Herpetofauna: funnel trapping

Version 1.0

This specification was prepared by Kelly M. Hare in 2012.

Contents

Inventory and monitoring toolbox: herpetofauna

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Department of Conservation
Te Papa Atawhai

Synopsis

Overseas, funnel trapping is a common method for monitoring and inventory of herpetofauna (e.g. Vogt 1947; Greenberg et al. 1994), and although still in its infancy, is becoming more common in New Zealand (e.g. Gebauer 2009; Bell 2010). Few studies using funnel trapping in New Zealand have been published, so the efficacy of this method for capture of native herpetofauna and its usefulness for providing data for analyses have yet to be confirmed. However, based on those published studies from New Zealand coupled with data from overseas studies, funnel traps are likely to be an effective method for obtaining data for distribution, inventory, indices of abundance, density estimates, population trends, site occupancy and estimates of survival.

Many types of funnel traps exist, from home-made mesh design (e.g. Fitch 1951) through to modified fish traps (e.g. g-minnow traps; Bell 2010). All funnel traps consist of a cylinder with an inverted funnel in either one (single-ended) or both (double-ended) openings (Greenberg et al. 1994). They work by animals climbing through the funnel opening and not being able to find the opening again. Funnel traps are useful for capture of terrestrial and arboreal lizards. Funnel traps are the most effective method for *trapping* geckos to date (other methods are also useful for sampling, e.g. systematic searches and pitfall trapping)—geckos can escape from artificial retreats and pitfall traps. Funnel traps can also be used to capture amphibians, but often result in desiccation of individuals (Fitch 1951) and are not recommended for use on amphibians in New Zealand. Funnel traps are not useful for tuatara (the traps are too small for adults); tuatara and frogs can be effectively surveyed using systematic searches and are not discussed further in this method.

Funnel trapping is similar to other types of trapping, such as pitfall trapping, and as such has similar assumptions, advantages and disadvantages. Pitfall and funnel trapping often give complementary results; that is, some species will be more easily captured in pitfall traps whereas others will be more easily captured in funnel traps (e.g. Greenberg et al. 1994; Christiansen & Vandewalle 2000). Therefore, the choice of trap type(s) will depend on the habitat type, target species and project goals. Some additional advantages of funnel traps (over and above that of pitfall traps) is that they can be set into habitats that pitfall traps cannot effectively sample (e.g. on vegetation, debris-dams and rocky screes), and can also effectively trap geckos. However, funnel traps also capture small birds, mammals and insects (Fitch 1951), therefore thought needs to be given to trap placement and potential predators of lizards (e.g. weasels, rodents, native centipedes, etc.) when installing traps. Furthermore, traps may be opened by larger mammals (e.g. dogs; Fitch 1951), which although a bigger risk overseas, may be a factor in some areas of New Zealand. Trapping in mammal-dense areas can be dangerous for lizards and is not encouraged.

As some species are captured more effectively using pitfall traps than funnel traps (e.g. common skinks (*Oligosoma polychroma*) and McCann's skinks (*O. maccanni*; M. Lettink, pers. comm.), the efficacy of funnel traps for capture of the species of interest should be determined using pilot studies. Funnel traps cannot be used for total counts as some individuals will not be caught in traps; total counts are not discussed further within this method.

Funnel traps are relatively non-destructive to the habitat (Gebauer 2009). Observer bias is low for funnel trapping and (as with pitfall trapping) relates only to the ability of the observer to identify species and handle herpetofauna effectively (Towns 1991). Funnel traps require less time to install than pitfall traps, but some designs can be more time-intensive during trap-checks (e.g. g-minnow traps that open in the centre need to be moved to open and remove animals and/or change bait). The total area used for trapping, sample sizes and sampling methodology undertaken (e.g. including mark-recapture v. without mark-recapture) will depend on the aims of the project and type of data being collected, as well as the species' behaviour and habitat type.

