates. Small management areas (< 1000 ha) have been unsuccessful in controlling rats and stoats to a level sufficient to recover long-tailed bat numbers (CFJ O'Donnell, DOC, pers. comm). Therefore, areas should be a minimum of 1000 ha and preferably in the order of 10,000 ha to ensure that bats are protected at their colonial and solitary roosts and while feeding. Roosting and feeding habitats are not randomly selected by bats, so understanding the habitat requirements of bats is important to ensure the management of appropriate sites.

Operations need not solely focus on the protection or restoration of bat populations. Existing biodiversity projects that aim to improve forest condition (e.g. Herangi Range, Eglinton valley and Pureora) will have the potential to benefit bat communities as well as many other threatened species associated with forests, such as kiwi (*Apteryx* spp.), kākā (*Nestor meridionalis*), whio / blue duck (*Hymenolaimus malacorhynchos*), mōhua / yellowhead (*Mohoua ochrocephala*) and mistletoes. However, specific projects may be required to protect *Dactylanthus taylorii* sites or special sites where significant bat populations occur but there are no other threatened species (e.g.

the Hanging Rock area in South Canterbury and Grand Canyon Cave in Maniapoto; O'Donnell 2000a, unpublished, see section **Error! Reference source not found.**, Notes; O'Donnell 2002).

Poisoning risks to bats in pest control areas

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

Long-tailed bats are likely to be at low risk from toxins because they rarely feed within forests and they feed entirely on the wing on flying insects that would be unlikely to come into contact with baits. Additionally, these bats are much less active in winter when many aerial pest control operations are planned. However, brodifacoum residues were found in one dead long-tailed bat at Grand Canyon, King Country (T Thurley, DOC, pers. obs.), and diphacinone residues have been detected in long-tailed bat guano collected at Pureora during summer (Dennis 2019).

By contrast, the feeding habits of lesser short-tailed bats make them vulnerable to toxins through both direct consumption and secondary poisoning through the consumption of arthropods that have fed on baits (Lloyd and McQueen 2000; Sherley et al. 2000), as high concentrations of 1080 and other vertebrate pesticides can persist in arthropods for several days after they have consumed baits (Eason et al. 1993; Booth and Wickstrom 1999; Fisher et al 2007).

Short-tailed bats were killed during a rodent control operation that used diphacinone in a cereal paste matrix (RatAbate®) (Dennis and Gartrell 2015), and 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 showed that they did not consume carrot- or grain-based baits that are commonly used with 1080 and second-generation anticoagulants (Lloyd 1994), and nor did they consume a non-toxic version of the cereal paste matrix that caused the death of wild bats when laced with diphacinone (Dennis 2019).

Several replicate trials would be required in a variety of circumstances and forest types before a generalised conclusion could be justifiably drawn about the mortality of lesser short-tailed bats during aerial and ground-based poisoning operations. However, studies in the field have given similar results, with no harmful impacts being detected in short-tailed bat populations that were monitored through two aerially broadcast poisoning operations using pollard baits – on Codfish Island / Whenua Hou using brodifacoum (Sedgeley and Anderson 2000) and in Rangataua Forest in the central North Island using 1080 (Lloyd and McQueen 2002). And while residues of diphacinone were detected in communal short-tailed bat guano deposits at Pureora during a rodent control operation using cereal pellet baits enclosed in bait stations, survival analysis and monthly measurements of prothrombin (blood clotting) time suggested that exposure of the bats to diphacinone was subclinical (Dennis 2019).

The risk to lesser short-tailed bats is greatest in areas where new baits or lures are being proposed for use. 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. Therefore, such proposals need to be carefully evaluated through non-toxic bait trials.

3.3.5 Protection of roost sites

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

Minimising the disturbance of cave- and rock-roosting bats is essential. Disturbance in caves in winter can reduce survival significantly. For instance, an apparent decline in the number of bats day-roosting in Grand Canyon Cave over the 5 years from 1992 to 1997 coincided with increased human use of the cave (C Smuts-Kennedy, DOC, pers. comm.), and there have been records of instances where all bats in the cave have taken flight while people have been watching them (N Miller, DOC, pers. comm.). Disturbance by rock climbers was also identified as a potential risk to long-tailed bats at Geraldine, particularly in winter when bats use the same limestone crevices as climbers (Griffiths 1996).

3.3.6 Protection of freshwater and terrestrial foraging habitats

The protection of foraging habitats can be achieved through statutory and non-statutory advocacy, the legal protection of significant sites, and the active management of sites that are 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. Because there have not yet been comprehensive surveys of all areas that are likely to have bats, surveys using bat detectors should be undertaken in areas near significant habitats that have already been identified.

3.3.7 Restoration of roosting and foraging habitats

There has been a significant loss of maternity roosts to habitat clearance, wood cutting and natural aging of remnant stands of native forest in some areas (O'Donnell 2000a). It is unlikely that many new roosts are being formed in South Canterbury because grazing inhibits forest regeneration, forcing bats to use poor quality roost sites, which results in low breeding success (Pryde et al. 2006), and other fragmented landscapes where bats are still present are likely to be under similar pressure – for example, a studied long-tailed bat population in the Waikato is under pressure from roading and housing development projects, causing the loss of roosts and habitat.

Maintaining and improving the habitat that bats currently use is likely to be essential to allow bats to persist in fragmented landscapes. It may be possible to restore bat communities within areas where numbers have declined by restoring forest and wetland remnants on agricultural land (foraging habitat). To achieve this, it is essential that the further loss of trees is reduced, and roosting tree species may also need to be planted in areas where natural regeneration has been inhibited. Forest remnants that contain roost sites are also likely to benefit from fencing and the exclusion of stock, which could take the form of the low-cost fencing of small patches around roost sites in areas where it is not possible to fence extensive areas of habitat, as well as predator control over a wider area.

Providing predator-proof artificial roost boxes may also be of benefit, although evidence of their effectiveness is limited and requires further study (Jones et al 2019). Artificial roost boxes have been used with very limited success in South Canterbury, with occasional use in the first 2 years but no evidence of their use after 5 years (CFJ O'Donnell, DOC, pers. obs.). By contrast, several artificial roost boxes have been used by long-tailed bats in Hamilton (Davidson-Watts 2019). However, artificial roost boxes require ongoing maintenance and are less effective at buffering temperature fluctuations than natural roosts, so they should only be used as a short-term measure in areas where there are currently few suitable roost trees until natural roosts are restored.

3.3.8 Translocations

No accepted techniques are currently available for translocating New Zealand bats to new sites. Two attempts to translocate short-tailed bats have been unsuccessful: 50 bats were moved from Codfish Island / Whenua Hou to Ulva Island, but were assumed to have subsequently returned to Codfish Island / Whenua Hou; and an attempt to translocate recently fledged juvenile short-tailed bats to Kapiti Island in 2005–2006 was only partially successful, with 60% of the bats surviving and remaining on the island in the first 9 months after release but then suffering from an unidentified infection that caused severe ear lesions (L Adams, DOC, pers. comm.).

3.4 Monitoring outcomes

Monitoring programmes should focus on monitoring several representative populations or operations so that the difference that is being made by management can be measured and reported, allowing appropriate management to be applied at other sites. For further information, see the Biodiversity and Inventory Monitoring Toolbox on the DOC website.

4. Finding bats with bat detectors

4.1 Bats and echolocation

Echolocation, or biosonar, is the biological sonar used by several mammals, such as dolphins, shrews, most bats and most whales. The term ‘echolocation’ 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, orientate and forage, often in total darkness. Bats generate high-frequency (ultrasonic) sound via the larynx and emit rapid ultrasonic pulses through the mouth or, less commonly, the nose. 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. Consequently, echolocation calls provide an opportunity to unobtrusively survey, monitor and identify bat species (Catto 1994; de Oliveira 1998; Russ 1999).

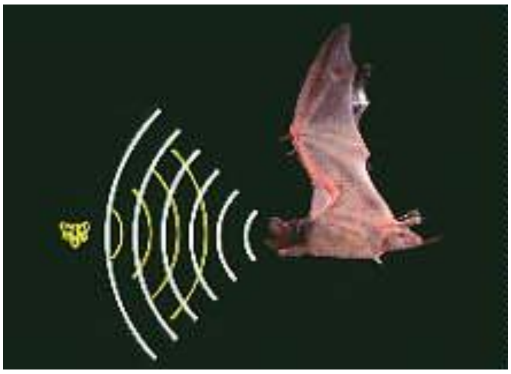


Fig. 6. Diagrammatic representation of a bat echolocating

4.2 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 for studying bats. Bat calls are picked up by the detector’s microphone and can be transformed into lower frequencies that humans can hear and/or displayed on a screen. There are three main types of hand-held bat detectors.

- Heterodyne
- Frequency division
- Time expansion

Each type uses a different technique for transforming ultrasound into audible sound, and some models can be used in conjunction with custom software to display a full-spectrum spectrogram using a phone, tablet or laptop (e.g. the Echometer Touch 2; see Fig. 7D below).

4.3 Choosing a 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 the calls collected, in the methods of storing, visualising and analysing calls, and in their ability to distinguish between calls made by different bat species. Therefore, the choice of bat detector will ultimately depend on its application. This section provides some background information on the three types of bat detectors and their relative advantages and disadvantages, as well as an overview of automatic bat detector and recording systems, to assist in selecting the most appropriate detector for specific research or survey and monitoring needs. More technical information (e.g. how the detectors work, options for storing calls, analysis techniques) can be found in Parsons and Obrist (2004). Several companies supply bat detectors commercially (e.g. Batbox Ltd, Pettersson Elektronik, Titley Scientific, Tranquility and UltraSound Advice), and some companies produce several types of detectors.

4.3.1 Heterodyne detectors

Heterodyning is a real-time method (i.e. you can hear the sound from the detector at the same time as it is being 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 and Walsh 1994) and the field (Parsons 1996). The most common bat detectors used in New Zealand are the Batbox III heterodyne bat detector and its successor, the Batbox IIID, which has the same sensitivity (Batbox Ltd, UK) (Fig. 7A). DOC uses both these detectors as the standard for surveying using handheld bat detectors along line transects (refer to the Toolbox method *Bats: Counting Away From Roosts – Bat Detectors on Line Transects* [see Appendix 1]).

Advantages

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

Disadvantages

- The narrow frequency band of tuneable detectors means that all bats calling outside the tuneable frequency range will be missed. Therefore, this kind of detector is of limited value in countries where there are numerous bat species calling at different frequencies.
- The 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 and Obrist 2004).

- It is often 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 of overlap in their calls, and the output from heterodyne detectors does not provide enough information to distinguish between them in all situations (see Species identification using bat detectors).
- It is crucial to calibrate heterodyne detectors before using them in the field to ensure that the frequency settings are correct (see Bat detector sensitivity and calibration below).

A

B

C

D

Fig. 7. Examples of hand-held bat detectors that use different methods for transforming ultrasound into audible sound. (A) Batbox Ltd Batbox III D heterodyne detector; (B) Titley Scientific Anabat Scout heterodyne and frequency-division detector; (C) Pettersson Elektronik D1000X heterodyne, frequency-division and time-expansion detector; (D) Wildlife Acoustics Inc. Echo Meter Touch 2 detector with heterodyne and real-time expansion modes (as well as visual output of a full-spectrum spectrogram).

4.3.2 Frequency-division detectors

Frequency-division (also called countdown or broadband) detectors transform the entire ultrasonic frequency range of a bat call without tuning. The output from frequency-division detectors is usually recorded onto an SD card. Computer software can then be used to visualise and analyse the call structure and aid species identification. In Australia, the Anabat system is widely used (Fig. 7B), which can store the recorded calls onto an SD card for later analysis. Calls are examined by first digitising them onto a computer using a zero crossing or full spectrum interface module (see Box 1 below) and then using Anabat Insight software to visualise and analyse the recorded calls (Parsons and Obrist 2004; Reardon 2010).

Advantages

- Frequency-division detectors 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.
- Among the Anabat systems, the Walkabout and Scout can store the recorded calls, while the Swift and Express have delay or time-switches, making these systems very suitable for remote/unattended surveys.
- The output from frequency-division detectors can contain more information than that from heterodyne detectors, including characteristics such as the maximum, minimum and average frequency, duration and time between calls.
- The output from frequency-division detectors contains 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 detectors (Parsons 1996).
- If the division ratio is set too low, calls of bats using high frequencies may be lost (although this probably is not an issue with New Zealand bat species).
- The methods by which frequency-division detectors transform bat calls can lead to misleading outputs (i.e. the outputs will not always accurately represent all the characteristics of a bat call) (Parsons and Obrist 2004).
- In Australia, there are several pairs or groups of bat species that cannot be reliably distinguished using the Anabat systems (Reardon 2010).
- Frequency-division systems (detectors, additional hardware and software) are more expensive than heterodyne detectors.

4.3.3 Time-expansion detectors

Time expansion is not a real-time method of transforming bat calls. Time-expansion detectors work by digitising a high-frequency output from the microphone at a high sampling rate. 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 and Obrist 2004). The slower speed reduces the frequency to an audible level that can 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 compact flash card. 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 Elektronik'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 and Obrist 2004) (e.g. Fig. 8).

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 that are 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.

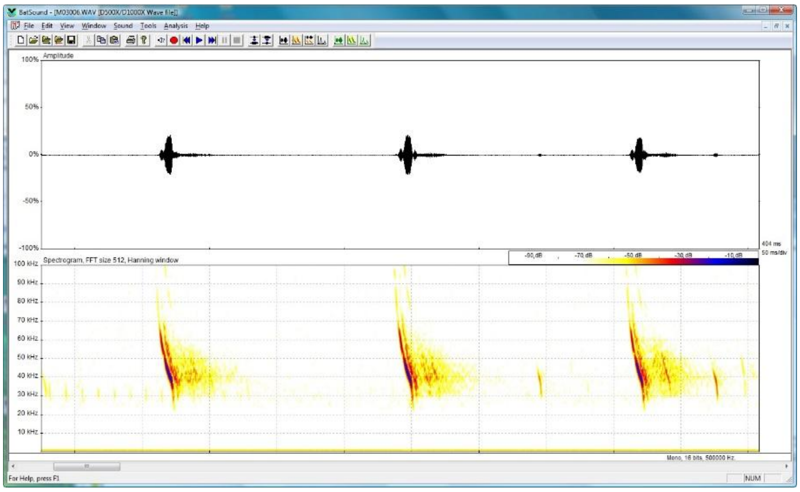
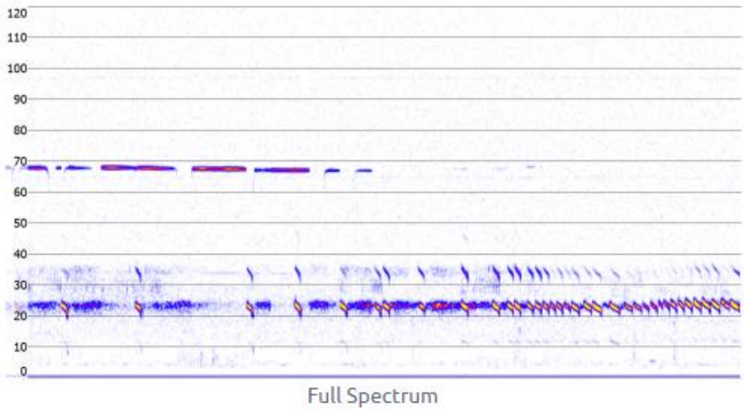
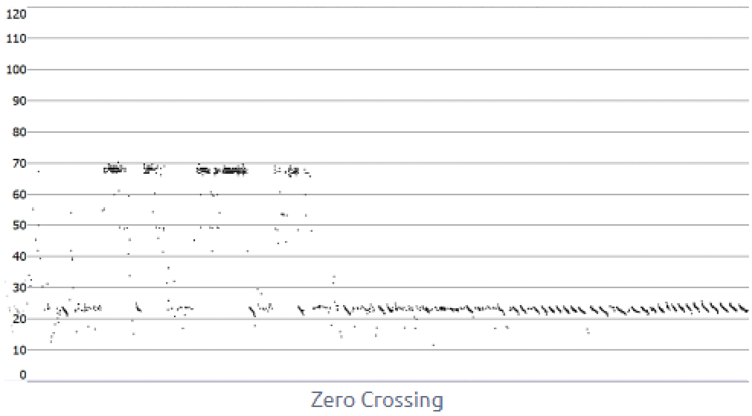


Fig. 8. Example output from a time-expansion bat detector system analysed using BatSound software from Pettersson Elektronik

Box 1 What is the difference between full spectrum and zero-crossing analysis?

There are two main recording formats for bat files: full spectrum (.wav) and zero-crossing analysis (.zc, .zca and .xx#). Full spectrum records the full spectral information within a sound file, just like a music file, whereas zero-crossing analysis renders the spectral information down into a series of time vs. frequency dots (see figure below).





Example outputs for full spectrum and zero-crossing recording formats

The advantages of full spectrum include the ability to see the call intensity, harmonics, multiple bats calling at the same time and faint bat pulses during high ambient noise, while the disadvantages include the much larger file size (typically 6 times the size of a zero-crossing file), the need for more processing power to record and the reduced speed of rendering on a computer for post-recording analysis.

The advantages of zero-crossing analysis are the small file size, meaning that memory space is not an issue, and the fact that many published guides to bat calls are based on zero-crossing analysis, while the disadvantages include the loss of spectral information (which may be helpful for species identification) and the fact that bat calls may not be recorded in full if there is high-frequency ambient noise (e.g. from insects).

4.3.4 Automatic bat detector and recording systems

Bat detectors can be used manually or remotely, and the outputs from detectors can be recorded and stored using an automatic bat monitor (ABM), which may also be referred to as an acoustic recorder or an automatic bat detector. Recording the output from a detector allows a permanent record of part or all of the night's activity to be kept.

Several automated systems have been developed that use different types of bat detectors and different methods for storing data. Consequently, their relative effectiveness and costs vary (Parsons and Obrist 2004). Many systems include timers and audio-activated delay switches that allow units to be left in the field and activated only when a call is detected, and the data are most frequently stored on SD cards.

The DOC Electronics Team developed a frequency compression recorder, the AR4 acoustic recorder, to provide conservation practitioners in New Zealand with a quality acoustic recorder at low cost (Fig. 9). This device has been designed to be light weight, weatherproof/waterproof, and small and easy to use, and includes the option of using inbuilt recording protocols for standardised monitoring. It can be set and left in the field for several weeks to record at set time periods and uses readily available low-cost consumables (AA batteries and SD cards). It also has an inbuilt GPS receiver for accurate location and time-stamping of recordings. The recorder is 'triggered' and records files when it detects bat-like sounds, and the files are saved as .BMP – a bitmap image of a compressed spectrogram. The files can be processed using the custom software BatSearch, which is provided by the Electronics Team, or by open-source software developed by AviaNZ. AR4 recorders are no longer available for purchase, however they have been superseded by the AR5 model, essentially the same as the AR4, and available from Alato (AR5 Acoustic Recorder).

Fig. 9. Department of Conservation Te Papa Atawhai AR4 acoustic recorder

The Anabat passive detectors (Swift and Express) from Titley Scientific (Fig. 10) are widely used for remote surveys in Australia. These detectors, which can record in both full spectrum and zero-crossing format, are well suited to unattended bat detector surveys, as they are weatherproof, have a built in GPS, use AA batteries and store files onto SD cards. Analysis of the output files requires the free Anabat Insight analysis software.

Fig. 10. Anabat Express passive detector

AudioMoth is another low-cost, full-spectrum, open-source acoustic monitoring device for monitoring wildlife (Fig. 11). This device is sensitive to both audible sounds and sounds that are well into the ultrasonic frequency range, making it suitable for use with bats. At this stage, it has not been used in

New Zealand, probably due to DOC's in-house production of the AR4, but some projects may consider it as an option given its lower cost. For further information, visit the Open Acoustic Devices website.

Fig. 11. AudioMoth acoustic monitoring device

4.4 Species identification using bat detectors

Echolocation calls provide an opportunity to unobtrusively survey, monitor and identify bat species (Catto 1994; de Oliveira 1998; Russ 1999). Detectors can be used manually (e.g. using hand-held bat detectors to count long-tailed bat calls along line transects) or remotely using automatic systems (see above).[2] In this section, we provide an overview of the call characteristics of New Zealand's bats and some information on how to use bat detectors to distinguish between them.

4.4.1 Call characteristics

The call structure and pulse repetition rate of 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. The peak amplitude of long-tailed bat calls is 40 kHz (Parsons 2001), and a Batbox III on full volume can detect long-tailed bats on average 43.5 ± 9.8 m away along forest edges (CFJ O'Donnell, unpubl. data). By contrast, the peak amplitude of lesser short-tailed bat calls is c. 27–28 kHz (Parsons 2001) and their calls can be detected from approximately 30 m away (Cockburn S, DOC, pers. comm)).

Call overlap

Unfortunately, there is some overlap in call structure (fundamentals and harmonics) between long-tailed bats and lesser short-tailed bats, as well as a significant overlap in frequency when calls are monitored using heterodyne detectors (Parsons 2001). This means that acoustic detectors set at 27–28 kHz will pick up the calls of both long-tailed bats and lesser short-tailed bats, and detectors set at 40 kHz will also pick up the calls of both bat species. Therefore, if walking-transect surveys are conducted using heterodyne detectors in a new area, or in an area where both long-tailed bats

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[2] For further information on surveying using line transects, refer to the Toolbox method *Bats: Counting Away From Roosts – Bat Detectors on Line Transects* (see Appendix 1) and O'Donnell and Sedgeley (2001). For more information on using automatic systems for inventory and monitoring, refer to the Toolbox method *Bats: Counting Away From Roosts – Automatic Bat Detectors* (see Appendix 1).

and lesser short-tailed bats are known to be present, it cannot be assumed that every call detected at 40 kHz is being made by a long-tailed bat and that every call recorded at 27–28 kHz is being made by a lesser short-tailed bat.

Key differences

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

Bat detectors and ABM units are important tools for determining the presence or absence of bats in an area. Both species of bats are endangered, so any record of bats from a new area is valuable, even if call identification is not 100% positive. Fortunately, with the AR4 and AR5 acoustic recorders, the echolocation sequences of the two New Zealand species are generally easy to tell apart, although there are situations where both species can alter the characteristics of their echolocation calls to the point that it can be difficult to distinguish them definitively. Recommended bat detector frequencies

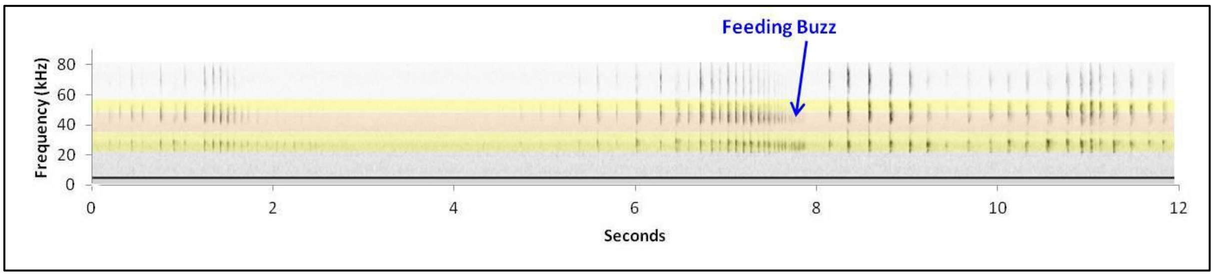
We recommend that the following frequencies are used on handheld detectors.

- **To detect long-tailed bats, set detectors at 40 kHz.**
  At 40 kHz, long-tailed bat calls are often loud and have longer call durations and slower pulse repetition rates than lesser short-tailed bat calls. Long-tailed bat calls have a relatively irregular rhythmical sound that can be described as a series of ‘slaps’ or ‘thwacks’.
- **To detect lesser short-tailed bats, set detectors at 27–28 kHz.**
  At 27–28 kHz, lesser short-tailed bat calls are often softer (unless the bat flies very close to the microphone) and have shorter durations and faster pulse repetition rates than long-tailed bat calls. Lesser short-tailed bat calls have a more even or regular rhythmic pattern that can be described as a short burst of staccato ‘clicks’.
- **To detect both species with a single ABM unit, use the AR4 or Ar5 acoustic recorder.**
  Both species can be reliably identified using the AR4 or AR5 acoustic recorder set on the Bat protocol and recording throughout the night. Although the feeding buzzes made by long-tailed bats, which are below 40 kHz, could be confused with the calls of lesser short-tailed bats, they are recorded far less often than the usual characteristic long-tailed bats calls and seldom occur by themselves, usually being heard at the end of a series of the more usual calls.

4.4.2 Examples of calls

Examples of the calls of long-tailed bats and lesser short-tailed bats obtained using AR4 or AR5 bat recorders are shown in Fig. 12 and further examples are provided in the *Bat Call Identification Manual for DOC’s Spectral Bat Detectors* (see Appendix 1) and Dennis (2021, unpublished, see section **Error! Reference source not found.**, Notes).

A



B

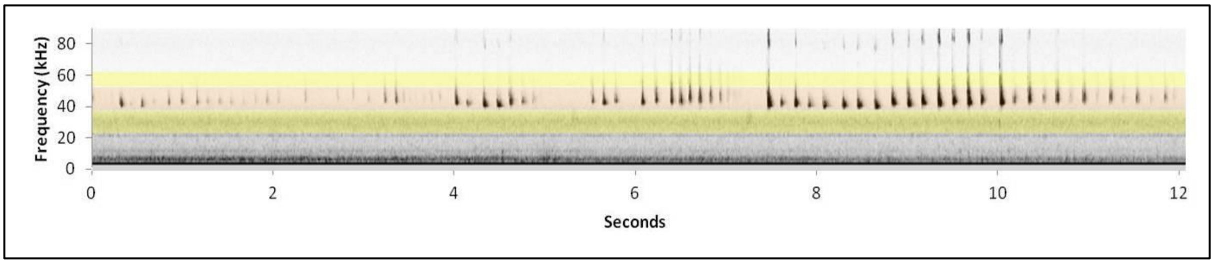


Fig. 12. Examples of calls of (A) short-tailed bats (*Mystacina tuberculata*) and (B) long-tailed bats (*Chalinolobus tuberculatus*) collected using the AR4 bat recorder

4.4.3 Bat detector sensitivity and calibration

It is important to test bat detectors before use, particularly if using old, used equipment. The sensitivity of bat detectors can vary between units (O'Donnell and Sedgeley 1994; Arkins 1999, unpublished, see section **Error! Reference source not found.**, Notes), with low sensitivity most commonly being due to under-charged batteries, damaged microphones and miss-aligned frequency dials. Therefore, it is recommended that all equipment is serviced at the end of each field season.

The easiest way to check the sensitivity and 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 detector's speaker at a distance of 40−50 m, provided that 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 and Sedgeley 1994).

The sensitivity of different brands and models of detectors can also vary (Parsons 1996; Smith et al. 2020). Therefore, if it becomes necessary to change the brand/model of detector at some point during long-term, standardised surveys, it is important to calibrate the gain between different brands/models to ensure that the receiving area of the new detector is comparable (O'Donnell and Sedgeley 1994: appendix 3).

The sensitivity of AR4 or AR5 bat recorders can be tested using the bat recorder tester app that is available on the Electronics team support and service page on the DOC website.

4.4.4 Distinguishing bat calls from other sounds on the bat detector

On heterodyne detectors, bat calls are heard as series of clicks as a bat flies into range. Each sequence of two or more audible echolocation clicks is defined as a 'bat pass' (Furlonger et al. 1987), and a period of silence separates 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' and 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 grab 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 such as cicadas and crickets can be very noisy on warm summer nights, 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, and non-bat sounds are more likely to be stationary and close to the ground. If the battery in the detector gets low, it can also create a feedback noise through the speaker.

5. Species identification in the hand

Long-tailed bats and lesser short-tailed bats are very easy to distinguish in the hand without the use of an identification key. This section describes the key differences between these 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 behaviours, as discussed in other sections in this report (see Handling, examining, measuring and releasing bats, Finding bats with bat detectors and Introduction to New Zealand bats).

Comparatively little is known about the characteristics of greater short-tailed bats, most of the distinguishing features of which are summarised in section 5.2.

The main differences among the three species are summarised in Table 2. For definitions of the various technical and morphological terms, see Handling, examining, measuring and releasing bats, particularly Fig. 46.

Table 2 Summary of the distinguishing features of the three species of New Zealand bats. Source: O'Donnell (2005).