Many factors can influence the capture probability and quality of the data obtained from funnel trapping. For example, capture probabilities vary widely among species (Greenberg et al. 1994). Additionally, capture probabilities of one species may vary among sites and microhabitats (Jamieson & Neilson 2007; Barr 2009; Gebauer 2009), and are strongly influenced by additional environmental factors such as weather and temperature (Fitch 1951). Other factors that may influence capture rate include age and sex of an individual, trap placement, and previous encounters with traps, as well as behaviour of the species and individuals (Christiansen & Vandewalle 2000). Even though funnel traps usually have an attractant included (bait), the placement of the trap opening is important as the target species needs to be able to find, or may avoid, the opening (Christiansen & Vandewalle 2000). Trap placement is very important for some species (e.g. chevron skinks, *Oligosoma homalonotum*; Jamieson & Neilson 2007; Barr 2009) and pilot studies should be used to determine best trapping microhabitats.

As animals are trapped and cannot escape until removed by the observer, ethical considerations are higher than for methods such as systematic searches. For example, protection against the sun is necessary for all traps and daily checking of traps is required in most situations (see 'Full details of technique and best practice'). Furthermore, as any traps left in the field will continue to trap animals it is vital that all traps are removed at the end of a trapping session (use GPS and flagging tape to pinpoint traps). Funnel trapping requires capture and subsequent handling of the animals and therefore requires some skill with herpetofauna. As animals are captured, marking of individuals and/or photo-resight identification methods can be easily employed. However, note that some species are very common and ID patterning from photo-resight will not be a suitable method of identification for monitoring. The design of trap-grids, placement of traps and effort put into checking traps depend on the aims of the study and these aims should be well developed and thought through before funnel traps are deployed. See 'Case study A' for examples of potential uses of funnel traps.

Assumptions

- The target species is/are catchable using funnel traps.
- All habitats are equal in their probability to be trapped, *or* habitat type is included as a factor in the analysis.
- The species of interest are truly absent from the area when none are detected.
- The sample area(s) is/are representative of the wider population(s).
- All observers have equal ability to identify and handle animals.

- All factors that can be controlled (e.g. the time of day that traps are checked) will be controlled, and those that cannot (e.g. temperature) will be recorded and used in analyses where appropriate.

A range of analytical methods can be used in conjunction with funnel trapping. Therefore, additional assumptions may apply depending on the capture technique employed and the aims of the study.

Advantages

- The only method currently available for *trapping* geckos.
- Easily repeatable between studies and over time. Traps can be removed temporarily, making it easy to repeat trapping efforts over long periods of time with minimal disturbance to the habitat. Therefore, funnel trapping is especially useful for long-term studies.
- Relatively non-destructive to the habitat.
- Usually easy to conduct.
- May be sufficient to describe basic biological patterns (e.g. distribution, abundance, etc.).
- Many factors affecting detectability can be controlled by standardisation of techniques (e.g. season, time of day, observer and species).
- Depending on data type being collected, or if data is collected in a standardised manner, then generally this method requires little statistical background.

A range of analytical methods can be used in conjunction with funnel trapping. Therefore, additional advantages may apply depending on the capture technique employed and the aims of the study.

Disadvantages

- Some funnel trap designs are expensive (e.g. g-minnow)
- May require a large time investment to initially install a trapping grid.
- Trap placement will affect capture probability.
- Traps can also capture potentially dangerous or at-risk ‘by-catch’. For example, funnel traps can effectively trap amphibians (which may desiccate), invertebrates (such as the native centipede, which may prey on trapped lizards), small birds (which may die from fright) and mammals (such as weasels and mice, which may prey on trapped lizards). Note: traps are only useful where mammals are negligible (e.g. mammal-free offshore islands) or where effective mammal-control is undertaken (and backed up with tracking-tunnel index cards).
- Many species will not be caught reliably in funnel traps (e.g. semi-fossorial species) and capture probabilities may vary widely among species.
- Not useful for tuatara or frogs.
- It may be possible that even within a species capture probabilities are not equivalent in different habitats or within the same habitat over time.
- Capture probabilities may vary substantially among individuals due to behavioural differences (termed: ‘trap-shy’ or ‘trap-happy’).
- Capture probabilities of herpetofauna vary with temperature and other weather variables. Although many factors affecting capture probabilities can be controlled by standardisation of

techniques (e.g. season, time of day) many factors (e.g. daily temperature fluctuations) cannot be standardised and must be measured and factored into analyses.