	LONG-TAILED BAT *Chalinolobus tuberculatus*	LESSER SHORT-TAILED BAT *Mystacina tuberculata*	GREATER SHORT-TAILED BAT *Mystacina robusta*
Onset of flying activity	Around sunset	After dark	After dark
Roost sites	Native and exotic trees, and caves	Native trees and caves	Native trees, caves and seabird burrows
Tail length and position	Almost as long as head and body; large 'V'-shaped interfemoral membrane; post-calcarial lobe present	Very short, partly free of interfemoral membrane, projecting c. 7 mm on dorsal surface	
Fur	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	Short and velvety, grey-brown, guard hairs over underfur	
Jaws	Fleshy lip-lobule at corner of mouth	No lip-lobule	
Hind legs	Small, delicate feet; legs enclosed within interfemoral membrane	Large, robust legs, not fully enclosed by interfemoral membrane	
Claws	Without spurs on toes and thumbs	With spurs	
Ears	Small, broad and rounded	Large and pointed, extend to or beyond muzzle when laid forwards	Large and pointed, do not reach muzzle when laid forwards
Nostrils	Small	Prominent and narrow	Short and broad
Forearm length	37–46 mm	39–46 mm	45–48 mm

5.1 Comparison of long-tailed bats and lesser short-tailed bats

Long-tailed bats and lesser short-tailed bats have very different appearances in the hand. Long-tailed bats are relatively small and delicate, with an adult body mass of 8.5–12.3 g and a forearm length of 38.7–40.5 mm, have chocolate brown or chestnut brown fur, and are generally docile when handled. By contrast, lesser short-tailed bats are stocky in appearance and can be a third larger than long-tailed bats, with an adult body mass of 11.4–22.0 g and a forearm length of 36.9–46.9 mm, have larger, more pointed ears, have fur 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).

5.1.1 Fur colour

Long-tailed bats

Fur colour is variable in long-tailed bats and also changes with age and can differ between adult males and females. Adult females usually have rich chestnut upper parts, sometimes with white tips to the fur, while 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. 14). In both sexes, the underparts are pale brown and 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), and the limbs, wing and tail membranes are almost naked and blackish-brown in colour (Fig. 14).

A

B

Fig. 14. Fur colour in long-tailed bats (*Chalinolobus tuberculatus*). (A) Adult female – note the chestnut fur colouring; (B) male/juvenile – note the darker fur colour. *Photos: (A) CFJ O'Donnell; (B) R Morris*

Lesser short-tailed bats

The fur of lesser short-tailed bats is generally grey-brown and is short, dense and velvety, sometimes appearing frosted. Guard hairs are present over the under-fur. As for long-tailed bats, fur colour can vary with age, with the fur of adults sometimes appearing 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. 15).

A

B

Fig. 15. Fur colour in adult lesser short-tailed bats (*Mystacina tuberculata*). *Photos: (A) BD Lloyd; (B) R Morris*

5.1.2 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. 16). 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).

A

B

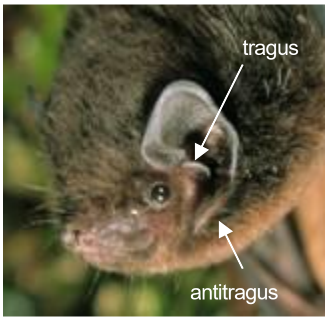


Fig. 16. Ears of long-tailed bats (*Chalinolobus tuberculatus*). The ears are smaller and more rounded than those of lesser short-tailed bats (*Mystacina tuberculata*), and the tragus is small and rounded. *Photos: (A) B Ebert; (B) R Morris*

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. 17A) (Lloyd 2005a, 2005b). When handled, lesser short-tailed bats can hold their ears in an upright position (Fig. 17B), partially curl their ears (Fig. 17C) or fold their ears relatively flat against their heads (Fig. 17D).

A

B

Fig. 17. Ears of lesser short-tailed bats (*Mystacina tuberculata*). (A) The ears are longer than those of long-tailed bats (*Chalinolobus tuberculatus*) and the tragus is long and pointed. (B) Ears fully erect. (C) Ear partially curled. (D) Ears flat. *Photos: (A) D Veitch; (B and C) J Sedgeley; (D) BD Lloyd*

5.1.3 Legs and tails

Long-tailed bats

The legs and tail of long-tailed bats are fully enclosed within a large V-shaped tail membrane and the tail is almost as long as the head and body combined (Fig. 18). 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 also 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.

A B

Fig. 18. Tails of long-tailed bats (*Chalinolobus tuberculatus*). The tail is long and fully enclosed within a large V-shaped membrane. Note the dark colour of the tail and wing membrane. *Photos: (A) C Hillock; (B) CFJ O'Donnell*

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 the dorsal surface of the membrane. The tail membrane is much shorter and more rounded than that of long-tailed bats. Like long-tailed bats, lesser short-tailed bats also 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. 19). The legs of lesser short-tailed bats are unusually robust, and their feet are stout and broad (c. 6 mm long) (Fig. 19). They also have a very fine talon that is 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

Fig. 19. Tail and hind legs of lesser short-tailed bats (*Mystacina tuberculata*). (A) Dorsal view showing the tail membrane tightly furled away and the tail protruding freely; (B) ventral view showing the tail membrane almost fully extended; and (C) dorsal view showing the tail membrane fully extended. *Photos: (A) L Lumsden; (B) J Sedgeley; (C) D Mudge Ngā Manu Images*

5.2 Summary description of greater short-tailed bats

Greater and lesser short-tailed bats share many general characteristics but, overall, greater short-tailed bats are larger and more robust (Fig. 13, Table 3), especially in the skull, and their molars and jaws are also proportionately larger. However, proportionately shorter ears, forearms and wing elements in greater short-tailed bats give rise to some overlap between measurements from the two species (Daniel 1990; Lloyd 2005a, 2005b). The potential for overlap is higher in southern New Zealand because the 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).

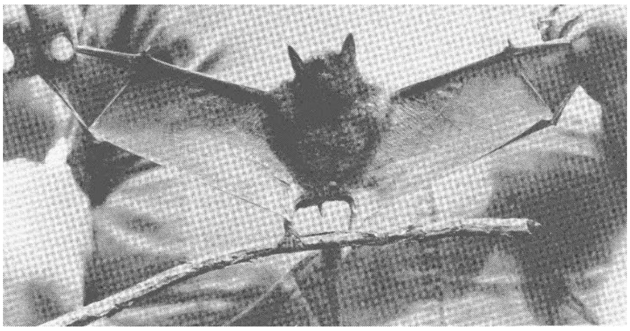



Fig. 13. Greater short-tailed bats (*Mystacina robusta*). *Photos: D Merton*

A 2004 study examined dental and skeletal measurements of lesser short-tailed bats and greater short-tailed bats from adjacent populations in southern New Zealand where the mean size of these species was closest: measurements were compared between 20 lesser short-tailed bats from Codfish Island / Whenua Hou and 8 greater short-tailed bats from the southwest Tītī / Muttonbird Islands off Stewart Island/Rakiura. It was found that the two species had complete size separation in most of the measured variables and minimal overlap in only a few (five tooth and two cranial measurements). All of the greater short-tailed bats were larger than any of the lesser short-tailed bats, had relatively smaller wing elements and had different tooth shapes. The percentage difference in size between the lesser short-tailed bats and greater short-tailed bats varied from 15% to 20% for many of the measurements but reached 34.5% for molar (M3) width and was lowest for forearm length (9.8%) and associated radius length (11.5%) (Worthy and Scofield 2004). Norberg and 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, indicating that they would have been faster in flight, with more rapid turns (i.e. more agile) but requiring larger turning radii (i.e. less manoeuvrable).

Table 3 Greater short-tailed bat (*Mystacina robusta*) measurements (mm). Data obtained from Lloyd (2005a, 2005b), Daniel (1990), Hill and Daniel (1985) and Dwyer (1962).

	TOTAL LENGTH	SNOUT-TO-VENT LENGTH	WINGSPAN	FOREARM LENGTH	TIBIA LENGTH	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. Since the greatest differences between greater and lesser short-tailed bats are largely skeletal, 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. Therefore, 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 the muzzle when laid forwards. It is extremely important to take as many measurements as possible, photographs with a scale indicated and, if possible, tissue samples (see Collecting samples).

6. Inventory and monitoring methods for counting bats

A large number of Inventory and Monitoring Toolbox specifications are available for New Zealand bats, all of which are available on the Bat inventory and monitoring page on the DOC website.

- Introduction to bat monitoring
- Bats: counting away from roosts—bat detectors on line-transects
- Bats: counting away from roosts—automatic bat detectors
- 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—field sign
- Bats: roost occupancy and indices of bat activity—automatic bat detectors
- Bats: casual reports

The best method will depend on the objectives or questions leading to the work you are doing, as well as the relative suitability for long-tailed bats or lesser short-tailed bats, as not all methods are appropriate for both inventory and monitoring or suitable for both bat species.

The first step is to read *Introduction to Bat Monitoring* (see Appendix 1), 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 for answering specific inventory and monitoring questions. These 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 then further arranged by methods that count bats away from roost sites, at roost sites or at both locations. There are 14 methods in total: 4 methods for use away from roost sites, 8 for use at roost sites and 2 that can be used in both situations. All methods are listed in 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 (e.g. 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 one or more methods, read the method specifications carefully to ensure that they meet your study objectives.

7. 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, 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 their home ranges and habitat use. Bats may be caught both in free flight and at their roosts using a variety of techniques.

Techniques that are commonly used 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. When deciding on a capture method, consideration should be given to how captured bats will be removed and handled in a timely manner. Methods that have not been trialled in this country but are proven techniques for other bat species are also mentioned briefly.

7.1 Mist nets

7.1.1 Description

Mist nets are made of fine nylon or polyester netting and are most commonly used in New Zealand for catching birds. 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 are usually 2 m high. The height of the net is divided into pockets or bags by three to five horizontal strings (shelf-strings) running the length of the net. Bats are usually trapped in these loose pockets of netting (Fig. 20). A tightly tensioned net with no pockets is much less likely to catch bats because they may simply bounce off.

Mist nets are also 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, while larger sized mesh might allow bats to fly through. Nets that are commonly used to catch birds are adequate for catching bats (30–40 mm mesh size), but specially designed bat nets may be preferable. Ecotone and Avinet are the most common net suppliers with specialised bat nets that have a mesh size of 38 mm and reduced pocket sizes to lessen the risk of entanglement. Silk nets from Germany (Solida Safety Line; 22 dtex, 4 m high with four panels) have also recently been used in the Eglinton valley and, although fragile, have been very successful at catching bats.

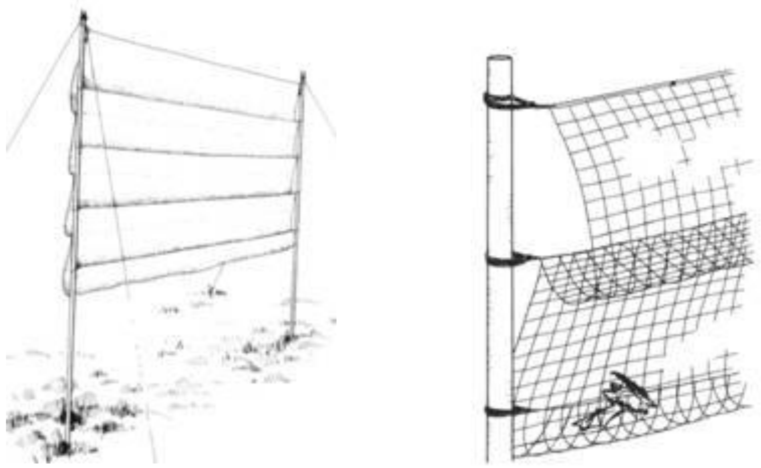


Fig. 20. Mist net setup with poles. The height of the net is divided into pockets by five shelf-strings running the length of the net, and bats are usually trapped in these loose pockets of netting. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

Advantages

- Mist nets are much less expensive than commercially available harp traps.
- Mist nets are lightweight and easily transportable when folded away into small bags.
- 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 become badly tangled in nets, so this technique may be more stressful for 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.

7.1.2 Setting the nets

When selecting a site to set a mist net, look for potential travel corridors (along streams, paths or open flyways).

Mist nets can be set using poles or with rope/string, and they can be set one net high or stacked one on top of the other (Fig. 21A). If more than two nets are set on top of each other, it is recommended that a pulley system is employed so that bats caught in the top portions of the net can be lowered easily (Kunz and Kurta 1988; Dilks et al. 1995).

A B

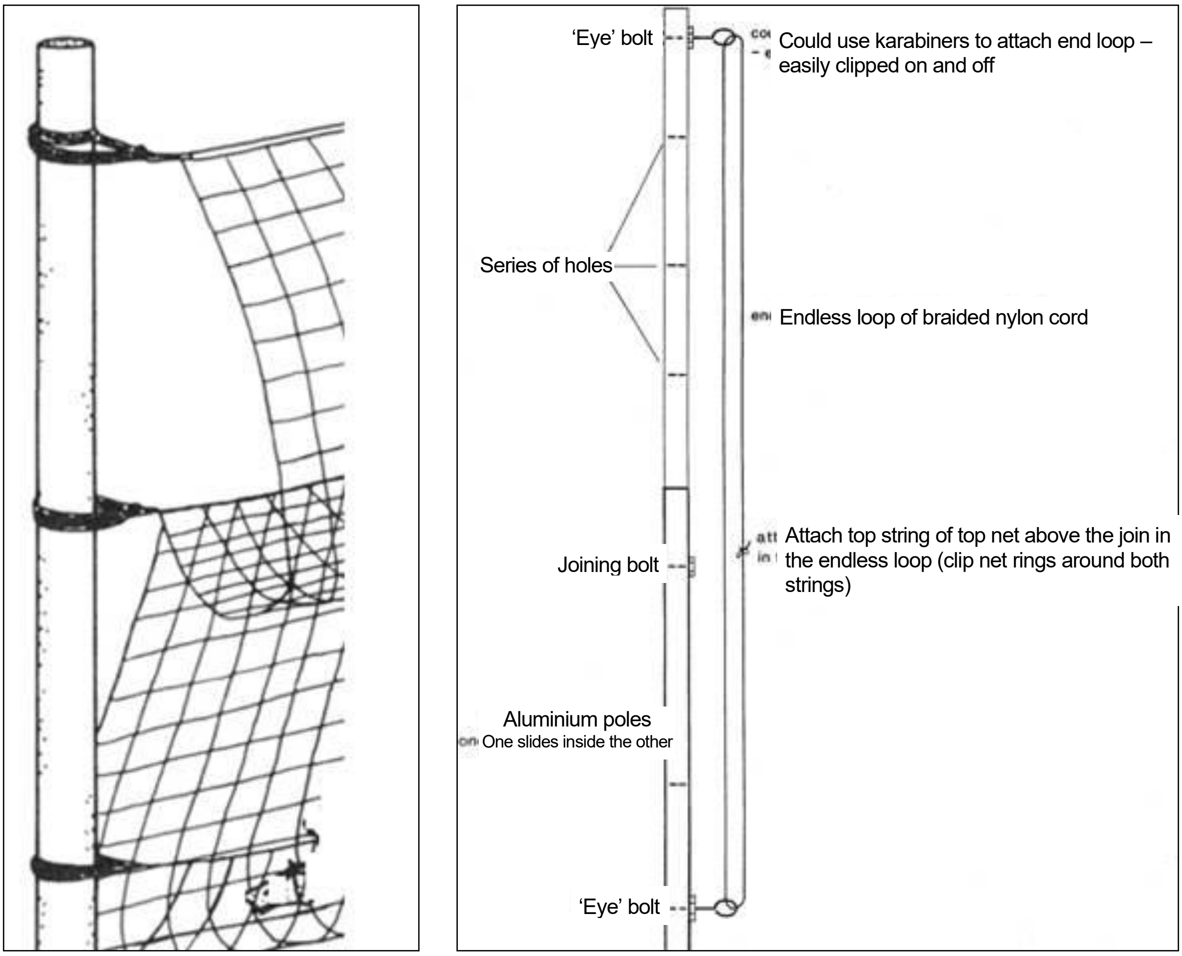


Fig. 21. Attaching mist nets to poles. (A) Wrapping end loops around the pole. (B) Aluminium pole with a simple pulley system attached for raising and lowering the net. Adapted from Dilks et al. (1995).

Pole systems

Ready-made mist net poles can be bought from retailers or, alternatively, aluminium tubing can be used as 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 with different diameters so that the smaller tube can slide inside the larger. The length of the pole can then be varied by placing a bolt 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 and Kurta 1988; Dilks et al. 1995; Fig. 21B).

An alternative method is to glue a shorter length (25 cm) of smaller diameter tubing into the top of a 1-m-long section of tube so that 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 that are 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 of the pole-holder is sharpened to create a ‘spade-point’ so that it can be hammered or pushed into the ground (Churchill 1998). .

Rope systems

Cord or string can be used instead of poles (Fig. 22). An endless loop of cord is passed over a branch and under a tree root or log. The addition of a karabiner or metal ring at the top and bottom, through which the endless loop is 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, however, else it can be difficult to achieve suitable net tensioning.

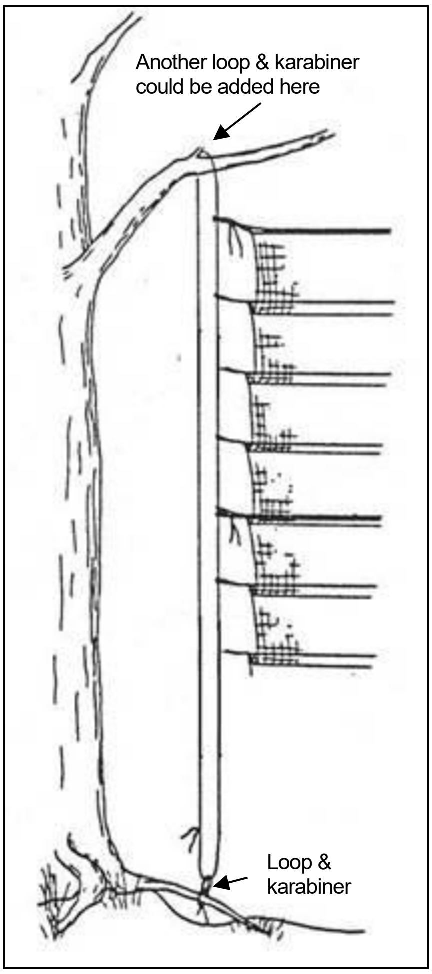


Fig. 22. A simple mist net rig using string instead of poles

If there are no supporting branches in the right places, a canopy rig can be erected, whereby the vertical endless loops are suspended from a horizontal top rope. Karabiners, metal rings or small pulleys are attached to the top rope at the required distances along the net (Fig. 23). See Dilks et al. (1995) and Kunz and Kurta (1988) for further details.

When shooting lines over branches, it can be helpful to use speargun rubber on a slingshot to reach a good height. Use braided fishing line (30 pounds) on a fishing reel attached to a c. 2-ounce sinker. The correct personal protective equipment (PPE) (helmet and safety goggles/visor) **must** be used by both the person shooting and the person holding the fishing reel. It takes practice to become good at shooting lines.

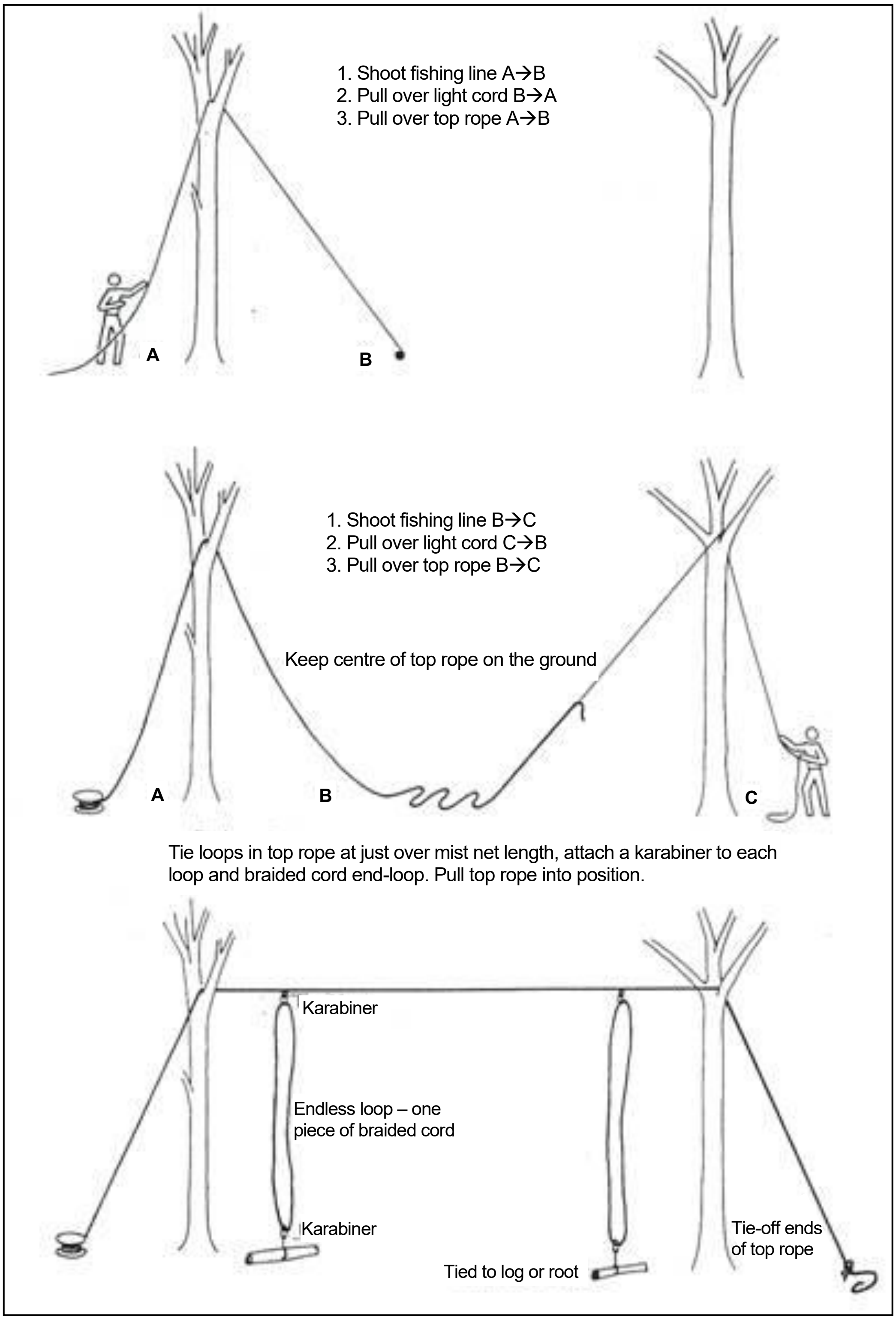


Fig. 23. Steps for erecting a canopy rig (Dilks et al. 1995)

If nets are to be stacked on top of each other, they need to be joined so that there are no gaps between them. Gaps can be prevented by overlapping the net attachment by one pocket. However, this will create a double layer of mesh, which will be easier for bats to detect and will also result in the bats becoming more tangled when caught.

Nets can be tied together with short lengths of sewing thread or string, wire bread-bag ties, or even grass (Fig. 24). 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 setup 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. 24).

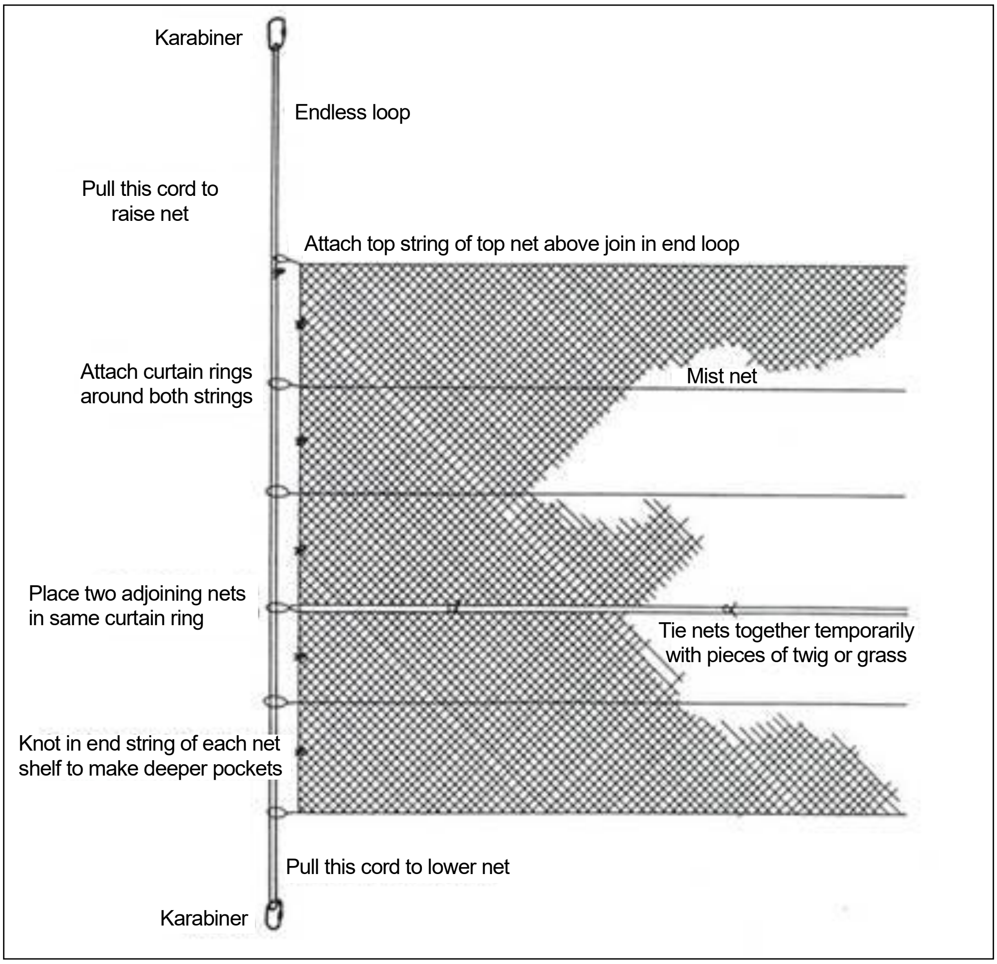


Fig. 24. Method for attaching nets to a cord pulley system (Dilks et al. 1995)

7.1.3 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 should 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 the net needs to be checked. 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 **must** be undertaken by a minimum of two people, and preferably three if using a rope pulley system where the net needs to be lowered. Two or more people are useful if more than one bat is caught in a net at a time.

Bats **must** be removed from nets as soon as possible to prevent unnecessary stress on the bats, to reduce entanglement and to prevent bats from chewing large holes in the net.

7.1.4 Extracting bats

Before handling any bats, you **must** be certified as competent in the relevant skills, or be under the supervision of an authorised trainer (refer to *Bat Handling Competencies* [see Appendix 1] and Handling, examining, measuring and releasing bats; also read the Health and safety section).

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, as the techniques are basically the same. However, 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, making it 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 to prevent its struggles from entangling it further or allowing it to escape. It may be necessary to lower the net to reach the bat. Do not try to lower the net by pulling down on the net close to the bat. Instead, lower the net from each end while avoiding lowering the bat so that it rests on other net pockets or on the ground, as this may cause it to become further entangled.

Lesser short-tailed bats can become aggressive when tangled in nets and may bite (sometimes drawing blood) when handled. Consequently, gloves should be worn when handling lesser short-tailed bats. However, it is almost impossible to remove bats from a fine mist net while wearing a pair of gloves so, as an alternative, one glove (or a bat bag) could be worn on the hand that is being used to restrain the bat and the other hand could be left bare to manipulate the net (Fig. 25).

Fig. 25. Using a bag to restrain a lesser short-tailed bat (*Mystacina tuberculata*) in a mist net. *Photo: CFJ O'Donnell*

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. First, 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 no netting is caught in the bat’s teeth. Six basic steps to remove a bat from a net are illustrated in Fig. 26, and two ‘real life’ examples of removing lesser short-tailed bats from nets are shown in Fig. 27. A short video of taking a long-tailed bat out of a mist net is also available (refer to *Extracting a New Zealand Long-tailed Bat from a Mist Net* [see Appendix 1]).