A range of analytical methods can be used in conjunction with funnel trapping. Therefore, additional disadvantages may apply depending on the capture technique employed and the aims of the study (e.g. observer variability when mark-recapture is used).

Suitability for inventory

Funnel trapping can confirm presence, but not absence, of herpetofauna and is suitable for inventory (e.g. Gebauer 2009). Funnel trapping is relatively expensive to set up (i.e. purchasing traps) but is no more expensive to run than other trapping methods (e.g. pitfall trapping), and is also strongly dependent on species' biology and habitat use (see 'Full details of technique and best practice').

Suitability for monitoring

Funnel trapping is generally suitable for monitoring of terrestrial and arboreal lizards. Funnel trapping is also dependent on biology and habitat use of the species as well as weather. Funnel trapping can be used with or without long-term marking and can be used in the following situations for monitoring:

- Species with some sort of individual identification (e.g. paint spots for short-term monitoring and ID patterning for long-term monitoring).
- Species without long-term marks where general changes in gross-abundance over long time periods (e.g. between seasons and years) are required. Note: without long-term marking other important data (e.g. longevity) cannot be collected.

Mark-recapture of permanently marked individuals (or using natural ID marks) is the most robust technique for long-term monitoring for population structure and survival of herpetofauna, provided all assumptions can be met and sufficient resources are available for a long-term study. This method is particularly powerful when counts are repeated annually over relatively long time frames (> 10 years), and when variation in observers, season and environmental conditions are minimised. Unfortunately, controlling for observer effects and changes in habitat are difficult under long-term monitoring scenarios. Short-term monitoring is often confounded by seasonal changes and other disruptions, but may be suitable to determine habitat use and population structure.

Skills

DOC staff may capture, handle and measure herpetofauna without permits, and tail-tipping (for genetic sampling) is covered by a standard operating procedure ('Sampling avian blood and feathers, and reptilian tissue (SOP)'—docdm-531081). Funnel trapping requires the following skills and training:

- Species identification
- Experience with handling reptiles
- Experience with funnel trapping
- Physical ability to install a trap
- Ability to write clear and thorough notes
- Proficiency using Microsoft Excel or other statistical software
- Basic understanding of statistics

See 'Full details of technique and best practice' section for more in-depth details.

Resources

Standard equipment for all funnel trapping techniques includes:

- One or more skilled worker(s)
- Datasheets/notebooks and pencils
- GPS
- Flagging tape to mark the start of the grid
- Traps
- Bait (e.g. pear or cat food for most species)

Useful, although not always essential items include:

- Covers (in open and sunny habitats)
- Moistened sponge (for dry habitats)
- Digital camera
- Temporary holding bags for captured animals (thin cloth bags are good for reptiles)
- A large pillowcase (for opening traps within)
- Non-toxic permanent markers for individually marking reptiles
- Animal ethics permits for some techniques (e.g. PIT tagging of larger lizards)
- Wildlife Act Permit if exporting samples overseas (e.g. for genetic analyses)

Hand sanitiser is also a good idea when handling lizards as some may have *Salmonella* (Middleton et al. 2010); transferral of *Salmonella* within and between species as well as to/from the handler may also be possible.

Minimum attributes

Consistent measurement and recording of the essential attributes is critical for the implementation of the method. Other optional attributes may be recorded depending on your study objective (see 'Full details of technique and best practice'). It is necessary for novices to obtain training from an expert herpetologist.

Essential attributes

At a minimum, the following should be documented:

- DOC staff must complete a ‘Standard inventory and monitoring project plan’ (docdm-146272).
- For all herpetofauna, New Zealand Amphibian/Reptile Distribution Scheme (ARDS) cards should be completed and forwarded to the Herpetofauna Administrator (address shown on ARDS card; Fig. 1).[1] Thorough, tidy and clear data entry is vital.

At a minimum, the following data should be recorded:

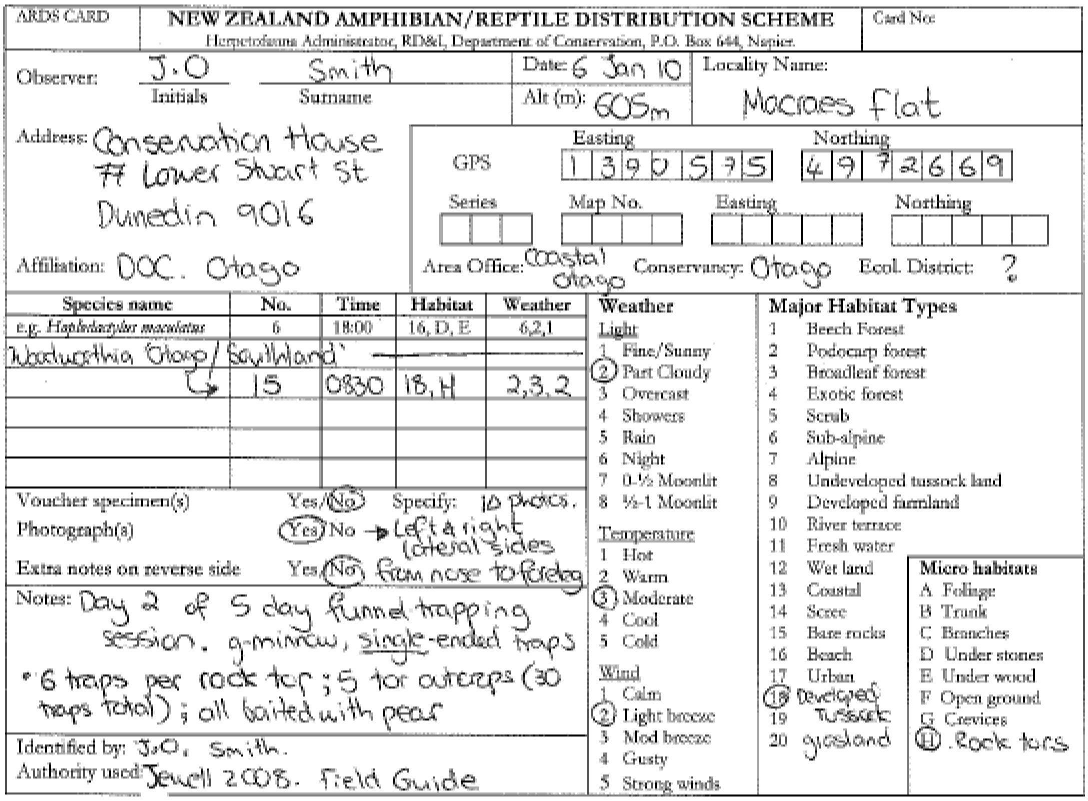
- Observer and/or recorder
- Date and time
- Location name/grid reference
- Trap number
- Weather/temperature data (either collect local temperatures during the study or get nearby weather station data after the fact)
- Number of lizards of each species present in each funnel trap

Optional attributes

Based on the specific goals of each project, other factors may be required. Additional attributes that may be useful to record while in the field, but are generally not required for funnel trapping, include:

- *Habitat characteristics*: location description, altitude, aspect, vegetation (including dominant plant species), available cover, temperature of substrate.
- *Weather characteristics*: ambient air temperature (shade, 1 m from ground), relative humidity, overnight minimum temperature, daytime maximum temperature, precipitation, cloud cover, wind direction and strength.
- *Individual morphological measurements*: snout-vent length (SVL; mm), vent–tail length (mm, include regeneration), mass (g) and records of natural toe-loss.
- *Sex*: the sex of individuals is also a useful parameter to record—see ‘Full details of technique and best practice’ for more detail.
- *Reproductive status of females*: see ‘Full details of technique and best practice’ for more detail.
- *Parasites*: see ‘Full details of technique and best practice’ for more details.