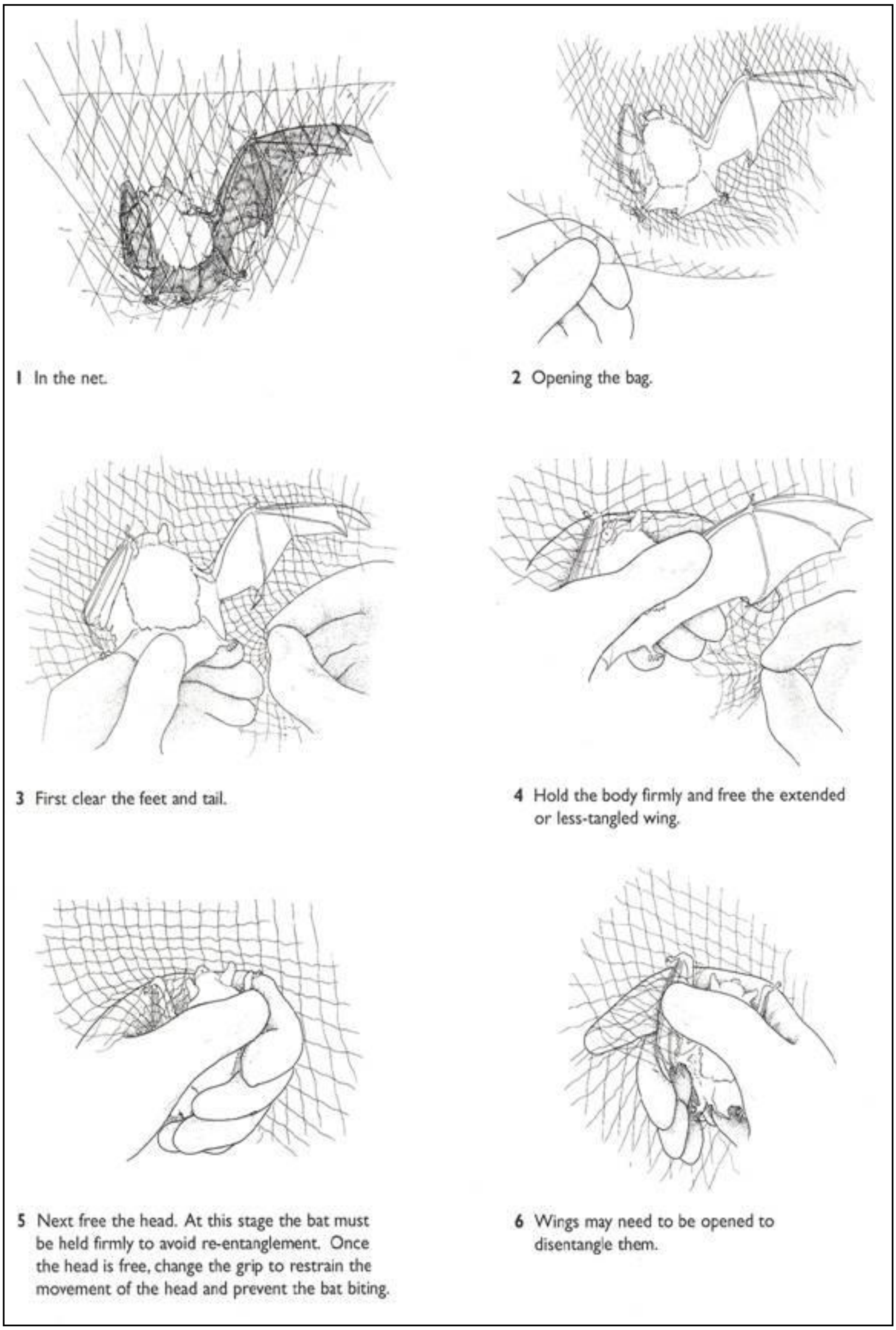


Fig. 26. Six basic steps to remove a bat from a mist net. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

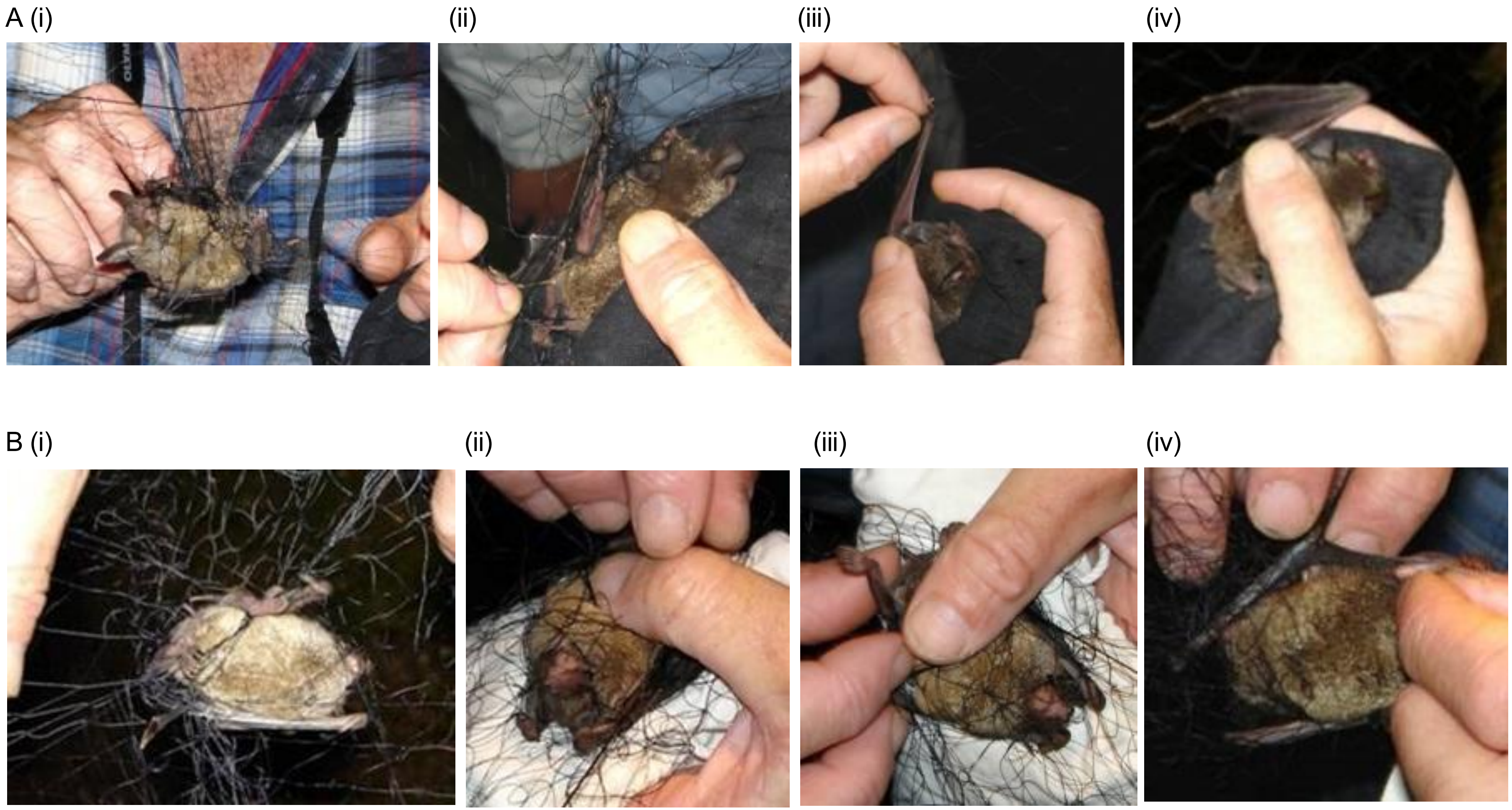


Fig. 27. Two examples of how to remove a lesser short-tailed bat (*Mystacina tuberculata*) from a mist net. (A) (i) In this sequence, the belly of the bat is tangled but its back is fairly free; (ii) the bat is secured in the bag and its feet are disentangled; (iii) its forearm is freed by gently easing the net over its wrist and thumb; and (iv) the wing is half opened 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; (iii) its feet are worked free; and (iv) the net is gently pulled over the bat's wings and head. *Photo: JA Sedgeley*

Occasionally, bats become so badly entangled that they cannot be freed quickly. In these situations, the net should be cut using fine scissors or a sewing ‘unpicker’ to prevent the bat from becoming 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 routinely be removed from nets, especially before nets are furled or folded away.

7.1.5 Capture rates for New Zealand bats

Long-tailed bats

Long-tailed bats often have very low capture rates in mist nets, even in areas of high activity, because they can easily detect and fly through small holes in nets. For example, in the Eglinton valley, capture rates of < 0.01 bats/net-hour have been recorded (O’Donnell and Sedgeley 1999), and the highest capture rates occur in February when young bats begin to fly for the first time, as these bats are relatively poor fliers and are less adept at avoiding nets.

Capture rates can be improved by setting nets close to foraging areas, such as ponds, and by using high mist net rigs that are 3–10 nets tall. Note that nets set for long-tailed bats do not need to touch the ground.

Lesser short-tailed bats

Lesser short-tailed bats can sometimes be caught within minutes of opening nets in areas of reasonably high bat activity. These 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 their time foraging on the ground, and sometimes fly at heights of less than 1 m, so 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 for this species (Fig. 28). Note that if the net is close to or touching the ground, remove bats entangled at ground level before commencing work on any other bats in the net.

Fig. 28. Lesser short-tailed bat (*Mystacina tuberculata*) caught in a mist net at ground level. *Photo: CFJ O'Donnell*

7.2 Harp traps

7.2.1 Description

Harp traps are specialised traps that were developed in America and Australia specifically for catching bats (Constantine 1958; Tuttle 1974; Tidemann and Woodside 1978). These traps typically consist of a 2 m × 2 m square frame of metal tubing that supports two banks of vertically strung monofilament fishing line and has a collecting bag attached beneath it (Fig. 29). The trap works on the principle that the banks of fine lines confuse the echolocation calls of bats, so while bats may be able to fly through the first set of lines, they find it difficult to avoid the second set, causing them to slide down the lines and land in the bag. The lines are tensioned so that bats do not become entangled, and the collecting bags are usually lined with polythene/plastic, which is attached at the top and extends downwards for about half to three-quarters of the height of the bag. The polythene is slippery and prevents any captured bats from climbing out, but allows them to crawl up the bag beneath it (Fig. 30). 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 the guidelines found in Tidemann and Woodside (1978), but commercially produced harp traps are also available from Titley Scientific.

Fig. 29. Harp trap

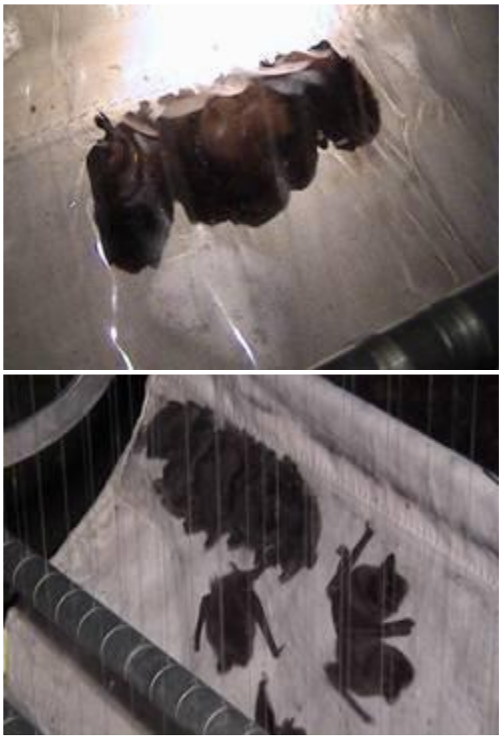


Fig. 30. Bats in harp trap bags

Photo: CFJ O'Donnell

Advantages

- Harp traps are reasonably portable over short distances and are easy to set up.
- Harp traps 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, often roosting 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.
- Harp traps 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.
- Lesser short-tailed bats have been observed climbing out of the trap bags.

7.2.2 Setting up harp traps

Assembly

Harp traps can be easily assembled by two people in a few minutes, or by 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 came with a very detailed information manual that includes instructions for assembly and maintenance, so if you borrow a trap, try to borrow a manual as well.

Once the trap is assembled, it is important to ensure that the strings are tensioned correctly – if the lines are too loose, any captured bats might get tangled, and if they are too tight, the bats might bounce off. Therefore, the trap should be adjusted so that the fishing lines are firm when pressed with an open hand, and any broken lines should be removed and replaced. The collecting bag also needs to be adjusted by hanging it on the trap and tying the end ties firmly around the hip-mounts or the legs of the trap to form a narrow V-shape. The aim is to prevent the bag from being loose and saggy, which will allow bats to escape, while avoiding tying it off too tightly, which will reduce the angle of the capture zone and could prevent bats from sliding down into the bag.

The commercial traps come with four telescoping legs, allowing them to 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. 31). Ropes can be attached for hoisting and lowering the traps.

Fig. 31. Harp trap suspended outside long-tailed bat (*Chalinolobus tuberculatus*) roosts in a tree and in a rock crevice. *Photo: CFJ O'Donnell*

A bridle or bracket should be attached to the top of the harp trap, and the centrally spaced lifting rope should then be attached to the bridle (Fig. 32 and Fig. 33). (Note: Avoid using the rope attachment method originally illustrated in Sedgeley and O'Donnell [1996] because it may warp the frame and affect the line tensioning.) It is also necessary to secure the top and bottom of the trap to prevent it from 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. 34). Guy ropes should then be attached to the trap at the four corners of the frame to aid in positioning the trap once it is in the air, to secure the trap in its final position and to assist in the lowering process.

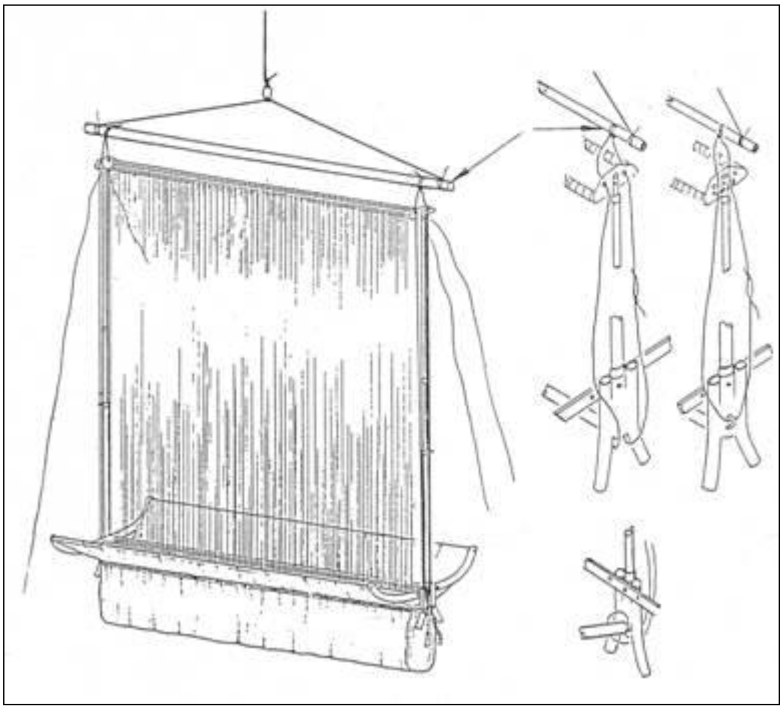


Fig. 32. Attaching the bridle to suspend a harp trap. Source: Faunatech harp trap instruction manual.

A

B

Fig. 33. Bracket for suspending a harp trap. *Photos: (A) W Simpson; (B) J Sedgeley*

Fig. 34. Securing the sides of a harp trap. *Photo: JA Sedgeley*

7.2.3 Monitoring traps

Harp traps do not generally require continuous monitoring because any bats that hit the fishing line will fall into and be held by the collecting bag. Consequently, harp traps positioned on the ground can be set up at dusk and checked at dawn, provided that the bats are released before it is too light. However, it is preferable to check the traps several times during the night so that the bats are not held for unnecessarily long periods of time. There have also been records of bats in harp traps being preyed upon by mammals and snakes in Australia. Predation at traps left unattended for long periods has not been recorded in New Zealand but is a potential problem as both possums and ruru/morepork have been observed visiting traps. Harp traps **must not** be set during rain as bats quickly become wet when caught in them.

During the breeding season, traps **must** be checked at least twice each night to enable captured lactating females to be released and 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, and the young of both species begin to fly at 5–6 weeks old (Lloyd 2001; O'Donnell 2001). Harp traps placed outside or near roosts **must** be monitored continuously from dusk onwards.

When trapping at roost entrances, the traps **must** be monitored continuously to estimate the number of bats captured. It is important that this number does not exceed the number that can be processed (banded, measured, PIT tagged, etc.) within 2 hours of capture by the team available to process the bats.

7.2.4 Extracting bats

Before handling any bats, you **must** be certified as competent in the relevant skills, or be under the supervision of an authorised trainer (refer to *Bat Handling Competencies* [see Appendix 1] and Handling, examining, measuring and releasing bats, as well as the Health and safety section).

After falling into the collecting bag, there is usually an initial period where the bats flap or run around the bottom of the bag. However, eventually, the bats tend to crawl under the plastic, move upwards and settle down to roost at the top of the bag – and if several bats are present, they will often form roosting clusters. This allows the bats to be easily 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 conditions are fairly cold, the bats will go into torpor. This usually means that the bats will simply be very sluggish and slow moving when they are handled. However, long-tailed bats that are coming out of torpor have very occasionally 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 bat's wings to its sides to enable it to be transferred into a holding bag.

7.2.5 Capture rates for New Zealand bats

Several studies using infrared cameras have shown that bats can detect and avoid harp traps, and that this ability varies among different species (reviewed in Gration 2002). However, no studies to date have compared the differences in capture rates between long-tailed bats and lesser short-tailed bats.

Long-tailed bats

Long-tailed bats have been caught successfully in harp traps in a range of locations and situations.

Lesser short-tailed bats

Free-standing harp traps appear to be less successful in catching lesser short-tailed bats than long-tailed bats, even when set in areas of high activity. For example, traps set in roosting areas have never caught a bat on Whenua Hou / Codfish Island and have only caught bats on a small number of occasions in the Eglinton Valley. Lesser short-tailed bats are not necessarily better at avoiding these traps, but rather may be better at escaping from them, as they are able to launch themselves into flight or climb out of the collecting bags – indeed, several individuals were observed climbing out of a trap set up outside a roost tree on Whenua Hou / Codfish Island (J Sedgeley, DOC, pers. obs.)

7.3 Other trapping methods

The methods described below have not been trialled in New Zealand but are proven techniques for bat species elsewhere. Trialling these methods in this country would require ethics approval.

7.3.1 Hand nets

Simple hand nets of the type that can be obtained from entomological suppliers (i.e. butterfly or dragonfly nets) are commonly used to capture bats at roost sites in other countries (Kunz and Kurta 1988; Churchill 1998; Finnemore and 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 and 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, hand nets could be used to catch bats in caves if they are roosting within reach, or 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).

If the bat is within reach, the net can be carefully placed over the bat before it flies. Alternatively, in places where bats are roosting high off 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 and Kurta 1988; Finnemore and Richardson 2004). As soon as a bat falls into the net, the frame should be rotated so that it cannot fly out again.

Hand nets should not be used to catch bats in free flight, as striking a bat with the hoop of the net can easily break bones and cause other injuries.

7.3.2 Cone and bag traps

Cone and bag 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. These 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 and Richardson 2004).

7.3.3 Trip lines

Trip lines are commonly used in Australia as a cheap and simple method for catching bats over water. Monofilament fishing line (c. 3 kg breaking strain) is criss-crossed across a water body multiple times at heights of 10–15 cm (Churchill 1998) or 6 cm (Reardon and 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 so swim to the edge before taking off in flight, allowing a person waiting at the edge to pluck the bats out of the water by hand or with a hand net. Note, however, that bats can swim surprisingly quickly.

7.4 Maximising capture rates in foraging areas and flight paths

Bats can frequently detect and avoid both mist nets and harp traps, but there are several ways of increasing the probability of catching them.

7.4.1 Trap placement

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 – for instance, nets and traps set across small pools and streams amongst trees where long-tailed bats frequently foraged proved to be effective in Pureora, South Canterbury and the Eglinton valley (Fig. 35). Suitable areas can be identified by using ABM 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, so nets and harp traps set across these gaps may be effective – and more than one harp trap can be used to fill a gap.

Care should be taken when trapping over water. Traps should be set close to the water to catch bats that are 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. Note that nets will sag over the course of a night and so may need to be readjusted – and it is also important to be aware of rising water levels.

There is some thought that bats can come to learn the position of traps, resulting in capture rates declining once the element of 'surprise' has been lost. Therefore, we recommend that traps are moved around and locations are given 'rest' nights.

Camouflage and confusion

Mist nets placed in front of vegetation are less likely to be detected by bats. However, it is also important to consider that if the 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 that vegetation 'funnels' the bat towards the trap/net (e.g. at the end of an enclosed track, beneath a large branch overhanging water or between a clump of trees and the bush edge). Harp traps work well when placed in small gaps in the vegetation, and any spaces around the traps can be filled with branches, vegetation or even shade cloth material (Fig. 35A).

A

B

Fig. 35. Harp traps set in long-tailed bat (*Chalinolobus tuberculatus*) foraging areas. (A) Pond used by foraging long-tailed bats in South Canterbury. (B) Old forest track in the Eglinton valley. *Photos: J Sedgeley*

The 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 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 also be less obvious to a bat than one placed at right angles (Reardon and Flavel 1987; Kunz and Kurta 1988; Churchill 1998).

7.4.2 Lures

It has been suggested that a bat can be lured into a net by flicking a small pebble upwards into 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. However, we know of no examples of this working in New Zealand.

Several researchers in this country have used Audubon bird squeakers to lure lesser short-tailed bats into nets, based on the theory that the squeaker produces a sound that imitates lesser short-tailed bat song. The use of squeakers may be more effective during the summer/autumn breeding season when singing activity peaks, but the effectiveness of this technique, and whether it is biased towards males or females, remains untested. The technique appeared to attract bats of both sexes into mist nets during the summer months in the Eglinton valley, with the most effective method being 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 were not successful at luring long-tailed bats, however, and generally scared these bats away (CFJ O'Donnell, pers obs, 1997).

Acoustic lures, which are devices that play back recorded or synthesised bat calls, have now been used to increase capture rates of different bat species in numerous countries (e.g. Hill and Greenaway 2005; Colins 2016; Samoray et al. 2018), and a range of lures are now available commercially. The Sussex Autobat lure has recently been used in New Zealand to increase the capture rates of both short- and long-tailed bats (I Davidson-Watts and CFJ O'Donnell, unpubl. data). However, the effect of acoustic lures on New Zealand bats is still poorly understood, so they **must** be used with caution and only when all other methods have been considered. Lures and/or lure speakers should be placed very close to the harp trap or net for them to be effective (e.g. see Fig. 36). Lures **must not** be played at high volume and there should be periods of silence when using lures so that they are not broadcasting continuously. Lures **must** also be turned off during the extraction of bats and traps **must** be checked regularly (between 1 and 4 hours maximum). Additionally, distress calls **must not** be played (unless part of a specific experiment / research objective) and lures **must not** be used within 80 m of known active communal roosts (based on Peterson detectors picking up an acoustic lure call at 80 m distance and the assumption that bats have better hearing than the detector). Research is ongoing to determine the most effective bat calls to use as lures in the New Zealand context. It should also be noted that lures may deter bats and could be counterproductive if used incorrectly, so getting some experience and training is advised.

Fig. 36. An acoustic lure setup at a harp trap. *Photo: CFJ O'Donnell*

7.5 Trapping at roost sites

Some research studies and management projects require large numbers of bats to be caught – 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 creates a much higher level of disturbance to bats than trapping in foraging areas, and can potentially cause bats to abandon their roosts. Several studies and management projects have involved catching long-tailed bats and lesser short-tailed bats as they emerge from their roosts (e.g. Sedgeley and O'Donnell 1996; Sedgeley and Anderson 2000; Lloyd and McQueen 2002; O'Donnell 2002b), and none of these studies recorded bats abandoning their roosts. However, this does not mean that the bats were not affected in other, less observable ways. Consequently, trapping at roost sites **must** only be undertaken if there is valid reason for doing so.

7.5.1 Mist netting at roosts

Note: When catching bats at a roost tree that is known to be occupied, it is preferable to use a harp trap rather than a mist net, as it has fewer associated risks.

Often when mist netting, the location and distance from occupied roost trees will be unknown. If nets are set close to a roost tree, there is the potential to catch many bats in a very short period (lesser short-tailed bat roosts may be occupied by thousands of bats) – and lesser short-tailed bats are often still caught even when the main roosts are more than 1 km away (e.g. if the nets are placed on a flyway).

it is extremely difficult to quickly extract large numbers of bats from the same net, as the bats can become very tangled and their removal can be very time consuming. Bats that remain in a net for a long period become distressed and can chew the nets, and will also sometimes bite themselves and other bats. Additionally, the sounds of bats squeaking in the net will draw in others.

If the catch is focused on processing large numbers of bats at once, you **must** ensure that there are enough handlers present to process the bats quickly and carefully. If a net is inadvertently placed in a position where too many bats are being caught, a soft cover such as a sheet or tent fly could be thrown over the net to prevent further captures – note that 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).

A large mist net rig (10 nets high) has previously been used in front of the main entrance of a long-tailed bat roost in Grand Canyon Cave (O'Donnell 2002b). The mist nets did not completely block off the cave entrance and could be quickly lowered by a pulley system. However, the bats here easily detected and avoided the nets, resulting in very low capture rates.

7.5.2 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. 37), and a line of four standing traps were used to cover the smaller southern entrance (Fig. 38). Despite their much smaller capture area, the harp traps in the northern entrance captured many more bats than the large mist net rig used there (O'Donnell 2002b).

A B

Fig. 37. Two traps suspended in the main (north) entrance of Grand Canyon Cave viewed from (A) inside and (B) outside the cave. *Photos: J Sedgeley*

Fig. 38. Four harp traps set at the rear (south) entrance to Grand Canyon Cave. *Photos: J Sedgeley*

Tree roosts

Using harp traps directly outside tree roosts can be very effective with correct positioning. Harp traps have been used to catch up to 100% of the long-tailed bats emerging from trees (including maternity roosts) in the Eglinton valley and did not appear to affect their roosting behaviour or cause them to abandon their young (Sedgeley and O'Donnell 1996). Harp traps have also been used successfully outside lesser short-tailed bat roosts in Rangataua Forest, in the Eglinton valley and on Codifish Island / Whenua Hou (Sedgeley and Anderson 2000; Lloyd and McQueen 2002).

There is no maximum number of bats that can be caught in a harp trap, but thought should be given to how long the bats are going to be held in the trap, how many bats can be practically processed in the timeframe and how many bats could potentially be caught. Large numbers of bats inside a collecting bag at one time may be unduly stressful for the bats and lesser short-tailed bat roosts may contain thousands of bats, so particular care **must** be taken to continuously monitor the harp trap and lower it quickly when needed when catching these bats outside roost sites. Mounting a small infrared video camera on the trap bag is useful for counting how many bats fall into the trap, particularly when catching lesser short-tailed bats, so that the trap can be lowered when the target number has been caught. A thermal camera can also be used from the ground to count bats being caught in a trap. In Rangataua Forest, a specialised bag was used to allow the ongoing removal of bats and continuous trapping (Lloyd and McQueen 2002; Fig. 39).

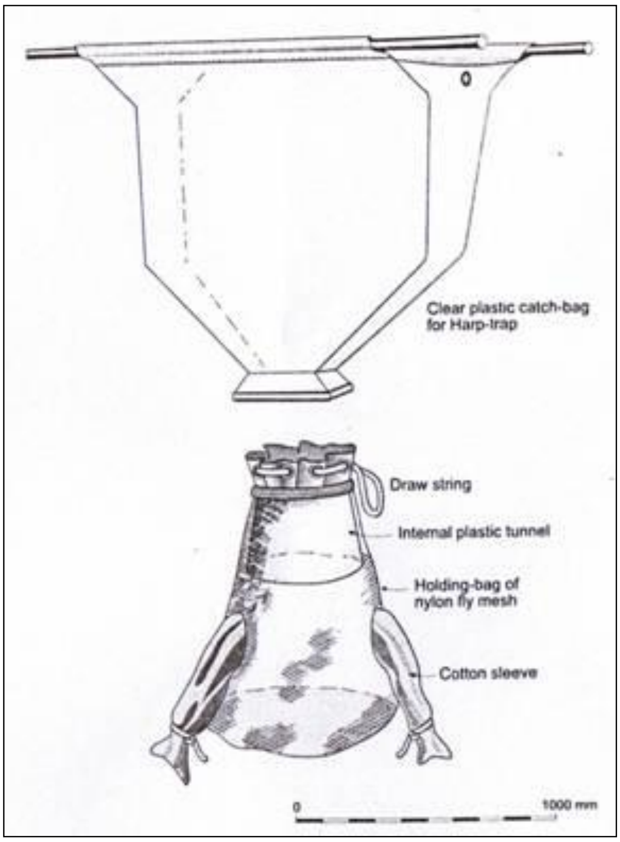


Fig. 39. Specialised harp trap bag used to allow the ongoing removal of bats during continuous trapping outside roost trees. Reproduced with permission from Lloyd and McQueen (2002).

The optimum trap placement for catching long-tailed bats exiting a roost appears to be hung across the roost entrance with the edge of the collecting bag as close to the exit hole as possible, often leaning against the tree (Fig. 40A), as this often results in bats falling 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 proved effective at catching lesser short-tailed bats as they returned to their roost site after foraging (Lloyd and McQueen 2002). However, the position of tree branches in relation to the roost hole can sometimes make it difficult to fit in a standard-sized harp trap. To address this, standard-sized commercial traps can be made smaller by shortening the sides of the trap and rolling up the line carriers. It is then necessary to secure the line carriers in their new position using insulating tape and to carefully re-tension the fishing lines and push them back into position (Sedgeley and O'Donnell 1996). Alternatively, a small-sized ('mini') harp trap can be purchased for trapping at roosts (Fig. 40B).

A B

Fig. 40. (A) Full-sized harp trap placed outside a long-tailed bat (*Chalinolobus tuberculatus*) roost; the arrow indicates the bottom of the roost entrance. (B) Mini harp trap placed outside a long-tailed bat roost with nearby branches; the view of the roost entrance is obscured by the trap bag. *Photos: (A) J Sedgeley; (B) GC Dennis*

7.6 When to set nets and traps

7.6.1 Time of day

Nets should be open by dusk, but it is important not to open them too early to avoid capturing birds. Long-tailed bats emerge from their roosts on average 2–30 minutes after sunset, but can emerge as early as 58 minutes before sunset, while 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 the night. Nets do not have to be dismantled during the daytime, but they **must** be furled to prevent the 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 from spinning loose. If several nets are stacked, the nets need to be lowered and all nets gathered together before spinning (Kunz and Kurta 1988; Dilks et al. 1995). Note that if nets are left set up for several days, they will begin to sag and will often require readjusting.

Harp traps can be set at any time and do not need to be dismantled during the 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. However, collecting bags **must** be removed when traps are not in use. If harp traps are left set for several days and the collecting bags become overly damp or wet, it is good practice to bring in the collecting bags and dry them out.

7.6.2 Weather conditions

We do not recommend that mist netting is 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 and so are not usually affected by wind, and the polythene liners in the harp trap collecting bag will protect bats to some degree from drizzle or very light rain. However, it is not good practice to leave bats inside cold and wet bags because they may enter torpor and subsequently become difficult to release.

7.6.3 Seasonality

Bat activity is usually reduced during cold conditions and 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 2000c). However, activity levels in relation to temperature do vary throughout New Zealand, with lesser short-tailed bats having very low activity levels in winter in the Eglinton valley but being active to temperatures as low as −2°C on Codfish Island / Whenua Hou (Daniel 1990; Sedgeley 2001b), where 399 bats were caught in a very short period of time during winter using a combination of harp trapping and mist netting (Sedgeley and Anderson 2000).

Trapping at roosts during the period when females are pregnant and before pups are flying has increased risks that should be taken into consideration before trapping is undertaken, as trapping may interfere with the ability of females to regularly feed their young and there is an increased risk of females abandoning their young. Long-tailed bats are visibly pregnant from early to late November and give birth from mid-November to mid-December, while lesser short-tailed bats give birth sometime between mid-December and mid-January, although the timing of birth for both species is variable; young of both species begin to fly at 5–6 weeks old (Lloyd 2001; O'Donnell 2001).

7.7 General care and maintenance of nets and traps

This section discusses general maintenance of the nets and traps. It should be read alongside the Animal health considerations section.

7.7.1 Mist nets

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

7.7.2 Harp traps

Harp trap bags should be thoroughly cleaned of twigs, vegetation, bat droppings and insects. It is also important to undertake regular repairs and maintenance so that the traps can be set up quickly and easily, as well-maintained traps will improve capture rates and reduce the possibility that bats will become entangled. Harp trap lines can snap or lose tensioning and parts of the frame and legs can corrode. The Faunatech harp trap manual provides detailed information on re-stringing, maintenance and repair. Harp trap catch bags **must** be disinfected (e.g. with Sterigene) if used in different study areas and should be carefully dried before storage.

7.8 Temporarily holding captured bats

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

7.8.1 Bag design

The most common devices for temporarily holding bats are soft cloth (cotton) bags with either drawstrings or ties. Bags that are used for holding birds are fine to use, but **must** be washed (e.g. with Sterigene) before and after being used for bats, and catch bags and gloves **must** also be disinfected before being used with different bat populations. Strands from fraying material can become tangled around bats' claws and teeth, so unless the bags have perfectly sewn seams with no frayed edges, turn them inside out so that the seams are on the outside. Bags made from a light-coloured material are best because it will be 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, so drawstrings or ties should be long enough to wrap around the bag at least once to secure the opening tightly, and should also be long enough to hang the bag up.

7.8.2 Holding captured bats

It may not always be possible to process bats immediately after they have been captured (weighing, measuring, attaching transmitters, etc.). Bags containing bats **must never** be put on the ground where they might be accidentally trod on or sat on – ideally, they should always be hung up in an allocated place. Bats **must not** be held in cloth bags for more than 2 hours, and it is preferable to release them as soon as possible.

A small number of bats (no more than five) 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. These species have never been recorded roosting together under natural conditions and differ in size and temperament, so there may be animal health implications resulting from holding them together.

Bats caught carrying young **must not** be held in bags but rather should be released immediately without processing. If young are separated from their mothers in the trap, they should be placed on a log or tree nearby to enable the females to collect them. The adults should then be processed as quickly as possible so that the females can be released to collect and feed their young before they get too cold.

Heavily pregnant bats should be handled as little as possible and, if possible, should not be fitted with transmitters or transponders. If they are to be banded or tagged, this should be done quickly, with no measuring or other procedures being undertaken to speed up processing times.

7.9 Animal health considerations

Little is known about potential disease risks in bats. However, when catching bats, common sense should be used and the following general principles apply.

- Disinfect or wash nets and harp trap bags between study areas to minimise the possibility of transferring parasites and diseases between sites.
- If possible, use different nets for birds and bats.
- Regularly empty bags of twigs, leaves, droppings, etc.
- Wash catch bags and gloves with Virkon®, Sterigene or F10 when moving between different sites.
- **Do not** use bags that have been used for holding birds unless washed first.

See the Health and safety section for more guidance. Also refer to the *Wildlife Health Management SOP* (see Appendix 1) for further information on disease surveillance and hygiene precautions.

7.9.1 SARS-Cov-2 (Human–bat transmission)

The IUCN bat specialist group recognises a low but credible risk of human-to-bat transmission of SARS-CoV-2. Therefore, all fieldwork involving catching bats **must** include an assessment of the level of risk that the project poses to bats and modify the fieldwork activities based on that risk assessment if necessary. See the risk assessment method recommended by the IUCN bat specialist group in July 2021 and check for up-to-date guidance from this group and on the Protecting bats/pekapeka page on the DOC website.

7.10 Additional equipment

A head torch is essential for taking bats out of nets and bags, and a spotlight or floodlight is useful for checking nets and essential for tall mist net rigs. Also, always have available some sharp fine scissors or an 'unpicker' when mist netting, and cloth bags to put captured bats into.

8. Handling, examining, measuring and releasing bats

When handling any live animal, it is important to consider how best to avoid stress and injury to both the animal and the handler. This section discusses how to safely handle and release bats. It also describes how to age, sex and assess the reproductive condition of individual bats, and the differences between species, as well as techniques for taking a range of morphological measurements and biological samples.

8.1 Handling bats

8.1.1 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 the bats and, if used carelessly, cause injury. Reasons for handling bats include:

- species identification
- marking
- research
- monitoring (e.g. outcome of predator control)
- rescuing injured and sick bats.[3]

8.1.2 Safety precautions and the use of gloves

Bats in other countries 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. There is also the *potential* risk of humans passing introduced viruses (e.g. SARS-CoV-2) on to native bats when handling them. See Animal health considerations.

Anyone planning to work on a project that will involve the routine handling of bats **must** read the Health and safety section 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 to the handler, a reflex withdrawal of the hand by the handler may harm the bat. Gloves should ideally 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 being used to control the bat, and keep the hand being used for manipulation purposes (e.g. for extracting bats from a mist net or

[3] A permit is not needed to help a sick or injured bat. However, bats **must** be handed in to a vet, 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 who are expert in such matters.

taking measurements) bare (Fig. 41). Depending on which 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. Where glove wearing is impractical (due to the difficulty of handling bats with gloved hands), hands **must** be disinfected/sanitised regularly.

A B

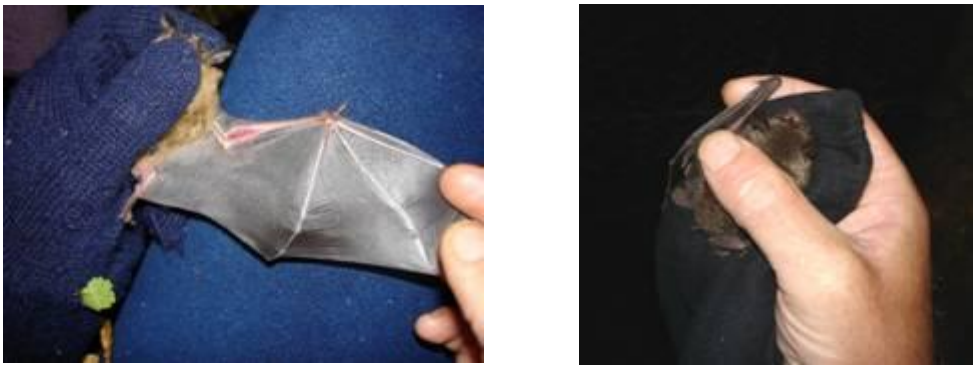


Fig. 41. When handling lesser short-tailed bats (*Mystacina tuberculata*), a glove should be worn on one hand or catch-bag folded around the bat to help protect the handler from bites, but the free hand can be left bare for manipulation. *Photos: (A) J Sedgeley; (B) CFJ O'Donnell*

Always wash your 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, liquid antiseptic or soap and water as soon after the bite or scratch as possible. Washing should consist of gently flushing the wound, not scrubbing it, as this could push particles further into the wound (see Health and safety). 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 always wear gloves when handling obviously sick bats or dead bats.**

8.1.3 Recommended grips

New Zealand bats are fairly small and fragile, particularly the finger bones in their wings – and both species can be very wriggly in the hand and can bite. Only people who have been certified as handlers by the Bat Recovery Group, or are being directly supervised by an authorised handler, may handle bats. Information on the bat competencies framework is available on the Protecting bats/pekapeka page on the DOC website.

It is important to develop a technique to hold a bat safely both to prevent injury to the bat and to avoid being bitten. It is generally easiest for workers to hold the bat in their non-dominant hand and take measurements with their dominant hand. We generally recommend that bats are 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 (Fig. 42 and Fig. 43). The bat may be held with the head protruding between the thumb and forefinger, which can be used to keep the bat's jaw shut (Racey 2004; Fig. 42). This method appears to minimise stress caused to the bat, and this grip is probably the best one to use when measuring the forearms and for opening the wings (Fig. 41 and Fig. 44) – but take care not to apply excessive pressure to the neck or back.

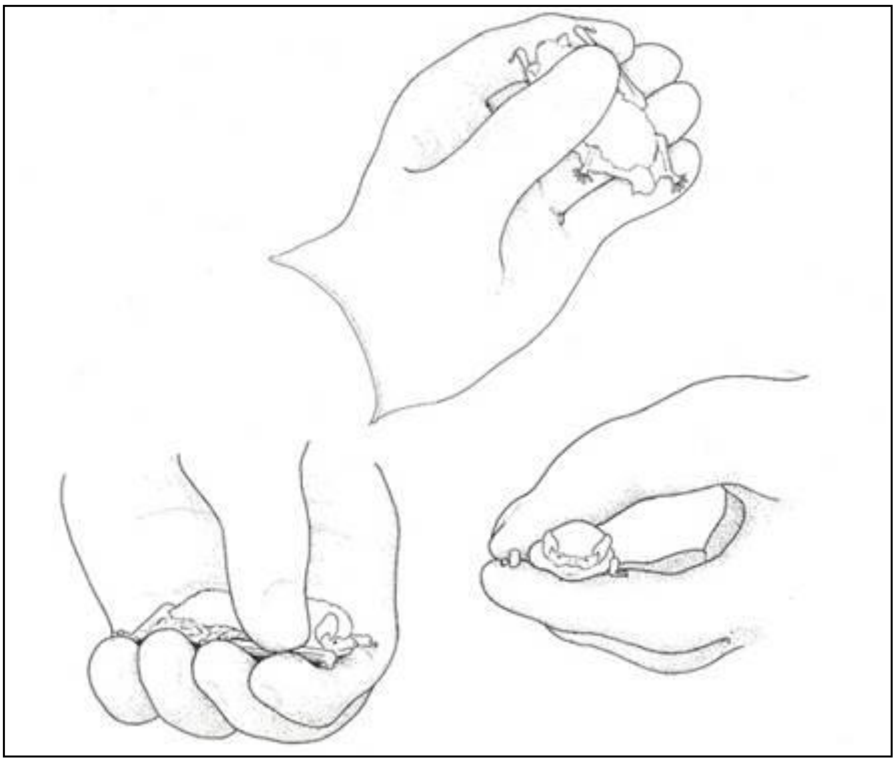


Fig. 42. Palm grip recommended for general handling and measuring purposes. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

Fig. 43. Using the palm grip on New Zealand bats. *Photos: J Sedgeley*

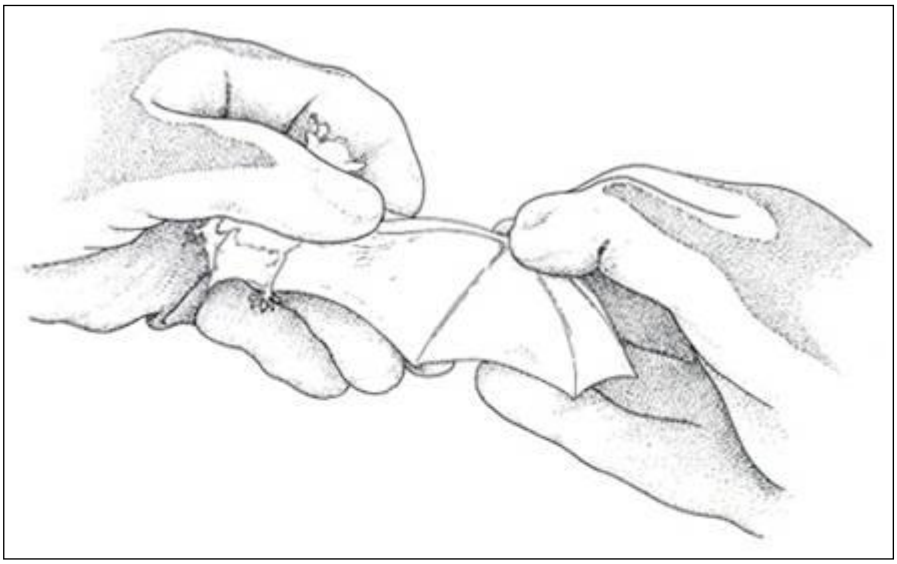


Fig. 44. Using the palm grip to examine a wing. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

An alternative grip that is commonly used in other countries for restraining larger bat species and is useful for holding a bat still for close examination is shown in Fig. 45. However, this grip appears to be more stressful for the bat than the palm grip and so should only be used for specific purposes. Care **must** be taken not to strain the forearms and flight muscles.

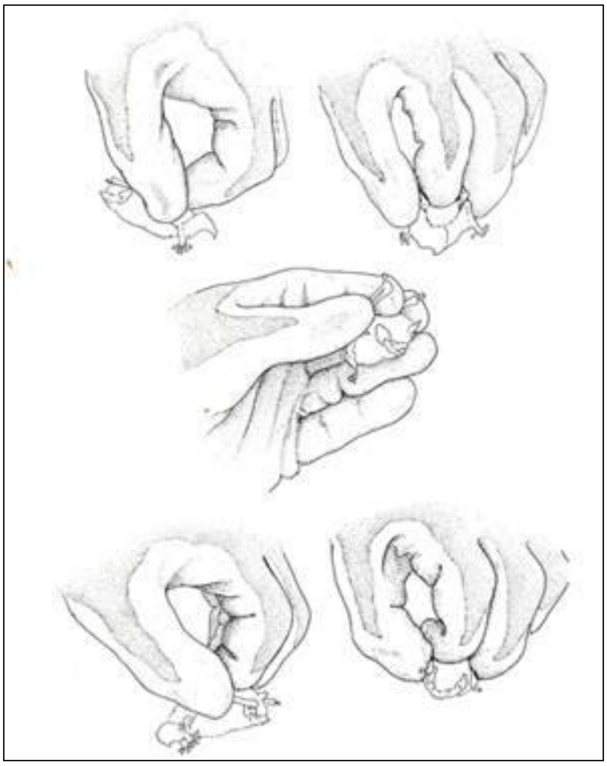


Fig. 45. Variations of an alternative grip that is recommended for holding a bat still for close examination. Care **must** be taken not to strain the forearms and flight muscles. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

8.1.4 Handling time

Bats **must** be processed (identified, measured and/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), but the time from initial capture through to release should be no more than 2 hours. The handling duration **must** be kept to a minimum while taking measurements, as 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**.**

We accept that people will want to take photographs while the bat is in the hand. Consider the impact of taking the photograph in each case, keeping in mind how long the bat has been in the bag and/or hand, whether it has shown signs of stress during handling, and anything else of concern that may warrant releasing the bat and not holding it back for a photograph. Where possible, do not use a flash – rely on torchlight only.

8.2 What should be measured and why

Measurements are usually taken 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 bat and long-tailed bat), as 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 (see Species identification in the hand).

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 46 shows the main features of a bat and the terms used to describe them, many of which are used in the sections below.

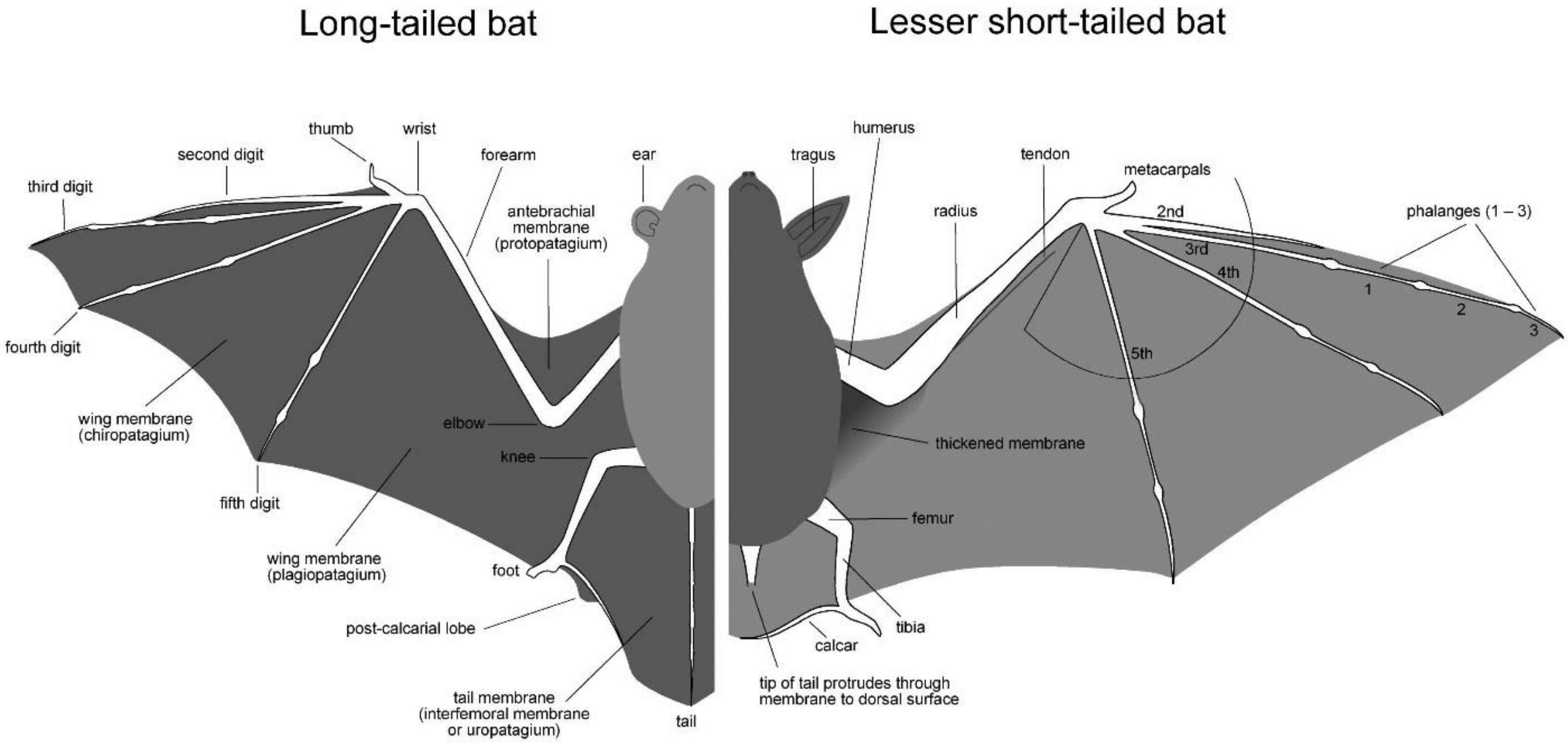


Fig. 46. The main features of a bat highlighting the key visual differences between lesser short-tailed bats (*Mystacina tuberculata*) and long-tailed bats (*Chalinolobus tuberculatus*)

8.3 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 that can also be examined, 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 at first flight; the term 'juvenile' is used to describe bats from the age at 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 yet reached breeding condition / sexual maturity; and the term 'adult' is used for bats that have reached sexual maturity.

8.3.1 Wing joints

At the time of first flight, a bat's 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 appear as pale bands either side of the joint, but as the cartilage is replaced by bone, the joint becomes more rounded and knuckle-like (Hutson and Racey 2004; Fig. 47). The joints usually appear fully ossified by the autumn, which means that this technique can only be used to reliably distinguish juveniles from sub-adults and adults for up to about 12 weeks after birth.

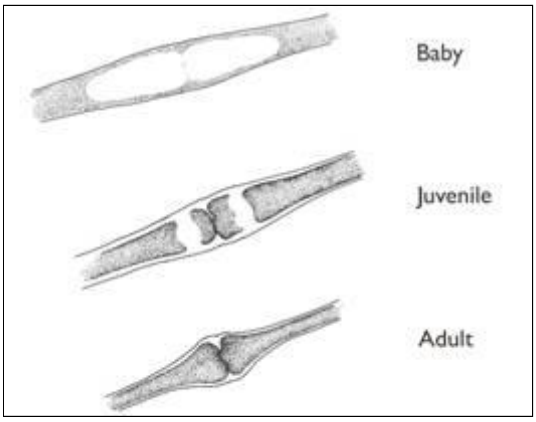


Fig. 47. Development of the joints of the finger bones, which can be used to reliably age bats. The joints of baby bats and juveniles appear thin and tapered with paler bands, while the joints of adults are more knobbly. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

The easiest way to examine the finger joints is to illuminate the wing from behind by holding it over a torch (Fig. 48 and Fig. 49). A torch with a reasonable diameter head is best. It is important to be aware of how hot the torch gets – for example, a spotlight could burn the bat's wing.

Fig. 48. Technique for examining the finger joints in long-tailed bats (*Chalinolobus tuberculatus*) illustrating the differences between (A) an adult and (B) a juvenile. *Photos: CFJ O'Donnell*

Fig. 49. Technique for examining the finger joints in lesser short-tailed bats (*Mystacina tuberculata*) illustrating the differences between (A) an adult and (B) a juvenile. *Photos: J Sedgeley*

8.3.2 Other features

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

8.3.3 When to look for juveniles

Young bats begin to fly at 5–6 weeks old. Young long-tailed bats have been recorded flying in the second week of December in South Canterbury and on 6 January in Hawke's Bay, while flying juveniles are caught from mid-January onwards in the Eglinton valley, although the date of first flight can vary by as much as 17 days annually (O'Donnell 2002, 2005). By contrast, lesser short-tailed bats give birth sometime between mid-December and mid-January throughout New Zealand (Lloyd 2005b), so flying young are unlikely to be caught until January or February – for example, at Pureora Forest, juveniles are normally flying by the last week of January.

8.4 Sexing bats and assessing their reproductive condition

8.4.1 Sexing

Male bats have a conspicuous penis (Fig. 50). Female lesser short-tailed bats have a pronounced clitoral pad above the vagina, but it is smaller and more domed than a penis (Fig. 51). 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 but are much less obvious once they begin to recede after lactation and in females that have never given birth. The easiest way to examine the nipples of a female bat is to hold the bat on its back, hold the wing open and gently part its fur where you would expect the nipples to be (Fig. 52 and Fig. 53).

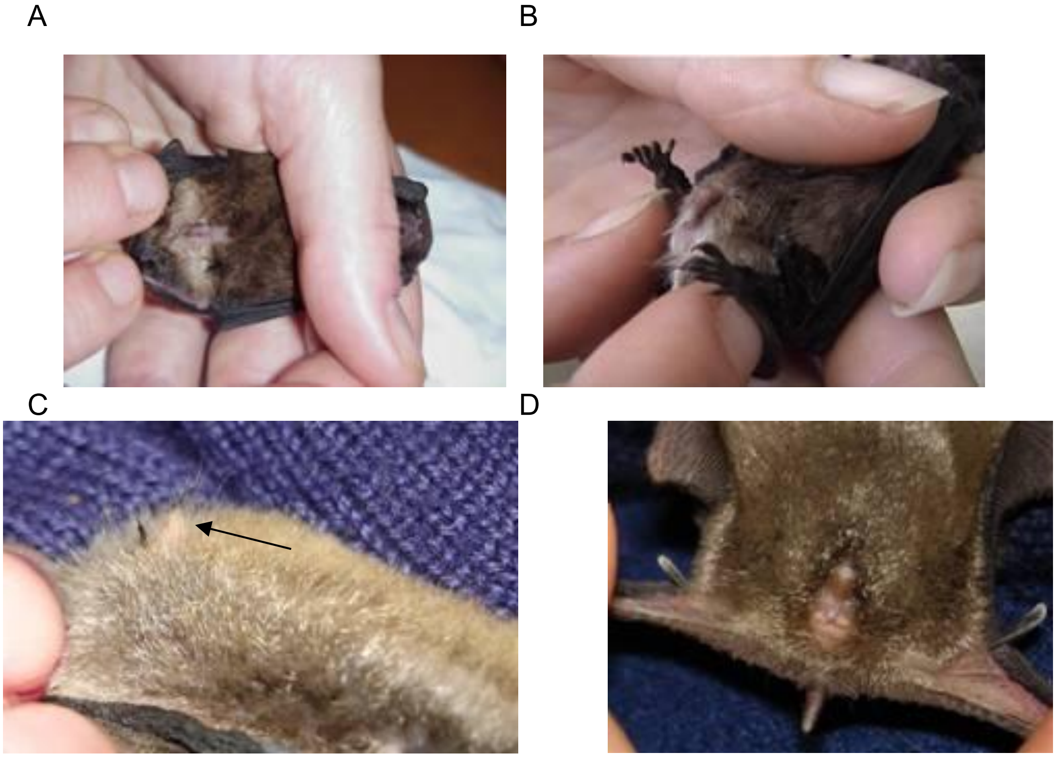


Fig. 50. Genitalia of male (A–B) long-tailed bats (*Chalinolobus tuberculatus*) and (C–D) lesser short-tailed bats (*Mystacina tuberculata*). *Photos: CFJ O'Donnell*

A B

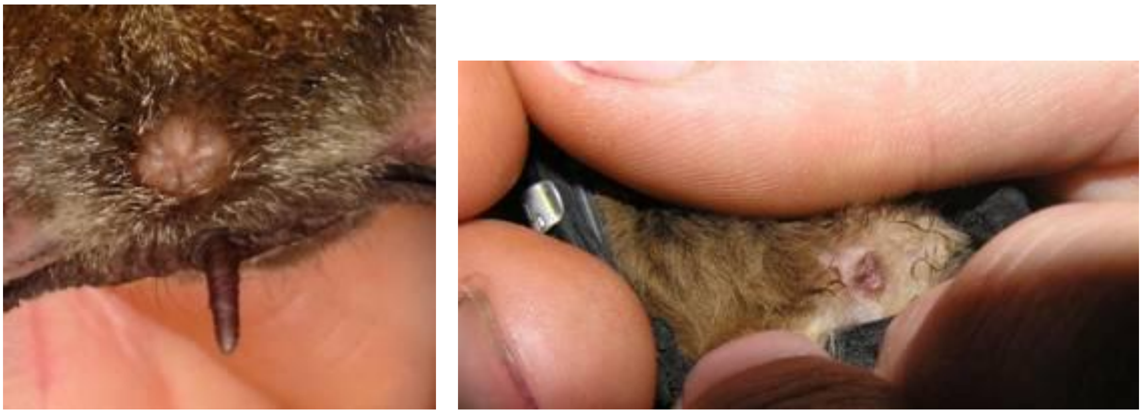


Fig. 51. Genitalia of a female (A) lesser short-tailed bat (*Mystacina tuberculata*), showing the clitoral pad, and (B) long-tailed bat (*Chalinolobus tuberculatus*). *Photos: (A) J Sedgeley; (B) CFJ O'Donnell*

8.4.2 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.

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. 52). In the Eglinton valley, nulliparous female long-tailed bats are usually 1–2 years old.

Fig. 52. Nipples of female long-tailed bats (*Chalinolobus tuberculatus*) that have never given birth (nulliparous females). *Photos: CFJ O'Donnell*

Parous females

Females that have given birth are termed parous and can be non-breeding or breeding. 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, but pregnancies that are half to two-thirds progressed can be diagnosed through gentle palpation of the abdomen. During pregnancy, the lower abdomen becomes very distended and it is possible to feel the single baby lying transversely by applying slight lateral pressure with two fingers, although 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 and 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. 53). Milk can sometimes be extruded from the nipple by applying gentle finger pressure to its base. When the young is being weaned, the nipple begins to shrivel and becomes darker and the hair gradually grows back around the nipple, although it always remains visible after giving birth once. It is very hard to say when the female has transitioned to post-lactating just by looking at the nipple, however. After the breeding season, the nipples regress markedly. In long-tailed bats, the nipples of parous females retain a larger and often darker appearance than the nipples of females that have never given birth and suckled young. By contrast, the 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 2005b).

A B

Fig. 53. Female (A) long-tailed bat (*Chalinolobus tuberculatus*) and (B) lesser short-tailed bat (*Mystacina tuberculata*) showing conspicuous bare nipples that indicate they are lactating. *Photos: J Sedgeley*

Female long-tailed bats have occasionally been recorded carrying small babies with them when they go out to forage (Fig. 54). 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 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.

Fig. 54. Female long-tailed bat (*Chalinolobus tuberculatus*) caught in a harp trap with a young baby attached to her nipple. *Photo: CFJ O'Donnell*

8.4.3 When to look for reproductive females

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

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), where births begin on 1 November, c. 1 month earlier than in other populations. By contrast, the average birth date in Hawke's Bay is around the last week of November, while bats in the Eglinton valley have the latest birth dates, with births occurring throughout December and two bats that were visibly pregnant being observed in the first week of January, although these were first-time breeders (O'Donnell 2002). It has also been shown that most births in the Eglinton valley are highly synchronous, with 70% of births occurring during a 10-day period in mid-December, and the sex ratio at birth is equal. Females first gave birth at 2–3 yrs old (O'Donnell 2002).

Lesser short-tailed bats

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

8.4.4 Reproductive status of males

In some bat species, the testes of juveniles are smaller than those of 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, but the 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 and Racey 2004).

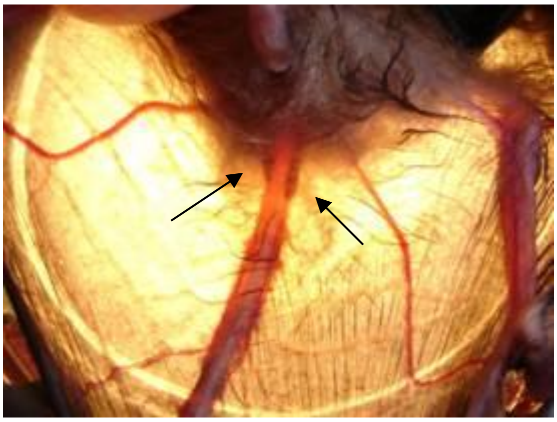


Fig. 55. Location of the epididymides in a long-tailed bat (*Chalinolobus tuberculatus*), the colour and distension of which can be used to assess breeding condition. *Photo: J Sedgeley*

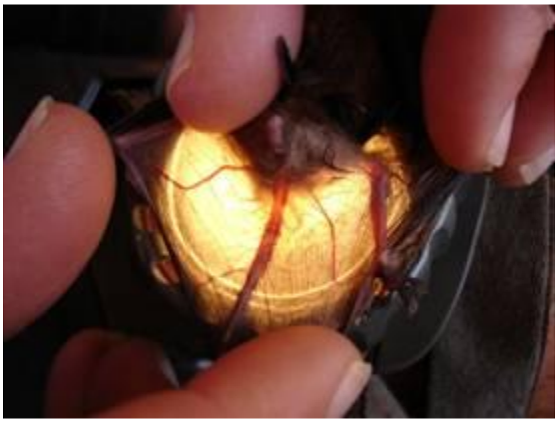


Fig. 56. Positioning a long-tailed bat (*Chalinolobus tuberculatus*) on a torch to examine the epididymides. *Photo: J Sedgeley*

Long-tailed bats

In long-tailed bats, the epididymides are attached to the testes and lie either side of the tail (Fig. 55). Therefore, the easiest way to examine them is by illuminating the tail from behind by holding it over a torch. A torch with a reasonable diameter head is best (Fig. 56), but it is important to be aware of how hot the torch gets – for example, a spotlight could burn the bat.

A 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 peritoneal sheath (the tunica vaginalis), which is densely pigmented in juveniles and sexually immature males, causing the epididymides to appear black and be clearly seen through the skin (Fig. 55 and Fig. 56). After spermatogenesis, sperm are released from the testes of adult males and pass through the epididymides into the caudae (tails of the epididymides). The caudae then become distended and stretch the peritoneal sheaths, separating the black pigment cells and causing the epididymides to appear clear, pale grey or white.

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 the testes nor the epididymides are visible externally at any time of the year. Other techniques that are used to assess the reproductive status of male bats in other countries include surgical examination and examination of the urine for the presence of sperm. However, neither of these techniques has been trialled in New Zealand.

8.4.5 When to look for reproductive males

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

8.5 Common measurements

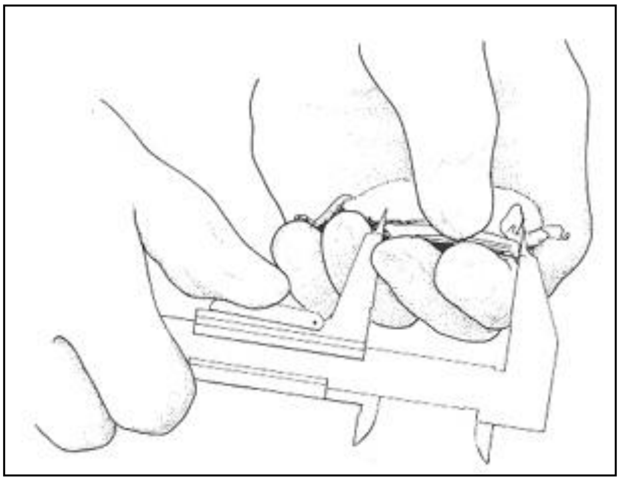
When taking any measurements, it is important to record a number of other details, including the date, time, place of capture, method of capture and reason for capture. Morphological measurements should also be supported by information on the species, sex, age and, if possible, reproductive condition. The fate of each individual captured should also be recorded – for example, 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) containing blank boxes 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, so it is important to routinely record who took the measurement so that any biases can be considered when undertaking analyses.

8.5.1 Forearm length

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

The 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 to use the dominant hand for manipulating the callipers (Fig. 57). The elbow of the bat is rested on the movable jaws of the callipers and the callipers are adjusted until you can see or feel a slight movement of the skin of the bat's wrist against the fixed jaw (Hutson and Racey 2004). Calliper measurements are usually taken to 0.1 mm accuracy.

A



B

Fig. 57. (A) Diagram and (B) photograph showing how to take forearm length measurements. *Images: (A) TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588; (B) J Sedgeley*

8.5.2 Body mass

The body mass of bats varies among different age and sex classes. Additionally, the body mass of an individual bat can vary greatly over a 24-hour period and seasonally, changing by as much as 30–50% depending on the time of year, reproductive condition and foraging success. The body mass of all bats should increase in autumn (late March) as they accumulate body fat reserves to help them survive through the winter months. The body masses of males are relatively stable outside this time, whereas those of reproductive females also vary according to the time of the breeding season (Hutson and Racey 2004).

This level of variation puts great limits on the usefulness of body mass data. However, body mass is a useful measurement in long-term studies of growth and body condition (Hutson and Racey 2004) and **must** be measured to ensure that a bat is heavy enough to have a transmitter attached, as the transmitter should weigh less than 10% of the bat's weight (see Choosing the correct tracking device).

The best way to measure body mass is to use a spring balance such as a Pesola. A 50-g Pesola balance (long scale) is ideal and can be read to an accuracy of 0.1 g. A range of other Pesola balances are also available (e.g. 30 g, 60 g, 100 g or larger), but some of these may give less accuracy because they can only be read to 0.5 g. Spring balances can be obtained through the DOC Banding Office (bandingoffice@doc.govt.nz). Small electronic kitchen-style scales can also be used but are seldom practical in the field because they require a flat, firm surface.

Before undertaking the measurement, ensure that the Pesola balance is set to zero. It may be necessary to calibrate the balance, which is done by turning the flat screw at the top. When reading the scale, it is important to hold the ring at the top so that the balance swings – if the body of the balance is held while taking measurements, the readings will be affected.

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, and a lighter bag will also improve measurement accuracy. Alternatives such as cloth cones (with the bats inserted down into the narrow end) or narrow sections of elasticated tights/pantyhose that can be wrapped around the bat can also be used, but plastic bags should be avoided because bats seem to get stressed in them despite being held inside for a very short time.

It is Important to regularly re-weigh the empty bag, particularly if measuring a large number of bats, as the bag will change weight if it gets damp or filled with debris or bat droppings. Ensure that the balance and bag are free of obstruction and wait until the bag and bat have stopped moving around before taking the measurement.

8.5.3 Body condition index

Because the body mass of individual bats can vary so much, it is only really a useful measurement 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, as this procedure corrects for differences in skeletal size between bats without the loss of mass units (O'Donnell 2002). The body condition index is calculated by dividing the individual's body mass by its forearm length and then multiplying by the mean forearm length for the total sample of bats that has been examined.

8.6 Other measurements

Many features of a bat can be measured (see Fig. 46), but 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] for Australian bats), so 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, limiting the usefulness of measurement data. However, limiting the number of people who are taking measurements will reduce this variability in long-term studies.

8.6.1 Features that can be measured

The more common measurements for bats include the:

- head and body length (from nose tip to anus)
- head length (from junction with neck to nose tip)
- body length (from junction with neck to anus)
- tail length (from anus to tail tip)
- wing depth at fifth digit (from inside of wrist to tip of finger)
- wingspan (from wing tip to wing tip)
- foot length (from 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)
- tragus width (maximum width)
- tragus length (maximum length from base to tip, ignoring any curves)
- outer canine width[4] (distance between the upper surfaces of the canines at the gum line).

Notes on any abnormalities (unusual colouration, injuries, deformities) should also be included.

[4] Note that this measurement is extremely difficult to take in live bats.

8.6.2 Wing depth at fifth digit

The easiest and most reliable way to measure the wing depth at the fifth digit (or fifth digit length) is to measure from the outside of the wrist to the fingertip. This is best done on a flat surface (Hutson and Racey 2004; Fig. 58). 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. Previous measurements of the fifth digit in long-tailed bats have been taken using the first method (O'Donnell 2005).

Fig. 58. Measuring the wing depth at the fifth digit of a long-tailed bat (*Chalinolobus tuberculatus*). Note that 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 that it is flat. *Photo: CFJ O'Donnell*

8.6.3 Wingspan

The wingspan is not a particularly useful field measurement because there is too much potential variation in the measuring technique (Hutson and Racey 2004). However, this measurement is 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. 59). It is very difficult to determine how far to extend the wings, and care **must** be taken not to overextend and injure the bat. **This technique is not recommended for lesser short-tailed bats.**

Fig. 59. Measuring the wingspan of a long-tailed bat (*Chalinolobus tuberculatus*). *Photo: CFJ O'Donnell*

8.6.4 Ear length and tragus measurements

Ear length is difficult to measure because bats often fold down their ears when being handled and it is important to ensure that 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, while tragus width is taken at the maximum width and tragus length is the maximum length from base to tip, ignoring any curved edges (Hutson and Racey 2004) (Fig. 60).

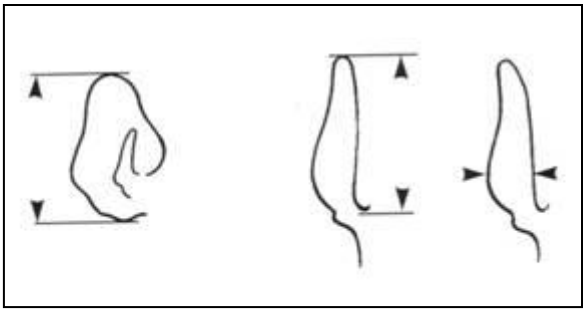


Fig. 60. Locations of ear and tragus measurements. *Diagram: TP McOwat, reproduced with permission from the Bat Workers' Manual, ISBN 1861075588, © JNCC 2004*

8.6.5 Wing tracing

Morphological data can be obtained using wing tracings. Standard aerodynamic measurements, such as wing loading, aspect ratio and wingtip shape index, can be calculated from wing tracings using the conventions outlined by Norberg and Rayner (1987). 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. The 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 bat's wing. The bat is placed face down on a sheet of paper that is attached to a hard surface such as a clipboard. It is important to open and draw only one wing, including the head and tail, and to keep the leading edge of the wing as straight as possible (Fig. 61).

Fig. 61. Holding a long-tailed bat (*Chalinolobus tuberculatus*) in position to take a wing tracing. *Photo: CFJ O'Donnell*

Other techniques can also be used as an alternative to tracing. For example, the bat can be laid face down with its wings gently extended and taped down with sticky tape, and a photograph including a reference scale can be taken using a digital camera. Image software is then used to take measurements (Fig. 62). **Note that this technique has not been trialled in New Zealand and ethics approval would be required to tape down the bat.**

Fig. 62. Taping an Australian free-tailed bat (*Mormopterus* sp.) to obtain wing measurements. *Photo: L Lumsden*

8.7 Collecting samples

8.7.1 Skin for DNA analysis

For information on how to obtain a skin sample for DNA analysis, see Taking tissue samples for genetic purposes.

8.7.2 Ectoparasites and associated insects

Many arthropods live on bats for at least part of their lives, and New Zealand bats are host to several ectoparasites. Both long-tailed bats and lesser short-tailed bats host several species of mites, and long-tailed bats also host the long-tailed bat flea (*Porribius pacificus*), while lesser short-tailed bats host an undescribed species of tick belonging to the genus *Argas* (*Carios*). Lesser short-tailed bats are also associated with the endemic and threatened New Zealand batfly (*Mystacinobia zelandica*), which is not a parasite but feeds on bat guano throughout its life cycle. Although superficially similar to other batflies, the New Zealand batfly evolved separately and has been placed in its own family (Lloyd 2005b). 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 and Racey 2004).

Parasites and batflies are found by inspecting the flight membranes, feet, ears, face and anus, and 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 these 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 it is, as 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 and Racey 2004). Stored specimens should be clearly labelled with full data, including the date, locality, name of collector and details of the bat host (e.g. species, age, sex, position on body). Refer to *Collection, Storage and Transport of Diagnostic Samples From Birds and Reptiles* (see Appendix 1) for more details.

8.7.3 Preserving dead bats

Dead bats should never be discarded immediately as they may be useful for a variety of purposes (e.g. as a voucher specimen, for exhibitions, for disease screening, for education and training, for 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 also an unfortunate fact that any project involving the 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, and 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 **must** be adhered to. For full details, refer to DOC's *Wildlife Health Management SOP* (see Appendix 1), which provides clear guidelines on when it is appropriate to send specimens away for post-mortem examination. If the bat is to be sent for necropsy, it should be sent to a veterinary pathology laboratory according to the instructions for submitting samples in appendix 2.3 of the *Wildlife Health Management SOP*.

Generally, a dead animal should be chilled to refrigerator temperature (c. 4°C) as soon after death as possible and dispatched for diagnosis on the earliest available transport. Freezing interferes with the examination of tissues and some aspects of microbiological culture and so should only be used as a last resort if the dead body cannot be delivered within approximately 24–36 hours. Alternatives include fixing the body whole in alcohol, or undertaking a field dissection and submitting the fixed tissues for histopathology. Researchers outside DOC should contact their local DOC office and send the specimen where directed.

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

If it is decided that a specimen will be sent directly to a museum (Museum of New Zealand Te Papa Tongarewa in Wellington or Canterbury Museum in Christchurch), it is best to preserve the bat in one of three ways, listed here in order of preference: 1. frozen, 2. preserved in 70–80% ethanol or 3. preserved 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 is propped open with a piece of matchstick. All specimens should be clearly labelled with the 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 methylated spirits or 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).

8.8 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 the implications of release for any bats that have been held for a longer period.

8.8.1 Where and when to release bats

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

8.8.2 How to release bats

Warm and active bats will often fly directly out of a holding bag once the top has been 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. 63). Having a bat detector or light switched on will help confirm that the bat has flown away rather than fallen to the ground – but do not shine the light directly into the bat's face. Ruru/morepork may be attracted to roosting or trap sites and have been observed pursuing released bats. If these birds are proving to be a problem, move to a less open release site to give the bats more cover as they fly off.

Young bats that have recently started flying and adult females that are heavily pregnant sometimes need a bit of extra lift to take off. To assist with this, find an open area to stand in that is free of obstructions and hold your hand high in the air. The ground should be fairly clear of low vegetation for several metres in front of you so that you can easily find and retrieve the bat if it lands. Sometimes it may be necessary to put a bat onto a tree so that 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 and generally occurs when a bat has been by itself in a holding bag (rather than with a group of bats in the same bag) or when bats have been captive in a harp trap bag for several hours (see Extracting bats). A bat can be warmed up by holding it inside loosely cupped hands or popping it inside a bag and carefully placing it down the front of your clothing and close to warm skin. Be mindful of your passenger by remaining calm and quiet and minimising activity. Care should be taken when handling lesser short-tailed bats because they may bite through gloves or a bag as they become active.

If a bat has fallen to the ground, be very careful not to step on it while searching for it. Ensure that everyone in the team knows that a bat is on the ground and keep the movement of team members to a minimum. When found, pick the bat up gently, warm it up and try to get a bit higher before release, or place the bat on a tree so that it can get as high as it needs to and take off when it is ready.

Fig. 63. Releasing a long-tailed bat (*Chalinolobus tuberculatus*) from the hand. *Photo: D Geddes*

8.8.3 Success rates of returning bats to the wild

Bats are long-lived animals, so it seems reasonable that they may have good long-term memories of their home range and abilities to home (Racey 2004; Guilbert et al. 2007). However, the success of returning bats to the wild may depend on the length of time they have been held captive and other factors, such as their flying ability (Racey 2004). Radio-tracking and banding studies have shown that bats that are caught, handled, examined and released on the same evening, or are released on the evening following capture, are able to integrate back into their colonies. Furthermore, 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 on the same night as they were released (Sedgeley and Anderson 2000).

Bats that have been born and raised in captivity may not be suitable for release back to the wild for several reasons, such as their 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 that the survival rates of these bats is low (Racey 2004). However, there have been instances of bats that were raised in captivity habituating to the wild (e.g. Devrient and Wohlgemuth 1997).

9. Banding and marking

Note: New Zealand bats are fully protected, and it is illegal to catch, handle, mark or keep them without appropriate permits and ethics approvals (see Permitting, ethics approval and training).

This section describes some of the more common techniques that are 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 Use of tracking devices).

There are several effective techniques for temporarily marking bats that are applicable to both extant New Zealand species, all of which are described below in Short-term marking of long-tailed bats and lesser short-tailed bats. However, the suitability of various long-term marking techniques and their effects differ between the two bat species due to the marked differences in their morphology (body size and shape) and behaviours, and not all techniques have been trialled on both species. Long-term techniques for marking long-tailed bats (both trialled and untrialled) are described in Long-term marking of long-tailed bats using metal bands and Long-term marking of long-tailed bats using other methods, while those for lesser short-tailed bats are described in Long-term marking of lesser short-tailed bats using PIT tagging.

9.1 Introduction to marking

9.1.1 Why mark bats?

The ability to recognise individual animals plays an integral part in ecological research and the 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).

9.1.2 Important considerations before undertaking marking

All marking methods will affect the subject to some degree, at least in the short term. Marking can affect animals by altering their 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 (Beausoleil et al. 2004).

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, and incorrectly placed or poorly fitted tags may lead to injury and loss of function.
3. The observation of marks almost always requires repeated recapture and handling, leading to further stress.

Therefore, workers **must** consider whether it is really *necessary* to mark bats to achieve the proposed research/management objectives, and the 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 on bats can be minimised by recognising the advantages and disadvantages of different marks and marking procedures and by choosing the most effective and humane way of applying the marks (Mellor et al. 2004). Before selecting a particular technique, a number of issues **must** be considered, including the (adapted from Barclay and Bell 1988; Beausoleil et al. 2004):

- specific objectives of the study and the nature of the data required
- level of recognition required
- species 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 the study and the length of time marks are required to last
- amount of time and resources available to researchers
- number of individually distinct marks required
- speed at which the bats will need to be marked
- required proximity of an observer to identify the mark
- necessity to recapture the bat to read the mark
- levels of training and experience required.

Only two methods are currently approved for the 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 associated with 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 that will help improve our understanding of New Zealand bats and aid in their conservation should be encouraged, but all projects **must** incorporate appropriate and adequate protocols for monitoring the potential effects of new techniques.

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

Short-term marks can last from 1 day to several weeks, depending on the technique used, and are generally far less invasive than long-term marks, being less likely to cause tissue damage, pain or stress, and unlikely to restrict movement.

9.2.1 Fur clipping

Fur clipping is a useful technique for identifying individuals or groups of bats and has no known harmful effects (Fig. 64). These 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.

Fig. 64. Marking a lesser short-tailed bat (*Mystacina tuberculata*) by clipping the fur 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 from biting. This type of restraint is not necessary for long-tailed bats (*Chalinolobus tuberculatus*). *Photo: CFJ O'Donnell*

Fur is usually cut from the dorsal surface of the bat in up to 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 fur clip in the middle of a bat's back could be confused with a mark left by a radio-transmitter. Using four clipped patches will allow many unique combinations, particularly if the age and sex of the bat are also recorded. The fur is most easily cut with sharp scissors or electric moustache/beard trimmers. When using sharp scissors, ensure that the bat is held securely and cover the bat's head to protect its ears.

Advantages

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

Disadvantages

- The marks can grow out within 2–3 weeks during the moulting season.
- If a large number of bats need to be marked, the individual mark combinations become more complex and consequently the handling time per individual increases.
- Bats have to be re-caught to read the marks.

9.2.2 Dye marking

Black hair dye has previously been applied to lesser short-tailed bats (B Lloyd, DOC, pers. comm.), but there are several disadvantages with this technique. Almost all human hair dyes contain harsh chemicals, such as peroxides, and the bat handling time is relatively long because the dyes do not 'fix' or 'set' immediately and will quickly rub up if not dry and set adequately. Quick-drying coloured antiseptic skin dyes such as 'gentian violet' or 'magenta' have also been trialled on bats. For example, an individual lesser short-tailed bat was marked with magenta during a captivity trial and it was found that, while the mark faded, it was still readable at the end of the 26-day trial (J Sedgeley, unpubl. data); and gentian violet has been used by wildlife carers in Australia to mark bats, although recent trials on lesser short-tailed bats in the Eglinton valley proved to be unsuccessful, with marked bats that were held overnight in cloth bags having lost their marks by the next day. Dye marks had also disappeared from wild bats that were both fur clipped and marked with gentian violet when they were recaptured 4 days after being marked (CFJ O'Donnell, unpubl. data). Magenta and gentian violet can be obtained from a chemist/pharmacy, but a prescription is required for gentian violet.

The use of small blobs of non-toxic paint could also be considered, but this technique has not been trialled. Toxic dyes or paints **must not** be used.

Dye marking using non-toxic dyes or similar has the same advantages and disadvantages as fur clipping. However, potential welfare effects (e.g. increased risk of predation) also need to be considered.

9.2.3 Chemiluminescent 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 and 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 (Lumsden et al. 1994; Christie 2003a).

The cheapest and easiest method of chemiluminescent tagging is to use small capsules filled with diphenyl oxalate (trademark name Cyalume) (Buchler 1976). Cyalume consists of a phosphor compound and a peroxide-based reactant that produce a bright 'cold' light when mixed. The brightness and duration of the light depend on the relative proportions of the chemicals, with equal proportions producing a very bright light that lasts 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 the 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), although other studies have found no evidence of this toxicity (Racey and 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 2003a). Chemiluminescent lures can be obtained from fishing shops and Kmart. 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 have been attached to the untrimmed fur of New Zealand bats initally using F2 Multipurpose Contact Adhesive (Ados Chemical Company Ltd, New Zealand), although now hospital-grade skin adhesives or those used for veterinary purposes ('Skin Bond' or 'Vet Bond' equivalents) are best, but the availability of such glues varies (and their brand names) change frequently. . 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 is pressed into place once the glue has dried (Fig. 65). Attachment of the tag to untrimmed fur allows the bat to groom it off in a relatively short period of time. There seems little point in attaching the tag more firmly if the luminescence wears off after a few hours, but if it is necessary to retain the tag on the bat for a longer period, the bat's fur can be trimmed before attachment. Note, however, that if the bat's skin is nicked or cut with scissors during trimming, tags **must not** be fitted.

Fig. 65. Long-tailed bat (*Chalinolobus tuberculatus*) fitted with a fishing lure Cyalume capsule. *Photo: CFJ O'Donnell*

Advantages of Cyalume tags

- No known harm is caused to bats by fishing lure tags – however, potential welfare effects (e.g. increased risk of predation) need to be considered.
- The tags are relatively quick and easy to apply.
- The tags are made of a plastic that appears to be thick enough to prevent bats from biting through them.
- The tags are very light weight (< 0.8% of lesser short-tailed bat body mass)
- The tags are very cheap.

Disadvantages of Cyalume tags in general

- The toxicity of Cyalume liquid to bats is questionable.
- The lifespan of Cyalume tags is extremely short.
- Cyalume tags provide very limited options for individual bat 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 a forest interior (Christie 2003a).

9.2.4 Glue-on tags

Various materials have been glued onto bats, but these are usually groomed off fairly quickly if the bat is active. However, such tags may remain in position for long periods when bats are hibernating

(Daan 1969; Stebbings 2004) and frequently stay on long enough to obtain useful data, such as information on roost use. For example, bats marked with radio-tags, Cyalume capsules or plastic disks can be clearly picked out entering and exiting roosts on video recordings (Sedgeley and Anderson 2000). Coloured reflective tape (attached using the sticky back or by 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 temporary tags but are more commonly inserted under the skin as permanent tags (see PIT tags below).

Advantages

- No known harm is caused to bats – however, potential welfare effects (e.g. increased risk of predation) need to be considered
- 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 noticed on video cameras, enabling bats to be identified without being recaptured

Disadvantages

- Glue-on tags are generally relatively short-lived.
- PIT tags can only be detected at distances of 1–30 cm and require a specialised reader.

9.3 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) is the only method currently approved for the long-term marking of long-tailed bats. Bands must be purchased from the DOC Banding Office[5] (bandingoffice@doc.govt.nz), not directly from Porzana, and bands (and mist nets) will only be supplied to people with the appropriate registered competencies.

9.3.1 Use of metal bands

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

Several countries, including Australia, Great Britain and the 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

[5] The DOC Banding Office stocks various equipment in addition to bands and mist nets. Visit the DOC website for a price list and order form.

New Zealand resulted in approval being granted 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, so banding is not currently approved for this species (see Long-term marking of lesser short-tailed bats using PIT tagging below).

Advantages

- After sufficient training, metal bands are relatively easy and quick to apply.
- Numbered bands allow the 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

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

9.3.2 Trials of metal bands on long-tailed bats

The most comprehensive banding trial of long-tailed bats to date 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, on 119 free-living bats. Half of these bats were fitted with standard bird bands, while the other half were fitted with modified bands that had the sharp corners filed down. After 2–4 weeks, bats were recaptured and assessed for wing damage and other injuries. Wing damage was recorded in 37% of recaptured bats with unmodified bird bands and 77% of bats with modified bands, with most injuries being judged as slight or moderate but six 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 (CFJ O’Donnell, unpubl. data).

Flanged bat bands (of different sizes) from Great Britain and Australia were trialled during the following summer season. It was found that the British bands, which were made of a softer metal and had smoother edges, did not cause any short- to medium-term injuries, whereas the Australian bands did cause some injuries. Consequently, only British bands continued to be used (Fig. 66).

In Eglinton valley, 4748 long-tailed bats have been banded , and only 13 injuries were recorded over a total of 21,018 recaptures. Injuries ranged from very mild to severe swelling of the forearm (CFJ O'Donnell, unpubl. data).

Fig. 66. A British 2.9-mm flanged bat band (ring) fitted to a long-tailed bat (*Chalinolobus tuberculatus*). *Photo: CFJ O'Donnell*

9.3.3 Applying bands

Bands are fitted over the bat's forearm. Some studies elect to use one forearm for females and the other for males, although this has limited application. To minimise damage, it is very important that the band is fitted as loosely as possible so that it can freely slide up and down the forearm. However, the gap should be sufficiently small to prevent the band from sliding over the wrist or elbow joint, trapping the finger bones. It is also important that the band is closed evenly, as a band that is pinched at one end and open at the other may cause injury (Barclay and Bell 1988; Stebbings 2004). The gap closure for bands fitted to long-tailed bats should be c. 0.7–0.8 mm (Fig. 67).

Fig. 67. Correct closure on a long-tailed bat (*Chalinolobus tuberculatus*) band. The closure (c. 0.7–0.8 mm) is small enough to prevent the band from sliding over the wrist or elbow, trapping the finger bones, and the band is closed evenly. *Photo: CFJ O'Donnell*

An effective banding technique is to hold the bat face down in the palm of the hand with the wing partially extended (Fig. 68A). The forearm **must** be supported by the first or index finger and the band is applied with 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 that needs to be applied to the band (see Fig. 68 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 bat bands so they retain the correct shape and gap closure.**

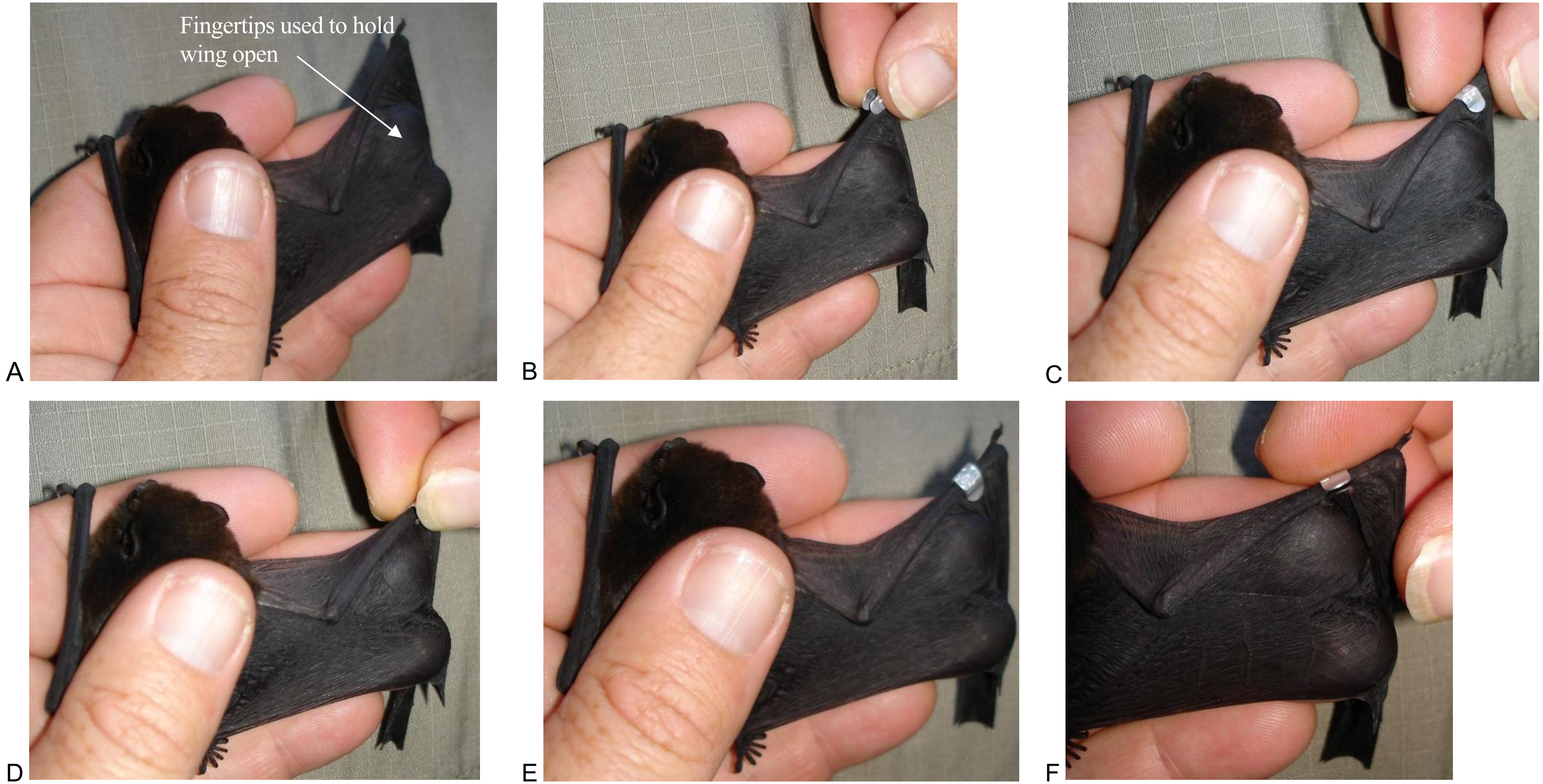


Fig. 68. Technique for applying bands to long-tailed bats (*Chalinolobus tuberculatus*). (A) Hold the bat face down in the palm of the hand with the wing partly extended; fingers can be positioned beneath the wing membrane between the bat's body and fifth digit to keep the wing open. (B–C) Slide the new open band into place on the bat's forearm using the thumb and forefinger. (D) Gently squeeze the band shut using slow and even pressure from the thumb and fingertips. (E) The closed band should not pinch the wing membrane and should be free to move up and down. (F) The gap should be sufficiently small (c. 0.7–0.8 mm) to prevent the band from sliding over the wrist or elbow joint, trapping the finger bones. *Photos: J Sedgeley*

9.3.4 Removing bands

If the band is closed too tightly or unevenly, it should be removed and a new one fitted. If any recaptured bats are injured or the band appears to be damaged, the band **must** be carefully removed using fine circlip 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 circlip pliers. However, the ends of the pliers need to be fine enough to insert inside the band, so the tips of the pliers may need to be filed down if they are not fine enough. If no circlip 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 fingernails or loops, it is very important that one person holds the bat and supports the forearm each side of the band while another person removes the band. The latter techniques are very fiddly and have a higher chance of causing injury, so it is far preferable to have a fine pair of circlip pliers as part of a standard banding kit.

9.3.5 Obtaining bands

The only type of band that is approved for use in New Zealand is a 2.9-mm (narrow) aluminium alloy flanged bat band manufactured by Porzana Ltd in Great Britain. These bands are now **only** available in New Zealand through the DOC Banding Office (bandingoffice@doc.govt.nz). The design of the band is the result of many years of experimentation and is characterised by a lack of sharp edges or burrs. **Bands will only be approved for those who have demonstrated a legitimate need to band, have appropriate permitting and ethical approvals, and can demonstrate that they have had sufficient training to band bats, or have arranged for appropriate training** (refer to *Bat Handling Competencies* [see Appendix 1].

9.3.6 Band recording scheme

A full record of all bands applied to bats **must** be maintained. As a minimum, this should include details of the band number, location, date, age, sex and reproductive condition of the bat, as well as the name of the person who undertook the banding. A copy of all banding records **must** be submitted to the DOC Banding Office (bandingoffice@doc.govt.nz).

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

New research and management projects may require alternative marking techniques for the long-term monitoring of long-tailed bats. A variety of techniques have been used on other bat species, the more common of which are described below. However, these either have not been trialled on long-tailed bats or have proven to be ineffective or unsuitable for this species, so trialling these methods in New Zealand would require ethics approval.

9.4.1 Coloured and plastic bands

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

Advantages

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

Disadvantages

- Split plastic bands have caused injuries in some bat species in Britain (Stebbings 2004), and this may also be an issue in New Zealand as split metal bird bands have injured long-tailed bats.
- These 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.

The use of plastic bands has not been trialled on long-tailed bats, but given that split metal bird bands have caused injuries to long-tailed bats, **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.

9.4.2 Reflective bands

Reflective coloured tape can be applied to metal bands to aid in the identification of individuals or sexes during flight or in the roost (reviewed in Barclay and Bell 1988). Reflective tape greatly enhances the visibility of bands in artificial light, or with camera or image intensifiers, and 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.

9.4.3 PIT tags

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

9.4.4 Necklaces

Necklaces have been used in some bat species where wing banding has caused injury and infection or there has been excessive band chewing. Necklaces **must not** be used on growing juveniles.

Bats have been marked using necklaces made out of bead-clasp keychains or ratchet-style plastic electrical ties, with the latter appearing to cause less abrasion around the bat's neck and allowing for finer size adjustment (Barclay and Bell 1988).

Necklaces made of thin ratchet-style plastic electrical ties were trialled on six free-living long-tailed bats in 1993/94. The ties were made of small knobbly beads and covered in coloured electrical heat shrink tubing in an attempt to make the necklaces smoother and less likely to irritate the bats. Different colour combinations of heat shrink were used to individually identify each bat. The bats were recaptured at regular intervals over the summer season and it was found that by the end of the summer, the necklaces had worn away the fur in a band around the bats' necks, leaving the skin bare but intact with no open wounds. All the necklaces were removed at the end of the trial because banding was considered to be more acceptable for the long-term marking of long-tailed bats (CFJ O'Donnell, unpubl. data). **The use of necklaces is not currently recommended as a technique for the long-term marking of long-tailed bats.**

9.5 Long-term marking of lesser short-tailed bats using PIT tagging

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

9.5.1 PIT tags

A PIT tag, or microchip, consists of a small, integrated circuit enclosed in a biologically inert glass capsule. Up until 2021, the PIT tags used in bats were 12 mm long and just under 2 mm in diameter. However, these tags are no longer being manufactured, so smaller sized tags that are approximately 1.4 × 8.4 mm are now being used. When being used as a long-term mark, PIT tags are inserted under the skin (subcutaneously) using a needle.

A PIT tag contains no power source of its own but rather 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 with a loop antenna or circular readers can detect tags from greater distances. The detection range depends on the reader type, power source and shape of the antenna and can range from about 1 to 30 cm. The antenna can be fashioned to go around the entrance of a roost.

Technological advances are likely to result in further miniaturisation of transponders and the 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. Therefore, it is advisable to contact the DOC Electronics Team to find out what types of PIT tags and readers are currently available and in use in New Zealand, and to find out more about new developments.

PIT tags are being used more widely as long-term marks for bats and, in some species, as an alternative to banding. Studies have shown that these tags are successful for examining roost use and social dynamics in Bechstein's bat (Kerth et al. 2002) and for mark–recapture survival estimates in the big brown bat (*Eptesicus fuscus*) (O'Shea et al. 2004), with 2073 bats being tagged in the latter study.

Advantages

- Each tag has a unique code, allowing a virtually unlimited number of bats to be individually marked.
- Tags are long lasting (> 10 years).
- The use of automatic readers and dataloggers at roost sites provides an opportunity for long-term monitoring without needing to recapture bats and is particularly useful for long-term survival studies.

Disadvantages

- The procedure for inserting PIT tags is highly invasive and may cause distress and pain.
- PIT tagging is a difficult procedure to use on a small animal and requires rigorous training. There are currently very few approved trainers – contact the Bat Recovery Group at bathandler@doc.govt.nz for more information.
- PIT tags can sometimes migrate underneath the skin, causing injury and pain, or can be expelled.
- Initial outlay for the transponders and particularly the readers is relatively expensive.

9.5.2 PIT-tagging trials in lesser short-tailed bats

PIT tags were first trialled on free-living lesser short-tailed bats in Rangataua Forest in 1997 The tags were inserted beneath the skin on the backs of 100 bats. Their skin was very thin and care was needed to prevent piercing the skin twice. Transponders were subsequently found ejected at the base of roost trees, and three or four bats had serious arterial bleeds (B Lloyd, DOC, pers. comm.;.

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 that were recaptured at 10 days and again at 3 weeks after they were released still had their tags in place, exhibited no evidence of scarring and were healthy. Further details can be found in Sedgeley and O'Donnell (2006, unpublished, see section 9.8, Notes).

Further successful trials in 2007 (Sedgeley and O'Donnell 2007, unpublished, see section 9.8, Notes) led to PIT tags being used to facilitate standard monitoring of the survival of lesser short-tailed bats. Automatic data logging systems have been developed by the DOC Electronics Team that record the presence of tagged bats at roost sites (refer to *Instructions for Setting Up RFID Readers, Dataloggers and Antennae* [see Appendix 1]) and the survival of tagged bats has been shown to be very high (O'Donnell et al. 2011).

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

9.5.3 Standard PIT tag for DOC work

When PIT tagging bats, DOC uses 12 mm or 8.4 mm tags, with a needle on a Henke Jet™ injector gun, which requires modification to allow it to work on the smaller sized tags.

9.5.4 Requirements to undertake PIT tagging in bats

Any staff involved in PIT tagging bats **must** have the appropriate skills for finding, capturing and handling bats as described in this manual.

Anyone wanting to PIT tag bats **must** also receive training from someone with previous experience in applying this technique to bats. There are very few approved trainers in New Zealand, so contact the Bat Recovery Group at bathandler@doc.govt.nz for more information.

We recommend selecting the appropriate person to learn to PIT tag bats based on their:

- Manager's approval
- ability to commit enough time to training, as well as to undertake PIT-tagging sessions over several seasons
- relevant skills (e.g. experience in PIT tagging other species, taking blood from animals, vaccinating farm animals)
- prior 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.

9.5.5 PIT-tagging procedure

Three people are generally required to PIT tag bats: one person to hold the bat (the handler), one person to insert the tag (the injector) and a third person to record the data (the recorder). We also recommend that a fourth person is used to handle the bat bags when dealing with large numbers of bats. Trainees should become familiar with and capable in all aspects of the procedure, although we acknowledge that some people may not wish to inject bats.

This guideline is split into three sections that follow the recommended staged approach to PIT tagging.

Assembling the PIT-tagging kit

A standardised and well-organised kit is essential for PIT tagging. We recommend having enough equipment to make up three or four kit bags. Each bag and its component items should be clearly labelled, and bags **must** be checked regularly to replace any items that have been used.

Before beginning tagging, trainees should familiarise themselves with the equipment and make up clearly labelled kit bags, each of which should contain (Fig. 69):

- two **closed-cell foam pads** or fold-up chairs for the 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); the DOC Electronics Team has developed a reader that connects via Bluetooth to a phone app to store PIT-tagging data before they are downloaded to a database
- an **insertion gun** – we strongly recommend using the Henke Jet steel insertion gun, as it is safer to use than the disposable blue gun that comes with the PIT tags, which is intended to insert only a limited number of PIT tags and can jam if overused
- a large supply of **PIT tags** – the tags are inside the needles, which 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 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 back 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
- a **rubbish bag** (for spare sticky labels and used alcohol wipes)
- **recording sheets** to record all new PIT-tag numbers and any PIT-tag numbers from recaptured bats; refer to *Transponder Field Recording Sheet for New Tags* for new bats and *Blank Field Recording Sheet for Transponder Recapture* for recaptured bats (see Appendix 1).

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

Fig. 69. 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 9-V 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 first be adept at handling, ageing and sexing bats.

The following procedure should be used to hold the bat in the preferred position for tagging.

1. The handler should reposition 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. 70), although some injectors 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 knee or thigh with the posterior of the bat pointing outwards to ensure that its back is sitting flat for the injection (Fig. 71). It is best if the handler’s knee is positioned slightly higher than the injector’s knees so that the injector is level with the bat. If the injector is right-handed, the bat should be held on the handler’s left knee and vice versa.

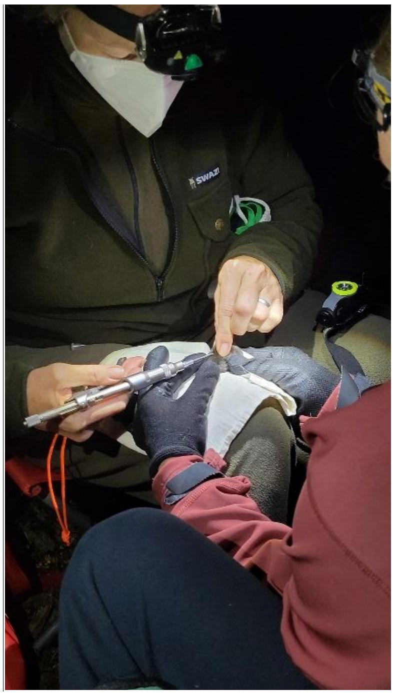


Fig. 70. The best position for PIT tagging is for the bat handler (left) and the injector (right) to sit facing each other. The bat needs to be on the top of the handler's knee or thigh with the posterior of the bat pointing outwards to ensure that its back is sitting flat for the injection. *Photo: T Thurley*

3. It is important to restrain the bat firmly and place its head underneath the edge of a cotton bat bag to prevent it from struggling or biting (Fig. 71). However, it is also essential to try to limit the holding grip to the bat's 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 their knee as possible 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.**

A

B

C

Fig. 71. The best position to hold a lesser short-tailed bat (*Mystacina tuberculata*) during PIT tagging. (A) The bat's head is restrained underneath the edge of a cotton bat bag to prevent it from struggling or biting. (B) The holding grip should be limited to the bat's wrists, elbows and feet to keep the skin as loose as possible. (C) The fingers and hands of the bat handler **must** be held away from the bat to afford the injector clear access to the injection site. *Photos: (A) J Sedgeley; (B and C) L McBride*

Learning how to inject PIT tags into bats

This section provides step-by-step instructions on how to inject a PIT tag into a bat.

Before starting, we recommend that trainees watch the short video clip *Micro-chipping a Lesser Short-tailed Bat* by Jane Sedgeley and Kate McInnes (see Appendix 1).

Inject a PIT tag using the following procedure.

1. Prepare the Henke Jet injector gun ready for tagging.
    a. Wipe the gun with an alcohol wipe.
    b. Set the gun to 'S' (twist the end of the gun to change settings; Fig. 72) – this 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 **must not** be used on a live animal. Remove the needle from its packet and gently twist the needle onto the gun.
    d. Uncap the needle. The needle should not be uncapped until it is on the gun and should not be totally uncapped until just before use. However, if necessary, the needle can be uncapped and left with the cap loosely covering it to protect it while preparing the injection site. The cap of the needle should then be eased off slowly and carefully.
    e. Align the needle. The front of the gun **must** be twisted until the needle is aligned correctly so that the bevelled edge faces upwards (Fig. 72).

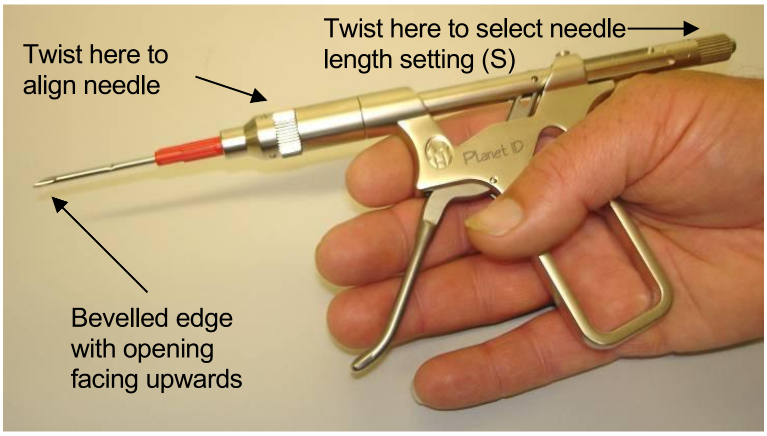


Fig. 72. Correct positioning of the needle in the insertion gun. *Photo: J Sedgeley*

2. If the injector is right-handed, hold the injection gun in the right hand and manipulate the skin of the bat using the left hand (and vice versa for left-handed injectors).

3. Insert the needle in a caudal to cranial direction (i.e. from the tail towards the head), 2–3 cm along from the shoulder blades (Fig. 73). It is very important not to insert the needle too close to the bat's head.
4. Hold the needle 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 a risk that it will go into the skin and then pass out again, creating both entrance and exit holes (a double puncture). Some PIT-taggers rest their gun hand on the handler's leg to steady the hand and make sure the needle is level.
5. Insert the tag sub-dermally (i.e. beneath the skin and not into the muscle). First, make sure the handler is holding the bat securely. Then, 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 the thumb and index finger – this action should loosen the skin and it should be possible to feel the difference between the skin and underlying muscle. A minimum of skin is then held pinched up between the balls of the thumb and forefinger to form a tent to inject into. Picking up a small fold of skin and inserting the needle at the base, close to the back of the bat, minimises the risk of double puncturing, as the needle punctures the skin almost immediately and is nowhere near the back end of the fold when you insert it another 5 mm and squeeze the trigger. By contrast, when a large tent of skin is used, the needle needs to be pushed about 5–10 mm under the finger grip before it punctures the skin, causing it to go very close to the back end of the fold and increasing the likelihood that it will pass out the back side when inserted to the normal depth.
6. Insert the needle below the fingers (so that it cannot be felt by the pinching fingers) 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.

Fig. 73. Close-up of the needle being aligned 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. Check the position of the tag. The aim is for the tag to be positioned between the bat's shoulder blades. It is difficult to judge the distance exactly when the skin is pinched up, and it does not 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. However, if the tag ends up too high (i.e. too close to the bat's head), it is much harder to manipulate it backwards.
9. Recap the needle, remove it from the gun and push it into the disposal container.
10. If tagging is unsuccessful, do not re-tag the bat. Tagging will sometimes be unsuccessful, usually because either the skin was punctured twice causing the tag to come out, or there was a misfire due to the needle failing to puncture the skin sufficiently or the needle not being pushed far enough under the skin. In any of these circumstances, the bat should not be re-tagged but can be fur clipped so that, if recaptured during subsequent tagging sessions, it can be identified as a 'failed attempt' and released immediately.
11. Limit the time you will work with the bats and the number of chips to insert. Between 20 and 30 tags per session is recommended to prevent the injector from becoming tired and potentially making mistakes (misfires).
12. Take compulsory 5-minute breaks with exercise, a snack and a drink after 1 hour of tagging.

9.6 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, but all with little success. To be able to develop new and more successful methods for the long-term marking of lesser short-tailed bats, the problems associated with techniques previously trialled **must** be carefully considered. Therefore, each of these techniques is described below.

9.6.1 Banding trials in lesser short-tailed bats

Captive trials

Five band types were trialled on six captive lesser short-tailed bats in Wellington Zoo. The bands selected for testing were size C and D celluloid split bird bands, a British 2.9-mm flanged bat band, 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. However, as the trial progressed it became apparent that this technique was unsatisfactory, so specialised banding pliers were developed specifically for the task (Lloyd 1995, unpublished, see section 9.8, Notes).

Both sizes of split bird bands caused swelling. Additionally, the C-band moved out of the insertion slit, while the D-band caused the insertion slit to enlarge to an 8 mm × 8 mm hole. The Australian 3.25-mm monel band also caused a hole in the wing membrane and swelling after 16 days. However, the British band caused no injury after 52 days. The 2.8-mm band was not on the bat on the first inspection after 7 days. The results of this initial trial led to further trials concentrating on the British bat bands, with a total of 15 being fitted to the six bats over a 5-month period. Eight of the British bands did not cause noticeable injury, but the remaining seven caused a variety of injuries, including holes in the wing membranes and, in one case, a wing membrane being caught in the tips of the band preventing the wing from opening. Table 5 summarises the characteristics of the bands that did and did not cause injury (Lloyd 1995, unpublished, see section 9.8, Notes).

Table 5 Characteristics of bands that did and did not cause injury to lesser short-tailed bats (*Mystacina tuberculata*) (Lloyd 1995, unpublished, see section 9.8, Notes)

CHARACTERISTIC	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

In the winter of 1996, 36 lesser short-tailed bats held in captivity on Codfish Island / Whenua Hou for 46 days were fitted with British Mammal Society 2.9-mm bat bands using fingers. No injuries were observed during the first 3 weeks in captivity but a large proportion of the bats exhibited minor irritation of the forearm tissue by the time of release, resulting in all bands being removed (Sedgeley 1996, unpublished, see section 9.8, Notes).

In 1998, 385 captive lesser short-tailed bats were fitted with 2.9-mm British bat bands. These were obtained from Lambornes in Britain and were made of an alloy called ‘incoloy’, which is harder than the magnesium-aluminium alloy Mammal Society bat bands previously trialled. However, all the incoloy 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 and 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 this method was discontinued after abrasions and swellings were seen on the forearms of banded individuals (M Daniel, pers. comm., cited in Lloyd 1995, unpublished, see section 9.8, Notes).

In 1996/97, 400 lesser short-tailed bats were banded in Rangataua Forest. However, 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 and McQueen 1997, unpublished, see section 9.8, Notes).

In 1995, 13 bats were banded on Codfish Island / Whenua Hou 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, unpublished, see section 9.8, Notes), 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 is not currently an approved technique for the long-term marking of lesser short-tailed bats, and the Bat Recovery Group recommends that banding **must not** be undertaken in this species until further notice . Given the range of band types already trialled, it seems unlikely that an acceptable method of banding for lesser short-tailed bats will be found. Some species of bats seem particularly susceptible to band injury, which is why banding is approved for some species but not others in several countries.

A combination of factors may explain why lesser short-tailed bats are particularly sensitive to the shape and closure width of bands and are more prone to band injury and infection compared with long-tailed bats and other bat species. For example, lesser short-tailed bats have a tendon extending from the wrist and running beneath the forearm that is not usually found in other bats species (Fig. 75). Bands can occasionally become clipped around this tendon (J Sedgeley, pers. obs.), and there may be a greater risk of injury if bands remain stuck in one place rather than freely moving up and down the forearm. Lesser short-tailed bats also frequently forage on the ground and amongst leaf litter, which may cause bands to become clogged with soil and other debris, contributing to increased levels of injury and infection.

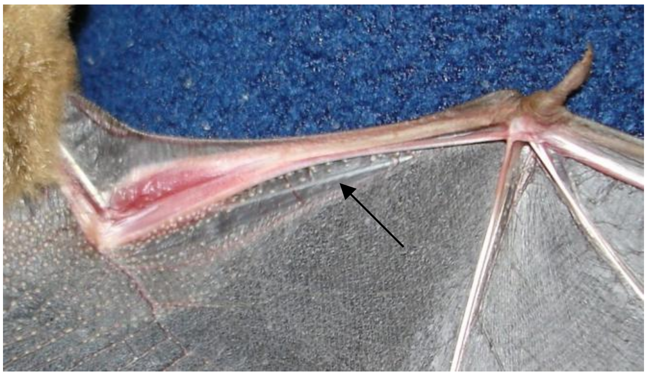


Fig. 75. Wing of a lesser short-tailed bat (*Mystacina tuberculata*) showing the tendon immediately below forearm. The presence of this tendon can sometimes restrict movement in forearm bands. *Photo: CFJ O'Donnell*

9.6.2 Tattooing

Tattooing is potentially one of the most permanent methods for marking wildlife, depending 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 and Bell 1988; Heideman and Heaney 1989).

Advantage

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

Disadvantage

- Tattoos on the wing membrane seem to only last a few months.

Tattooing trials in lesser short-tailed bats

This technique was trialled on lesser short-tailed bats held in captivity at Wellington Zoo but was considered to be unsuccessful (BD Lloyd, DOC, pers. comm). **The use of tattooing in lesser short-tailed bats is not recommended.**

9.6.3 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).

Advantage

- 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 the optimum application time. Applying the brand for too long will produce extensive skin sloughing and scarring, while applying it 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).

Freeze branding trials in lesser short-tailed bats

This technique was trialled on lesser short-tailed bats held in captivity at Wellington Zoo but was considered to be unsuccessful (BD Lloyd, DOC, pers. comm). **The use of freeze branding in lesser short-tailed bats is not recommended.**

9.6.4 Necklaces

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

10. Use of tracking devices

The following bat tagging techniques were approved for general use by the DOC AEC on 25 September 2017. The use of techniques that vary from those outlined below will require AEC approval.

In this section, we describe the reasons for tracking bats, the standards for attaching transmitters and the methods for tracking bats, and also provide a brief note about possible analysis techniques.

10.1 Types of tracking devices

10.1.1 Radio transmitters

Radio tracking is a standard technique for studying bats and a necessary precursor for inventory and monitoring and measuring the outcomes of management. Radio tracking is a specific technical skill; however, trackers will also need to be skilled in identifying areas of bat activity to find locations in which to place traps or nets; setting up harp traps or constructing mist net rigs; and handling bats competently. Training may be needed to learn how and where to place traps to optimise capture rates.

10.1.2 GPS tags

New GPS and satellite tracking technologies are currently emerging for use with bats as small as New Zealand species (e.g., Gonsalves et al. 2024). **The principles and guidelines outlined below are also applicable to the use and attachment of GPS tracking devices**. For example, they should not exceed the weight limits described below. GPS tags that are < 1 g in weight have the advantage of collecting precise (almost continuous) data on locations of bats up to the data storage capacity of the tag. **Specific Animal Ethics Committee permits are not required if they follow best practice for radio transmitters.**However, if it was proposed to use tags heavier than the radio transmitter weight thresholds, an AEC application and approval would be required first.The small tags that are suitable for New Zealand bat species do not currently have remote download capacity (i.e. the bat needs to be recaptured to download the data).

10.2 Reasons for tracking

Radio and GPS tracking can be used to:

- locate 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
- determine home range size and design adequate reserves
- identify movement patterns and important habitat types
- identify if development proposals (e.g. dams, roads or wind farms) might affect bats
- measure short-term survival in relation to management (O'Donnell et al. 1999)

- study behaviour.

10.3 Disadvantages and problems with tagging

- The biggest issue with tracking bats is keeping the tags attached for long enough to last through any desired monitoring operation. Although currently available transmitters can have a battery life of up to 4 weeks, they rarely stay attached to the bat for longer than 2 weeks.
- The sample size (e.g. the number of bats tracked or roosts found) is generally small due to the cost of buying tags and then following the animals, limiting the potential inference capability of studies.
- The individuals or age and sex classes selected for tracking may not represent the typical behaviour of 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.

10.4 Choosing the correct tracking device

A wide range of tiny transmitters are available that are suitable for tracking New Zealand bats. Most people in New Zealand have been using transmitters from Holohil Systems due to their small size and proven reliability and robustness. However, use is not restricted to this brand, with other brands available including Advanced Telemetry Systems and Lotek.

Additionally, 1-g GPS tags are also becoming available although, at the time of writing, these had yet to be used on New Zealand bats.

To choose the right transmitter or GPS tag, you should consider the weight of the tag relative to the weight of the bat, as well as the reliability and robustness of the transmitter/tag. Five basic principles will influence the choice.

1. Transmitters/tags should be as small and light as possible and **must not** weigh more than 5–10% of the bat’s weight so that the bat does not have to carry a heavy load.

2. The handling times of bats is a key ethical consideration in tracking studies – as few bats as possible should be caught and handled to achieve the project’s aims.

3. Transmitters/tags should be reliable and remain attached for as long as possible to justify catching bats in the first place.

4. The transmitters/tags should be optimum for the period you need to track bats for – for example, if you only need to track a bat for 3–7 days rather than 10–30 days, you should use one of the lighter transmitters that is available.

5. Transmitters should have relatively long antenna lengths (c. 160 mm) for maximum range relative to transmitter size.

The standard Holohil transmitters that are used to track adult long-tailed bats and adult and juvenile lesser short-tailed bats are 0.62 g (model BD2 with aerial length 160 mm).

Standard Holohil transmitters have a battery life of c. 28 days, while the standard life of the transmitter is estimated at 21 days.

Generally, researchers aim to use transmitters/tags that are c. 5% of the bat's body mass and never above the generally accepted threshold of 10% (Aldridge and Brigham 1988). Long-tailed bats are capable of carrying weights up to 80% of their body weight when carrying their young to new roosts each night (O'Donnell et al. 1995), and long-term studies of the survival of long-tailed bats using transmitters weighing 5–7% of their body mass have showed no ill effects on survival, breeding success, movements or roosting behaviour (e.g. O'Donnell 2000b, 2001, 2002a, 2002b; O'Donnell et al. 2017).

Flying juvenile long-tailed bats that are < 2 weeks old can be lighter than 8 g and should not be tracked unless using lighter transmitters because they have poor flight abilities.

10.4.1 Pros and cons of smaller transmitters

Radio transmitter producers make even smaller transmitters (< 0.19 g) that are suitable for tiny bats and even insects. These transmitters may be suitable for use on lighter juvenile long-tailed bats but they only last for very short periods (typically 3–7 days) and generally have a very short range, increasing the risk of not being able to detect the bat again. Trials in New Zealand have also shown that smaller transmitters are groomed off the bats after a few days, necessitating the capture and tracking of many more individuals**.**

10.4.2 Ordering radio transmitters

Transmitters should be ordered several months in advance to avoid delays, with delivery arranged 1–2 weeks before the start of the project. Test all transmitters when they arrive to make sure that none of the magnets have moved and accidentally switched a transmitter on during shipping. This will avoid the battery draining without you knowing. Store transmitters in a cool, dark place (Holohil recommends the refrigerator). Unused or recovered transmitters can be refurbished by the manufacturer at reduced cost.

When ordering radio transmitters, request:

- a flat package (so it is does not sit too high on the bat's back)
- deactivation by magnet if possible (which makes it very easy to start the transmitter by removing the magnet)
- an aerial base that is reinforced with c. 1 cm heat shrink to strengthen it (otherwise the bats will bite off the antenna)
- a receiver frequency range of 160.1 MHz (channel 00) to 160.6 MHz (channel 47) for New Zealand conditions.
  - Note that the use of frequencies from 160.6 to 161.11 MHz (channels 48–99) requires an additional licence (contact the Banding Office at bandingoffice@doc.govt.nz). Frequencies of 173–174 MHz can also be used; however, TR4 receivers do not work for these frequencies. It is also 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.

Before ordering transmitters, check to see if any other radio-tracking studies are being conducted in your study area. If so, it will be important that you order transmitters with different frequencies from those already in use – it can be very frustrating if you end up tracking the wrong animal.

10.5 Attaching transmitters/tags

Transmitters/tags are attached between the scapulae (shoulder blades) on the upper back of the bat (Fig. 76).

Before attachment, the fur is usually trimmed over an area the size of the transmitter to increase the length of time the transmitter/tag will remain attached. However, recent trials in the Eglinton Valley have indicated that tags remain attached for similar periods on trimmed and untrimmed long-tailed bats (CFJ O'Donnell, DOC, pers. obs).[6]

The transmitter/tag is glued onto the fur so that the bat does not need to carry a bulky harness and to ensure that it will fall off. This also allows the transmitter to be recovered and reused once the battery has been replaced.

Transmitters/tags can be attached using a variety of glue types. Hospital-grade skin adhesives or those used for veterinary purposes ('Torbot' or Uro-Bond® V equivalents and 'Sauer') are best, but the availability of such glues varies (and their brand names) change frequently. **Do not** use 'Super Glue' variants because they are carcinogenic, and the use of contact adhesive glue F2® (Ados Chemical Co, Auckland, New Zealand) is no longer recommended.

If trimming fur before attaching a tag, the procedure involves:

1. Sit the bat on a flat surface (Fig. 77) and hold its forearms firmly to immobilise it. Short-tailed bats wriggle a lot, so make sure your grip is tight without squeezing the bat too hard. It helps to place the bat flat on a holding bag and fold one end of the bag over the head of the bat to calm it down. Sitting the bat across the outer curve of your thigh helps to arch the bat's back and makes it easier to trim the fur.
2. Trim the fur with very sharp scissors or electric nose trimmers. The trimmed area should be no larger than the area of the transmitter and the trimmed fur should be a couple of millimetres long.
3. Use a cotton bud to push surrounding fur away from the trimmed area and then use the cotton bud to apply a thin coating of glue to the trimmed area as well as the underside of the transmitter.
4. Follow the specific instructions on the glue's container to determine how long to hold the transmitter before attaching to the bat, then carefully place the transmitter on the back of the bat and release. If the fur is left untrimmed, ensure that it is wrapped around the transmitter when attached. Transmitters usually fall off after 2 weeks (maximum recorded = 28 days), although they may fall off earlier if the animals are moulting.

[6] It would be useful to trial attaching transmitters to untrimmed bats at other sites and comparing the duration of attachment in trimmed and untrimmed bats.

Fig. 76. Photograph of a lesser short-tailed bat (*Mystacina tuberculata*) with fur trimmed to the correct length.
Photo: J Sedgeley

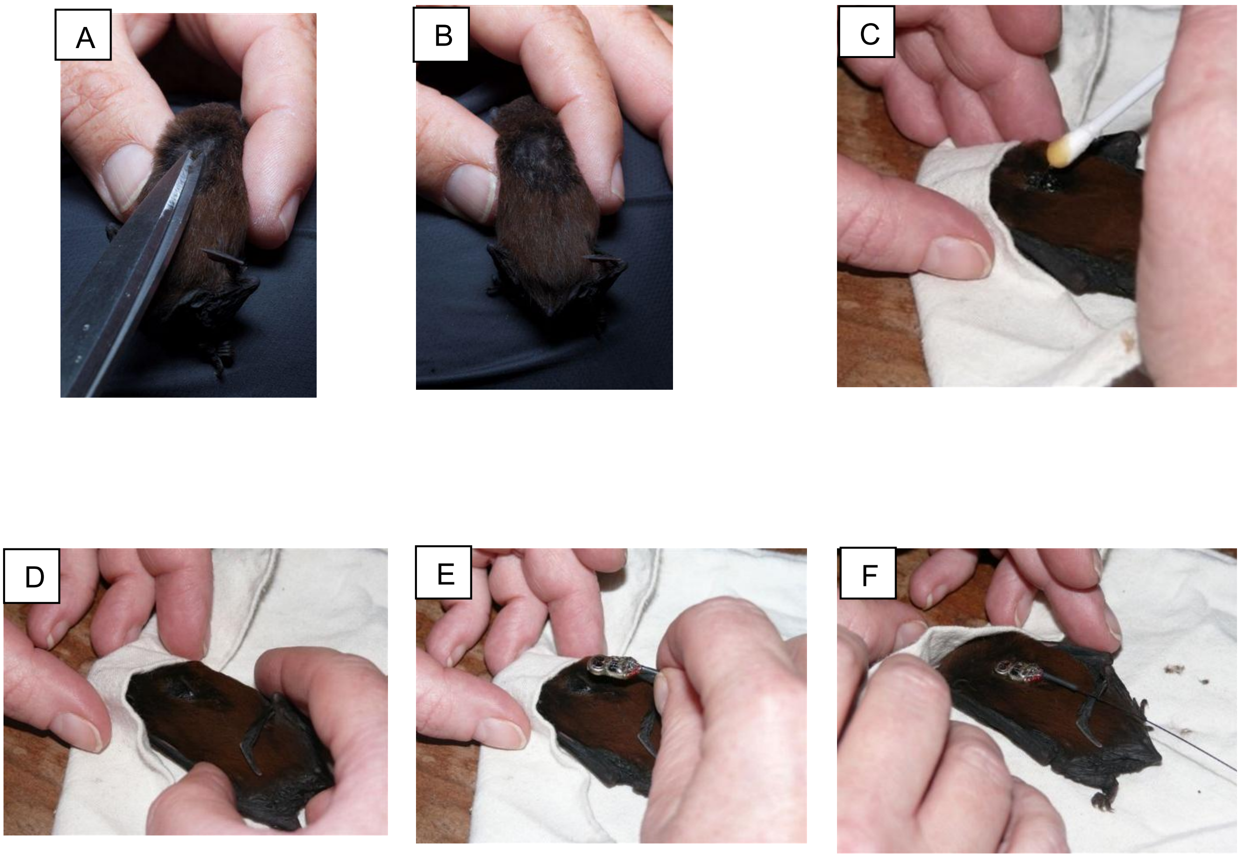


Fig. 77. Procedure for attaching a radio-transmitter to a long-tailed bat (*Chalinolobus tuberculatus*). *Photos: (A) S Bernert; (B–F) J Sedgeley*

10.6 Recording the locations of tagged bats

If using radio transmitters, the signals of bats are picked up using radio receivers attached to antennae tuned to 160.1–161.11 or 173–174 MHz (see Ordering radio transmitters).

A wide range of receiver models are available, all of which function well. The most commonly used receivers are hand-held TR4 receivers (Telonics, Mesa, Arizona, USA) and Lotek Ultra receivers (Lotek, Havelock North, New Zealand) with hand-held, three-element Yagi aerials (Fig. 78).

Bats can also be tracked by vehicle using TR4 or scanner-receivers (a variety of models are available), as well as vehicle-mounted omnidirectional aerials.

Three-element Yagi aerials are directional, so following the direction of the loudest signal will get you moving towards the bat. It can be a bit of an art form to learn the many tricks for interpreting signal strength and direction, and learning how to radio track has to be done in the field – there is no substitute for getting training in a real situation. However, the following training modules are available on DOC's Intranet (see Appendix 1).

- Use of radio transmitter frequencies
- Ground-based radio tracking protocol
- "Sky ranger" radio tracking system

Two training modules are also available in DOCLearn (see Appendix 1).

- Animal Radio Tracking
- Radio Direction Finding

Aerial tracking can be a good option for picking up the initial signals if the bat roosts being monitored are spread over a large area. This can be done using a fixed-wing aeroplane (with specially-fitted external aerials) or a helicopter. If you are doing this, then it is important to locate the bat roosts on the ground on the same day, otherwise the radio-tagged bats may move overnight. The person undertaking the aerial tracking in the aircraft pinpoints a GPS location of the transmitter signals from the air, and then teams of people on the ground go in to locate the actual roost trees. Try to get the plane up as early as you can in the day to allow the teams on the ground as much daylight time as possible to track in to the roost trees. Note that to undertake aerial tracking, you will require training or assistance from an experienced person.

Aerial tracking is not essential if the bat population roosts are in a small, known area. In this case, go to the known roost area on the ground and try to pick up the transmitter signals – trying near known roost clusters and from high points is best.

Finding long-tailed 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 arrive at the roost in time, you will see bats entering the roost cavity, which will save time later when you go to set up a bat trap or carry out an emergence count.

Another option for locating a roost entrance is to use a thermal camera that can pick up the heat signature of an active roost entrance and sometimes provides information on the general numbers of bats using the roost (< 10, 10–50, > 50). This requires the outside temperature of the tree to be significantly lower than the temperature of the roost, so it is better to do this earlier in the morning.

Other options for locating the roost include climbing the tree with a radio-receiver (following DOC's *Tree Climbing SOP* [see Appendix 1]) or, alternatively, firing a line over a branch (using a slingshot and nylon line) to allow a TR4 (without a Yagi aerial) to be pulled up the tree, noting when the signal is loudest and close to a cavity entrance (note that this does not work using a Lotek Ultra).

The Holohil BD2 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. A bat that is high in a roost tree on slightly or moderately sloping terrain can typically be heard 1–1.5 km away.

Fig. 78. Tracking a bat with a three-element Yagi aerial. *Photo: S Bernert*

10.7 Home range analysis

When undertaking any tracking study, it is best practice to consider the objectives of the study at the outset and the range of analysis techniques intended to be used so that bats are not caught and tagged unnecessarily.

There is a very wide range of options for analysing home range data (the point location and habitat data you may collect), and analyses can be implemented in a range of software packages, including GIS platforms. Good general references for analysis techniques include White and Garrott (1990), Kenward (1987) and Amelon et al. (2009), while examples of home range analyses can be found in

O'Donnell (2001) for long-tailed bats and O'Donnell et al. (1999) and Christie (2003) for lesser short-tailed bats.

11. Taking tissue samples for genetic purposes

Tissue sampling for bats is covered by the *Tissue Sampling for Bats SOP* (see Appendix 1).

The purpose of this section is to describe this SOP, which aims to:

- provide guidance to DOC staff or independent science providers commissioned by DOC who do not work for an institution that has an AEC and are planning to collect scientific tissue samples from native bats (long-tailed and lesser short-tailed bats) in the field
- provide a formal mechanism for ensuring that any impacts on manipulated animals are minimised during tissue sampling
- 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 Handling, examining, measuring and releasing bats section in this best practice manual.

Operators must seek separate AEC approval for any project involving the manipulation of animals outside routine DOC species management practices.

11.1 Reasons for taking tissue samples

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
- the genetic effects of population bottlenecks
- taxonomic statuses and relationships, and evolutionary ecology
- parentage
- the level of hybridisation
- the sex assignment of individuals (e.g. juveniles for translocation, adults lacking sexual dimorphism)
- metapopulation dynamics.

11.2 When should a tissue sampling project proceed?

A tissue sampling project can only proceed if:

- there is a 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.

11.3 Preparations for undertaking tissue sampling

The tissue sampling for bats SOP contains key information to assist project managers and trained operators in:

- identifying the need for tissue sampling
- seeking separate AEC approval where appropriate
- planning the project safely and appropriately
- ensuring that relevant experience is accredited to the operator
- collecting tissue samples from native bats, with animal welfare being paramount
- storing and transporting samples safely.

Users of the SOP **must** note the following.

- **DOC staff:** The 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 that 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.

11.4 When to take tissue samples

Bat workers should avoid handling bats at times of the year when potentially negative impacts have been identified. The impacts of handling and sampling can include compromising the condition of:

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

Because bats enter torpor more often during winter, it is necessary to sample them during the breeding season. Therefore, care should be taken to ensure that the bats are processed as quickly as possible so that lactating females can resume parental care.

Bats should also 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 the rain and wind (e.g. tent fly, tarpaulin).

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

11.5 Handling time

For each species, it is important that operators:

- minimise individual handling times
- know how individuals are going to respond to prolonged handling and manipulation.

Wherever possible, operators **must** ensure that multiple captures are housed appropriately while individuals are being sampled. Bats can be held in cloth holding bags that are hung securely at a low height. To prevent overcrowding and aggression, the recommended maximum number of bats in a bag is 10 for long-tailed bats and 5 for short-tailed bats.

11.6 Training requirements

The Bat Recovery Group Leader **must** approve all tissue sampling trainers. Refer to Permitting, ethics approval and training.

Specialised training **must** be undertaken 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.

11.7 Occupational health and safety requirements

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

11.8 Tissue sampling protocol

Step 1 – Assemble the team

The team **must** include at least one experienced handler, who will usually be responsible for:

- restraining the bat firmly and confidently to prevent any movement during tissue collection (Fig. 79)
- stretching out the wing to its full capacity to provide a taut surface for the biopsy punch (see Step 6).

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

Step 2 – Prevent further captures

It is essential that the trapping device (e.g. mist net, harp trap) is closed or removed immediately after the bats are removed from it to prevent the capture of excessive numbers of bats if not enough personnel are present to attend to them.

Failure to do this could severely compromise the safety of any excess bats trapped, as well as 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.

Step 3 – Prepare the equipment

Before commencing sampling:

- ensure that all equipment is laid out ready and close to hand
- pre-fill vials with 70% ethanol and keep several of these 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 that the hard surface the sampling will take place on 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 place it 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
- ensure that surplus cotton buds, biopsy punches and sterile tweezers can be easily accessed.

Step 4 – Restrain the bat

Handlers can make restraint during tissue sampling easier and minimise stress to the bat by:

- holding the bat steady with its back against the prepared hard surface
- informing the operator if the bat is about to struggle
- processing the bat as quickly and efficiently as possible.

For lesser short-tailed bats, the handler should wear a polypropylene or other suitable glove on the non-dominant hand to avoid being bitten. If the bat does happen to bite the handler, the handler **must** ensure that 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.

Step 5 – Check the suitability of the bat for tissue sampling and collect any other data required

Before taking a tissue sample:

- identify the bat (check the band number or PIT tag) and check that a tissue sample is required (e.g. the bat has not been sampled before – note that previous punches heal quickly and are difficult to recognise)
- assess the condition of the bat – tissue samples should not be taken from bats that are in poor condition (e.g. less than 85% of the mean weight for the species), mothers with young attached to the nipple, heavily pregnant females or juveniles in their first 4 weeks of flight
- perform any other necessary manipulations (e.g. morphometric measurements).

Step 6 – Take a wing biopsy

Handler

1. Hold the bat face up against the sterilised hard surface with the non-dominant hand (Fig. 79).

2. Use the dominant hand to stretch the wing out to its full capacity, parallel and as close as possible to the hard surface.

Operator

3. Remove the protective cap from the biopsy punch and unscrew the lid from a prepared ethanol vial.
4. Press the biopsy punch firmly down on the main part of the wing, between the fifth finger and the body, avoiding major blood vessels. The wing should be completely flat, and the punch completely vertical.
5. Twist the punch gently 360 degrees to both the left and right, ensuring that 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.
6. Carefully remove the biopsy punch from the wing. The sample will either be lodged in the punch or stuck on the cutting board. In case of the latter, the handler **must** be very careful to avoid moving the sample as they remove the bat from the sampling area.

    - **If the sample remains in the 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 70% ethanol can be flushed through the punch with a pipette to dislodge the sample. Once the sample is in the vial, sterilise the punch by shaking it again in the ethanol vial.

    - **If the sample remains on the 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.

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 requirement.

Fig. 79. Taking a tissue sample from a long-tailed bat (*Chalinolobus tuberculatus*). *Photo: J Sedgeley*

11.9 Hygiene

Basic hygiene measures should be taken to prevent the spread of infectious diseases and parasites between bat individuals and populations. These include:

- washing hands or wiping hands with medical hand wipes (if no water is available) between bats handled/sampled
- discarding and replacing holding bags as soon as they become soiled (e.g. with 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 **must** also be sterilised.

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

- placing cold or torpid bats in a cloth handling bag and keeping them 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
- having a bat detector or light switched on to help confirm that the bat has flown away rather than fallen to the ground – but do not shine the light directly into the bat's face.

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

11.10 Healing times

Wing holes are typically fully healed within 3–4 weeks, without impairment to flight or reproductive success, at which time they 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.

11.11 Labelling samples

It is vital that all tissue samples are labelled with key information that relates back to the electronic database, including the:

- individual ID (e.g. PIT tag, band number)
- date of collection.

Other useful information can include the:

- colony group
- wing (left or right)
- geographic location of the collection site.

Vials should be labelled with sticky labels that wrap around the entire vial to ensure that 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 when they come in contact with alcohol.

11.12 Storing samples

Samples should be kept in 70% ethanol and refrigerated. If samples are being sent overseas, the International Air Transport Association provides guidelines for shipping dangerous substances, including alcohol (which is 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 information about the:

- species
- number of samples
- quantity and strength of ethanol
- commercial value of the samples.

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

12. Guidelines for temporarily keeping bats in captivity for research purposes

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

12.1 Permitting requirements

In addition to normal permitting requirements:

- AEC approval **must** be obtained to keep bats in captivity for research purposes
- applicants **must** demonstrate a willingness and ability to keep bats in captivity using the captivity guidelines below
- a full report of the captivity procedures, including the number of bats kept, the number of bats released, husbandry techniques (amount of food, housing), health, any deaths and causes of death **must** be provided at the end of the work
- a report summarising the preliminary research findings should be provided at the end of the fieldwork, and copies of final research findings and any published results should be provided at a later date.

12.2 Important considerations

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

12.3 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. However, they **must** be housed in more spacious holding boxes and exercised if they are to be kept any longer than this. If bats need to be kept for more than 3 days, they **must** be held in a free-flight enclosure (see Requirements for keeping bats in longer-term temporary captivity).

12.3.1 Keeping bats in cloth bags

Note that bags and gloves must always be thoroughly washed and disinfected between different study areas or if they have been used previously to hold other species. Bags and gloves should be washed using a disinfectant detergent (e.g. Sterigene) and rinsed thoroughly. This can be done in a washing machine.

Aim to keep bats in bags for as short a time as possible. A bat may be kept in a bag overnight (which almost always means the next day as well) provided that 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 that the bats do not become too hot or dehydrated. The bottom of each bag should be moistened to provide humidity, and the bats **must** be offered food and water during this period (see Food and water).

It is preferable to keep only one bat in each bag to ensure that it gets enough food and to avoid any aggression. However, two bats could be kept together in a larger bag.

It is acceptable to have more than one bat in a bag just prior to release. In fact, this often makes release easier because the bats will warm each other up and will be more likely to be active and ready to fly.

12.3.2 Keeping bats in wooden cages

Note that holding boxes must be carefully disinfected between use by birds and bats.

If a bat is to be held for more than 1 night, it **must** be kept in a specially designed holding box. The box **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 the bats to grip while roosting. Soft material can be attached to the walls so that the bats can choose to roost under the cloth.

Store the box in a quiet location with an appropriate ambient temperature. For most research purposes, it is easier to keep bats warm and active at temperatures ranging from 25 to 28°C. In very hot conditions, it may be necessary to provide extra humidity by placing a dampened cloth or paper on the floor of 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. 80). 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).

Figure 81 provides an example of a holding box design that is commonly used in the UK and is recommended in the UK *Bat Workers' Manual* (Mitchell-Jones and McLeish 2004).

Fig. 80. Bird transfer box adapted for use by bats. Each compartment contains a small bowl of water and a separate bowl with mealworms. *Photo: J Sedgeley*

We have kept up to five bats in each section of the bird transfer boxes shown in Fig. 80, but we have only held that many bats for 1 night. We do not recommend holding more than three bats per section for longer periods. We have also kept same sex bats together and mixed males and females together. However, when mixing the sexes, we would try to keep the sex ratio balanced or have more females than males.

It is important to note that whilst we had no problems keeping bats under the conditions described above, this 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 and water).

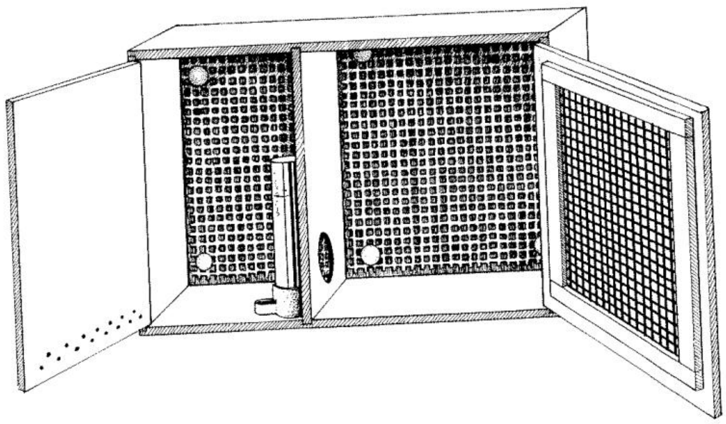


Fig. 81. Bat holding box design that is commonly used in the UK. Note that water is provided using a water-feeder that is used for feeding captive birds. *Illustration: ©TP McOwat, reproduced with permission from the Bat Workers' Manual, © JNCC 2004, ISBN 1861075588*

12.3.3 Food and water

Captive bats **must** be provided with food and water every day, even if they are only being held for 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 from a damp cotton wool bud or using a pipette / eye dropper. When offering water by hand, ensure that the bat is held with its feet higher than its head to prevent the bat from accidentally aspiring water into its lungs. Also note that long-tailed bats will usually need to be fed mealworms by hand.

Bats will only eat if they are active, so they should ideally be kept at temperatures of 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 part of their natural 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. However, if the bat is being held inside a bag, it will have to be offered water by hand using a damp cotton wool bud, a small spoon or a pipette / eye dropper.

Long-tailed bats are aerial insectivores, feeding primarily on moths and other flying insects. Consequently, they do not recognise mealworms as natural food and will 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.

The following guidelines should be followed when hand feeding and providing water to a captive bat.

- Remove the bat from 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 that the bat is held with its feet higher than its head to prevent the bat from accidentally aspiring water or fluids into its lungs.
- Water can be offered using a damp cotton wool bud, small spoon or pipette / eye dropper.
- When offering a mealworm, first 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.
- Once fed or watered, the bat should be replaced inside its bag or holding box and the container put in a warm place (so the bat stays active to digest the food).
- Live mealworms should also be placed inside the container being 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 then be made of the number offered so it is possible to see if the bats have learnt to feed themselves.

We found that 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 and 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.

Bats that are being kept for up to 7 days can be fed on ordinary commercial mealworms. However, if they are being kept for longer, they will require vitamin and mineral supplements. This is most easily achieved by feeding the bats nutrient-enriched mealworms that have been taken from their bran medium and transferred into a container of Wombaroo® Insectivore Rearing Mix, where they are held for 1 day before being fed to the bats.

12.4 Requirements for keeping bats in longer-term temporary captivity

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

Bats can rapidly lose condition if kept in captivity with no exercise, so if they are to be kept for longer than 3 days, they should be allowed free flight daily where possible e.g. in the free-flight enclosure at Knobs Flat (Fig. 82) where roost boxes allow bats to exercise at night. Food and water dishes need to be provided (large Pyrex® or glass baking dishes work well). The enclosure can comfortably hold 30 to 40 bats at a time, but this many bats should be provided with at least two or three roost boxes and several food and water dishes to avoid competition. Where free flight aviaries are not an option, bats could be exercised in a room and then recaptured to return them to their holding boxes, but it is likely that this will cause them to become stressed.

Fig. 82.   The free-flight enclosure located in the forest behind Knobs Flat Field Station. This enclosure is c. 10 m long × 5 m wide × 2 m high, making it large enough for the bats to fly around inside. It contains vegetation to provide bats with a natural environment and a choice of several roost boxes. Feeding and water dishes need to be supplied. *Photo: J Sedgeley*

12.5 Monitoring weight and health

Bats **must** be weighed daily if being held in short-term captivity and weekly if being held in longer-term captivity. The bats should also be checked for signs of ill health (e.g. lethargic, dull sunken eyes) or injuries (e.g. bites, wing tears). If bats are to be kept for longer than 3 days, it is recommended that an experienced bat keeper or vet is present.

Weights should be carefully monitored and the 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 (see Food and water).

Sometimes an individual bat will lose weight or fail to put on weight at the same rate as other bats being held with it, in which case it may be necessary to isolate this bat from others to ensure that it gets enough food.

For temporary captivity, it generally seems best to keep bats warm and active and make food freely available. However, bats will sometimes put on large amounts of weight, particularly if they have had little exercise, in which case it may be necessary to cut back the food, although this has not generally been a problem for lesser short-tailed bats being held in bags or boxes for a few days or free-flight enclosures for up to a month.

12.6 Limiting research procedures

To reduce stress on the bats, limit the number of research procedures performed on each bat to only one per day.

12.7 Release

Bats should be released as close to the 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, 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. This can be achieved 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.

12.8 Feedback and reporting

This manual provides general guidelines for keeping bats in captivity. However, feeding and exercise requirements will vary among individuals and species and at different times of the year.

It is important, therefore, to carefully document the exact procedures that were used, including the number of bats that were kept, how they were housed, the amount of food and exercise they were given, 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.

12.9 Other useful information

Useful information on first aid care can be found on the DOC website Resources for bat workers: Bats/pekapeka (doc.govt.nz) and in DOC's *Wildlife Health Management SOP* (see Appendix 1).

13. Permitting, ethics approval and training

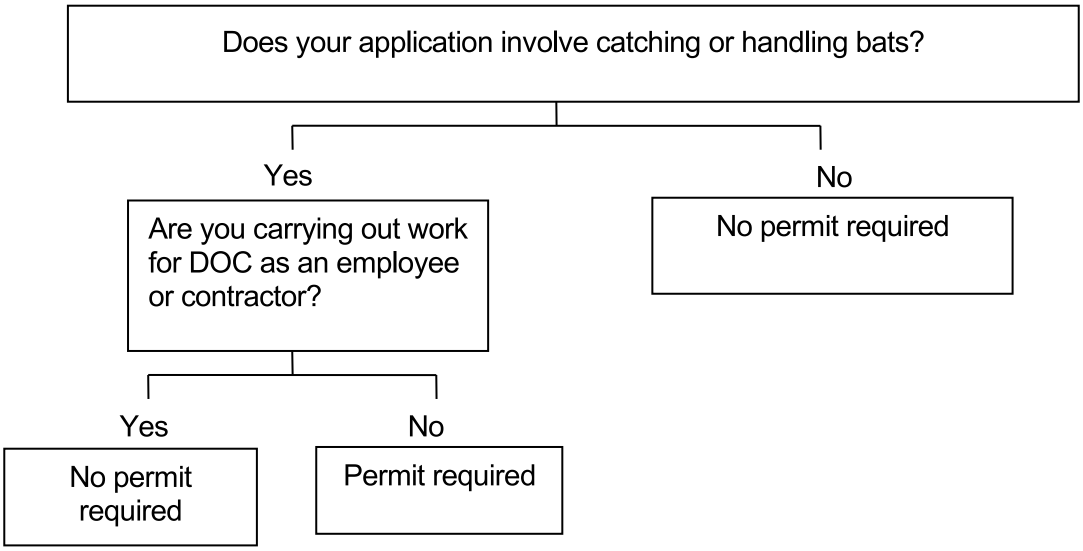
13.1 Permitting

13.1.1 Do you require a permit?

All New Zealand bats are protected under the Wildlife Act, which makes it an offence to trap, capture, pursue, molest or disturb them.[7] Therefore, if your proposal involves catching or handling bats, authorisation (i.e. a permit) from the Director-General of DOC is required.

DOC employees carrying out DOC work do not require permits. This is because DOC employees who are 'acting in good faith in pursuance of your duty' are carrying out the work of DOC in accordance with its functions and with the powers of the Minister of Conservation and Director-General of DOC. However, they **must** hold the appropriate level of competency as per *Bat Handling Competencies* (see Appendix 1).

The decision tree below will assist you in determining if you require a permit.



13.1.2 Role of the Bat Recovery Group in permitting

The Bat Recovery Group is an advisory group, not a permitting authority. If required, it can provide advice to decision-makers on applications under the Conservation Act 1987 and the Wildlife Act to manipulate bats. Generally, the Bat Recovery Group will support proposals if the proposed study/project will:

- enhance knowledge of bats
- benefit bat conservation

[7] Section 65 of the Wildlife Act makes it an offence to hunt or kill wildlife. By definition, in section 2 of the Act, hunting or killing includes trapping, capturing, pursuing, disturbing or molesting wildlife and also includes any attempts to do so.

- not unduly impact on bats or have a low likely impact
- use only approved techniques (Animal Ethics Committee and this manual).

13.2 Animal ethics approvals

13.2.1 DOC's obligations under the Animal Welfare Act 1999

The DOC 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.

All proposed projects except those using the standard manipulation techniques covered in this manual are subject to scrutiny and approval by 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
- consider and determine, under section 84(1)(a) of the Animal Welfare Act, applications for the approval of projects
- set, vary and revoke conditions of project approvals
- monitor compliance with conditions of project approvals
- monitor animal management practices and facilities to ensure compliance with the terms of the *Code of Ethical Conduct for the use of animals for research, testing and teaching* (see Appendix 1)
- consider and determine applications for the renewal of project approvals
- suspend or revoke project approvals, where necessary
- recommend to the Director-General of DOC amendments to the Code of Ethical Conduct.

Section 80 of the Animal Welfare Act, 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, as it provides guidance on the circumstances under which animals can be manipulated. Such guidance is particularly important for the AEC when considering project proposals. The following principles apply.

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.

13.2.2 Applying for AEC approval for bat work

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

Bat workers within DOC **must** follow DOC's procedures for applying for AEC approval – details can be found on the 'Animal Ethics Committee (AEC)' page on the DOC Intranet (see Appendix 1).

Where external bat researchers are applying for permits under the Wildlife Act or Conservation Act, DOC **must** ensure that they have appropriate ethics approvals if necessary. Examples include approvals for banding bats, inserting transponders and tissue sampling.

13.2.3 Do you need AEC approval?

The Ministry for Primary Industries (MPI) has produced a generic flow chart to assess whether AEC approval is required. However, if bat workers are uncertain whether they need to apply, the DOC AEC strongly recommends that they approach the current chair of the AEC via aec@doc.govt.nz.

13.2.4 How to apply to the AEC

Proposals require identification of the species and numbers of animals to be used, the nature of the manipulations, justification for the use of animals, 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 so they can be easily understood by an intelligent lay person. For further guidance, read 'How to apply for AEC approval' on the DOC Intranet (see Appendix 1).

13.2.5 AEC application deadlines

The DOC AEC usually meets on the first Wednesday of every month and applications for consideration by the committee **must** be submitted by the 20th of the preceding month, with the exception of December (the AEC does not meet in January). However, check the 'Do I need Animal Ethics Committee approval?' page of the DOC Intranet (see Appendix 1) for scheduled meeting dates and submission deadlines for the upcoming year. Approval times for AEC applications can take between 1 and 3 months, depending on the complexity of the research and quality of the application.

13.2.6 Application form

A link to the DOC AEC application template for manipulating live animals is available in Appendix 1.

13.3 Training and competencies

13.3.1 Acquiring training and demonstrating competencies

The purpose of this section is to outline the ethical standards required to be registered as a competent, authorised bat worker by the New Zealand Bat Recovery Group.

Certification by the Bat Recovery Group is required for any permits that require the handling of bats (for Wildlife Act authorisations). Both species of New Zealand bats are threatened and individuals are small and delicate, making them vulnerable to injury if handled incorrectly. Therefore, anyone that handles them **must** have levels of competency that ensure they are handled ethically. A competent handler will know how to catch, hold and release bats appropriately, understand if a bat is in torpor or not and adjust their handling appropriately, and know when and how to attach monitoring devices. Bat workers can reach a level of competency in up to 25 skills (listed below), each of which is represented by a separate competency. Details of the skill requirements are outlined in this best practice manual.

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 them during their project, as this approach may better allow the necessary skills to be targeted.

13.3.2 List of potential trainers

A list of the names and contact details of people with the skills necessary for training in certain techniques is available from bathandler@doc.govt.nz. Note, however, that it should not be assumed that these people will always be willing or available to offer training.

13.3.3 Definitions and registration processes

- **Registered Bat Trainee:** A person who has registered with the Bat Recovery Group as a Trainee.
- **Bat Banding Trainee:** A person who has registered with the New Zealand Banding Office as a Level 1 Bat Bander.
- **Trainee Log:** A logbook of all training sessions undertaken, with each session signed off by an Authorised Trainer. Logbooks are available from bathandler@doc.govt.nz, from the Resources for bat workers page on the DOC website or from DOC's internal file management system, docCM (see *Bat Competency Training Sheet*, Appendix 1).
- **Competent Bat Worker:** A person who has been certified as 'Competent' in a particular skill by the Bat Recovery Group.
- **Authorised Trainer:** A person who is registered as competent in a particular skill AND has been authorised by the Bat Recovery Group to teach and supervise Registered Bat Trainees in that skill (but only if they are working under an existing Research or Collection Permit and Wildlife Act Authority or if they are a DOC Trainer). The Trainer **must** be present for all training and inspect all competency activities.

13.3.4 Training

- While people are designated as Trainees, training **must** occur under the direct supervision of an Authorised Trainer (see above).
- Once one or more competencies have been signed off by the Bat Recovery Group, the Competent Bat Worker can work independently with respect to the skill(s) (if they have the appropriate permits).
- The Trainee will keep a logbook that describes their experience in each competency (refer to *Bat Competency Training Sheet* [see Appendix 1]). This needs to be signed by one or more Authorised Trainers.
- The Trainee **must** have read and understood this best practice manual.
- Trainees can be certified in either individual competencies or multiple competencies. Like bird banding in New Zealand, it is envisioned that it may take several years for most Trainees to achieve all competencies because opportunities for hands-on bat work are limited.

13.3.5 Application for certification in a competency

- When Trainees reach the target handling levels described under each competency, they may apply to the Bat Recovery Group, via bathandler@doc.govt.nz, for certification in that competency. However, reaching the target level does not automatically give the applicant certification and an application for certification **must** be accompanied by a letter of endorsement in writing from at least one Authorised Trainer.
- Applicants can apply for certification for single or multiple competencies.
- Applications **must** include a short summary of the applicant's bat handling experience, copies of signed training logs, and the names of two Authorised Trainers who can attest to the applicant's competency.
- Applications will be reviewed by the Bat Recovery Group at its monthly meeting.
- Applicants will receive confirmation of certification from the Bat Recovery Group within 2 months of applying.
- If certification for competency in banding long-tailed bats is being sought, then the Trainee **must** apply to the DOC Banding Office for Level 1 Bat Bander registration (bandingoffice@doc.govt.nz) after filling in the appropriate Level 1 banders form.

13.3.6 Rescinding certification in a competency

The Bat Recovery Group may rescind certification if practitioners are no longer considered competent or do not follow best practice.

13.3.7 Becoming an Authorised Trainer

- Competent Bat Workers may apply to become an Authorised Trainer in writing to the Bat Recovery Group, via bathandler@doc.govt.nz.
- Authorisation is at the discretion of the Bat Recovery Group, and discussion with the Recovery Group is recommended before applying.
- Applicants can apply for authorisation for training against single or multiple competencies.
- Applicants **must** be able to demonstrate:

a. a deep understanding and experience of the ecology of New Zealand bats
b. considerable experience well beyond competency levels in catching, handling and manipulating bats
c. a strong aptitude for, and experience in, teaching others about bats
d. a clear understanding of teaching standards
e. knowledge of the Wildlife Act and Wildlife Regulations 1955 as they apply to working with bats
f. effective communication skills, an understanding of health and safety requirements, and experience in records administration.

13.3.8 List of competencies

Bat workers can reach a level of competency in each of the following skills.

1. Catching bats
   - 1.1 Use of mist nets
     - 1.1.1 Extract, bag and store correctly a total of 30 individuals of either species
     - 1.1.2 Demonstrate correct mist net placement, set up, smooth operation, appropriate mist net attendance, assessment of risks and safe extraction and handling on 10+ different nights
   - 1.2 Use of harp traps (free standing)
     - 1.2.1 Lead identification of appropriate harp trapping sites and set up and monitor trap(s) on 10+ different nights
     - 1.2.2 Extract 10+ bats appropriately from free standing traps
     - 1.2.3 Demonstrate harp trapping protocols (animal welfare considerations, trapping in the breeding season, rain, repair and maintenance etc)
   - 1.3 Use of harp traps (at roost entrances)
     - 1.3.1 Lead set up and monitoring of trap(s) on 10+ different nights
     - 1.3.2 Extract 10+ bats appropriately from traps hoisted up trees
     - 1.3.3 Demonstrate harp trapping protocols at roost entrances (safe trapping at tree roosts (risk management), predation risks, disturbance risks, animal welfare considerations, trapping in the breeding season, rain, repair and maintenance etc)
2. Handling bats
   - 2.1 Bagging, storage, handling, measuring, weighing, sexing, aging, temporary marking and releasing appropriately:
     - 2.1.1 For long-tailed bats: 50 individuals
     - 2.1.2 For short-tailed bats: 50 individuals
   - 2.2 Banding long-tailed bats:
     - 2.2.1 50 individuals
     - 2.2.2 Demonstrate knowledge of how to remove bands safely (2 methods; demonstrate on model bat)

2.3 Pit-tagging insertion in short-tailed bats:

2.3.1 Pit-tag insertion to short-tailed bats

2.3.2 Bat handling for pit tagging

Note that that transponder skills require exacting standards and specialised training from a select few people, and if people need this skill, they should contact the Bat Recovery Group to apply to get trained.

2.4 Attaching radio transmitters (should first be competent in 2.1 and/or 2.2):

2.4.1 For long-tailed bats: watch 5 individuals having radio transmitters attached by a Competent Bat Worker or Authorised Trainer

2.4.2 For long-tailed bats: attach radio transmitters to 5 individuals correctly under supervision

2.4.3 For short-tailed bats: watch 5 individuals having radio transmitters attached

2.4.4 For short-tailed bats: attach radio transmitters to 5 individuals correctly under supervision

2.4.5 Demonstrate understanding of reasons for attaching transmitters, Animal Ethics issues (risk management and animal welfare considerations, trapping)

2.5 Taking wing biopsies

2.5.1 Watch 5 individuals having biopsies taken by a Competent Bat Worker or Authorised Trainer

2.5.2 Take biopsies from 10 individuals under supervision

2.5.3 Understand and follow the Tissue Sampling for bats SOP (available on request from the Bat Recovery Group Leader)

3. High-risk activities – Roost felling (all of these competencies include the understanding of what to do when bats are found during tree felling as per appendix 6 of *Initial Veterinary Care for New Zealand Bats* (Borkin and Shaw 2019)

3.1 Assessing roost tree use using automatic bat monitors - Demonstrate correct timing, placement and interpretation of data 10+ times according to DOC's bat roost protocols.

3.2 Undertaking roost watches / emergence counts - Undertake roost watches / emergence counts at 10+ occupied roosts where the entrance is visible.

3.3 Evaluating roost tree features - Evaluate 10+ potential roost features in trees (e.g. cavities, peeling bark, epiphytes) in at least two different forest/habitat types, including the forest/habitat type where trees are going to be assessed.

14. Initial veterinary care for New Zealand bats

Guidelines have previously been developed for situations when a bat may be injured or found on the ground (Borkin 2023). These guidelines, which are for use in the initial few days of care, provide advice for the first responder as well as the veterinarian and are current best practice.

The full guidelines, and appendix 6 of the guidelines which provides bat care advice for first responders, can be found on the DOC website.

15. Health and safety

15.1 Requirements for health and safety plans

DOC staff who are working with bats **must** ensure that they operate under approved safety plans produced through DOC's primary health and safety management tool, Risk Manager.

The *Risk Manager User Manual* (see Appendix 1) provides detailed, module-by-module instructions on how to use this web-based system, starting with a basic introduction to navigating around the system.

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

DOC safety plans are generally approved by the Operations Manager in the area where the work is being undertaken. External researchers working under the auspices of other organisations **must** have an approved safety plan from their parent institution, and the Operations Manager for the area should assess these plans when processing applications to undertake research on public conservation land or protected species.

It is important that hazards relating to bat monitoring are properly identified, assessed and controlled and are linked to the 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 are also worth reading (see Armstrong and Higgs 2002).

General hazards relating to working in the field almost always apply to working on bats. However, several hazards that are specific to bat-related work are highlighted in Table 6.

Table 6 Important hazards relating to working on bat projects

HAZARD	DESCRIPTION
Visiting bat roosts and catching bats at night in forests and/or caves	People working at night should generally work in pairs. The only exception is when undertaking bat roost counts < 200 m from the road where 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, entering caves and/or climbing limestone cliffs to find bat roosts and/or set up harp traps	Climb according to DOC's Roped tree climbing SOP (see Appendix 1). New staff **must** be trained to a standard of competency and by an authorised DOC instructor (as per the SOP), and climbing equipment should be upgraded according to the SOP and **must** be of UIAA standard. Only one person is to be climbing at a time and at least one other person who is trained in rescue techniques is to remain on the ground.
Radio tracking bats at night from a vehicle	Maintain a roster of night workers to avoid over-tiredness. Shifts should be no longer than 3 hours. The vehicle **must** have a current warrant of fitness and registration and must undergo regular servicing. Drivers **must** have appropriate driver licences and must carry a first aid kit in the vehicle.
Rigging trees with slingshots and ropes or strings for mist nets, climbing and bat traps, which may result in falling objects	Approved NZSS/UIAA standard safety helmets **must** be worn on the ground at all times and anyone using slingshots must also wear a safety visor. Correct knots and pulley systems must be used and security tested by the designated safety supervisor. All equipment must be of a high quality.
Bites received from handling bats resulting in possible infection, including a very remote chance of rabies-related lyssavirus	Follow the Bat Recovery Group guidelines (see Disease risks to human health). Restrict the number of personnel handling bats and ensure that gloves are worn at all times, except when bare hands are necessary (e.g. attaching a transmitter or extracting a bat from a mist net). A first aid kit must always be on hand, and staff who will be handling short-tailed bats without gloves should receive a rabies vaccination (and booster every 10 years).
Lithium batteries used in camera monitoring of bat roosts posing a potential risk through degassing/exploding	Do not overcharge batteries and keep them out of direct sunlight when charging. Also, make sure the batteries do not get dropped or knocked. Refer to *Lithium Battery Use and Issues* (see Appendix 1) for more information.

15.2 Disease risks to human health

15.2.1 Lyssavirus

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

What is lyssavirus?

The term 'lyssavirus' refers to a group of viruses that 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 is also present in other countries in the region where bats are found, including New Zealand. This is a relatively new virus, so the information provided below is based on its close relative, common rabies.

Contracting lyssavirus

Lyssavirus could be contracted from a bite by an infected bat or from the saliva of an infected bat if it gets into an open wound (or other orifice) of a person.

Symptoms

Symptoms are similar to those of rabies (refer to *Rabies* [see Appendix 1]), which initially causes illness with numbness and weakness of the limbs, progressing to coma and death from encephalitis.

Treatment

Immunisation against rabies can be 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 to seek medical attention immediately. Avoiding handling affected or sick bats and wearing protection such as gloves are the best precautions for avoiding risk of exposure.

According to *Initial Veterinary Care for New Zealand Bats* (Borkin 2019):

> If bitten, NSW WIRES Inc. (2018)[8] suggests gently washing the wound for 15 minutes with soap under running water. This should be a "wash and flush" approach. Do not scrub the wound, to ensure that debris is not pushed further into the wound. Betadine should be applied after washing.
>
> People who regularly handle or care for bats should be vaccinated for rabies as a precaution, although this is not currently known to be present in New Zealand (K. McInnes, Department of Conservation, pers. comm., 27 March 2019). Unless there is an incident where transmission is considered likely, rabies antibody titer levels should be checked every two years, to ensure that levels are greater than four (Dr Tania Bishop, Australia Zoo, pers. comm.).

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[8] New South Wales Wildlife Information, Rescue and Education Service Inc. 2018. The Bat Manual: identification, rescue, rehabilitation and release. An unpublished manual developed by the WIRES Bat Management Team for WIRES, Brookvale, NSW 2100, Australia. 334 p.

15.2.2 SARS-CoV-2 (bat–human transmission)

There is no evidence of SARS, SARS-CoV-2 or SARS-CoV-2-like viruses being present in either New Zealand or Australian species of bats. There is also no evidence to suggest that bats or other wildlife in these countries pose a risk of SARS-CoV-2 (the causal agent of COVID-19) infection in humans. Nevertheless, the Bat Recovery Group makes the recommendations outlined below in relation to all disease risk.

15.2.3 Recommendations from the Bat Recovery Group

Bat workers in New Zealand should take the following precautions when handling bats, as is currently advised in Europe, America and now Australia.

- All bats that appear to be sick **must** be handled with gloves. If gloves are not available, then bats should not be handled.
- The bodies of sick or dead bats should be necropsied and the results added to the national database, as per DOC's *Wildlife Health Management SOP* (see Appendix 1).
- The number of workers handling bats on a particular project should be minimised.
- Workers handling short-tailed bats (which bite often) should wear gloves whenever possible.
- Where glove wearing is impractical (due to the difficulty of handling bats with gloved hands), hands **must** be disinfected/sanitised regularly.
- Workers handling short-tailed bats without gloves should obtain rabies vaccinations, and get a booster every 10 years.
- 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.

15.2.4 Further information

- *Infectious Diseases Information System* (see Appendix 1)
- *Rabies* (see Appendix 1)
- Australian bat lyssavirus
- Coronaviruses in Australian bats
- SARS-CoV-2 and precautions when in contact with Australian wildlife
- Bat Conservation InternationalBat Conservation International

15.3 Disease risks to bats

There has been limited research on diseases in New Zealand bats. The research undertaken up to 2003 was summarised by Duignan et al. (2003), who did not identify any significant disease issues at that time (see Borkin [2019] for a summary). However, an absence of records does not mean that there is no risk of diseases being transmitted among populations of bats. Therefore, a precautionary approach should be taken. DOC's requirements relating to wildlife health are set out in the *Wildlife Health Management SOP* (see Appendix 1).

15.3.1 SARS-CoV-2 (human–bat transmission)

The IUCN bat specialist group recognises a low but credible risk of human-to-bat transmission of SARS-CoV-2. Consequently, an assessment of the level of risk that a project poses to bats **must** be carried out before undertaking any fieldwork that involves catching bats, and fieldwork activities should be modified based on that risk assessment if necessary. See the risk assessment method recommended by the IUCN bat specialist group in July 2021 and check for up-to-date guidance from this group and the Bat Recovery Group.

16. Biosecurity incursions

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

16.1 Accidental importations of bats

Six exotic species of bats from three microchiropteran families have arrived dead in New Zealand as stowaways in cargo. Three of these species were in family Vespertilionidae: a Japanese pipistrelle (*Pipistrellus javanicus abramus*) arrived in a cargo of car parts (Daniel and Yoshiyuki 1982), an Australian lesser long-eared bat (*Nyctophilus geoffroyi*) arrived in a cargo of timber (Daniel and Williams 1984) and an Australian little forest bat (*Vespadelus vulturnus*) was found in a crate of aircraft parts (O'Donnell 1998). Additionally, in 2002, a small unidentified bat belonging to family Molossidae was transported in a shipment of bananas from Ecuador and found in a Queenstown shop; in October 2004, a dog-faced fruit bat (*Cynopterus brachyotis*; family Pteropidae), which is 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; and in June 2005, another molossid, the wrinkle-lipped free-tailed bat (*Tadarida plicata*), was found dead in Papakura in a vehicle imported from Thailand (CFJ O'Donnell, DOC, pers. obs.).

16.2 Risks

Exotic bats may pose potential threats to the native fauna and people in New Zealand, particularly through disease risk. For example, Pteropidae are known to carry the rabies-related lyssavirus, which can be fatal to humans and other bats. In an overseas study, antibodies to Nipah virus (family Paramyxoviridae), which caused disease in pigs and humans in peninsular Malaysia in 1998–99, were raised in *Cynopterus brachyotis* (Johara et al. 2001), the same bat species that was found in Queenstown in 2004.

So far, all interceptions have been of dead bats, and there is only a slight risk of disease transmission to New Zealand from such cases. Additionally, most of the bats arriving in this country 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.

16.3 Pre-border interception

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

16.4 Post-border interception

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

For advice on procedures to follow in this situation, refer to *Reporting Procedure for Suspected New Organisms* (see Appendix 1).

Under the Biosecurity Act, every person in New Zealand has a legal obligation to report any suspected new organism as soon as practicable. Therefore, if you think you have seen a potential new bat species or disease or pest symptom:

- call the MPI Hotline on 0800 80 99 66; or
- report it through the MPI website.

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, including disease symptoms that you may not have seen before or an organism that might be new to New Zealand.

MPI has access to experts who have experience in diagnosing new species, and often they can do this over the phone with the information you provide.

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Appendix 1

Internal references for Department of Conservation Te Papa Atawhai (DOC) staff

A1.1 References to DOC's internal file management system, docCM

FILE NAME	FILE NUMBER
AEC application template	DOCDM-737250
Animal pests SOP definitions and FAQs	DOCDM-51708
Bat call identification manual for DOC's spectral bat detectors	DOC-6469402
Bat competency training sheet	DOC-6228629
Bats: counting away from roosts – automatic bat detectors	DOCDM-590733
Bats: counting away from roosts – bat detectors on line transects	DOCDM-590701
Bat handling competencies	DOC-6169671
Blank field recording sheet for transponder recapture	DOCDM-130631
Code of Ethical Conduct for the use of animals for research, testing and teaching	DOC-6130638
Collection, storage and transport of diagnostic samples from birds and reptiles	DOCDM-54407
Extracting a New Zealand long-tailed bat from a mist net	DOCDM-22907
Infectious diseases information system	DOCDM-383258
Instructions for setting up RFID readers, dataloggers and antennae	DOCDM-379889
Introduction to bat monitoring	DOCDM-590958
Lithium battery use and issues	DOC-5888747
Micro-chipping a lesser short-tailed bat	DOCDM-131439
Operational planning for animal pest operations SOP	DOCDM-1488532
Rabies	OLDDM-544362
Reporting procedure for suspected new organisms	DOCDM-290718
Risk Manager user manual	DOCDM-611915
Roped tree climbing one page SOP	DOC-1544989
Tissue sampling for bats SOP	DOCDM-929116
Transponder field recording sheet for new tags	DOCDM-130625
Use of second-generation anticoagulants policy	DOCDM-97398
Wildlife health management SOP	DOCDM-442078

To obtain a copy of these documents, please contact bathandler@doc.govt.nz.

A1.2 DOC Intranet pages

PAGE TITLE	LINK
Animal Ethics Committee (AEC)	http://intranet/natural-heritage/managing-natural-heritage/animal-ethics/
Do I need Animal Ethics Committee approval?	http://intranet/natural-heritage/managing-natural-heritage/animal-ethics/when-to-seek-approval/
Ground-based radio tracking protocol	http://intranet/natural-heritage/terrestrial/innovations-and-techniques/ground-based-radio-tracking-protocol/
How to apply for AEC approval	http://intranet/natural-heritage/managing-natural-heritage/animal-ethics/how-to-apply-for-aec-approval/
"Sky ranger" radio tracking system	http://intranet/natural-heritage/terrestrial/innovations-and-techniques/sky-ranger-radio-tracking/
Use of radio transmitter frequencies	http://intranet/natural-heritage/terrestrial/innovations-and-techniques/use-of-radio-transmitter-frequencies-in-nz/

A1.3 DOCLearn modules

NAME	LINK
Animal Radio Tracking	DOCLearn
Radio Direction Finding	DOCLearn