[1] The ARDS card is available online: http://www.doc.govt.nz/conservation/native-animals/reptiles-and-frogs/reptiles-and-frogs-distribution-information/species-sightings-and-data-management/report-a-sighting/



ARDS CARD | NEW ZEALAND AMPHIBIAN/REPTILE DISTRIBUTION SCHEME | Card No:

Herpetofauna Administrator, RD&I, Department of Conservation, P.O. Box 644, Napier.

Observer: J.O (Initials) Smith (Surname)

Date: 6 Jan 10

Alt (m): 605m

Locality Name: Macraes Flat

Address: Conservation House, 77 Lower Stuart St, Dunedin 9016

GPS Easting 1390595 Northing 4972669

Series | Map No. | Easting | Northing

Affiliation: DOC. Otago

Area Office: Coastal Otago Conservancy: Otago Ecol. District: ?

Species name	No.	Time	Habitat	Weather
e.g. *Hoplodactylus maculatus*	6	18:00	16, D, E	6,2,1
Woodworthia 'Otago/Southland'	15	0830	18, H	2,3,2

Voucher specimen(s) Yes/No Specify: 10 photos.

Photograph(s) Yes/No → Left & right lateral sides

Extra notes on reverse side Yes/No from nose to forelegs

Notes: Day 2 of 5 day funnel trapping session. g-minnow, single-ended traps
• 6 traps per rock tor; 5 for outcrops (30 traps total); all baited with pear

Identified by: J.O. Smith.

Authority used: Jewell 2008. Field Guide

Weather

Light: 1 Fine/Sunny; 2 Part Cloudy; 3 Overcast; 4 Showers; 5 Rain; 6 Night; 7 0-½ Moonlit; 8 ½-1 Moonlit

Temperature: 1 Hot; 2 Warm; 3 Moderate; 4 Cool; 5 Cold

Wind: 1 Calm; 2 Light breeze; 3 Mod breeze; 4 Gusty; 5 Strong winds

Major Habitat Types

1 Beech Forest; 2 Podocarp forest; 3 Broadleaf forest; 4 Exotic forest; 5 Scrub; 6 Sub-alpine; 7 Alpine; 8 Undeveloped tussock land; 9 Developed farmland; 10 River terrace; 11 Fresh water; 12 Wet land; 13 Coastal; 14 Scree; 15 Bare rocks; 16 Beach; 17 Urban; 18 Developed tussock grassland; 19; 20

Micro habitats

A Foliage; B Trunk; C Branches; D Under stones; E Under wood; F Open ground; G Crevices; H Rock tors

Figure 1. Example of how to fill in a New Zealand Amphibian/Reptile Distribution Scheme (ARDS) card. Note that either a GPS location or a map series number is sufficient. Also, try not to leave blank spaces—instead leave an indication that those data were not available or collected. If further notes are collected these can be included under 'Notes', and continue on the back of the page if necessary.

Data storage

The following instructions should be followed when storing data obtained from this method. Forward copies of completed survey sheets to the survey administrator, or enter data into an appropriate spreadsheet as soon as possible. For all herpetofauna, ARDS cards should be completed and forwarded to the Herpetofauna Administrator (address shown on ARDS card; Fig. 1).[2]

Collate, consolidate and store survey information securely, as soon as possible, and preferably immediately on return from the field. The key steps here are data entry, storage and maintenance for later analysis, followed by copying and data backup for security. Summarise the results in a spreadsheet or equivalent. Arrange data as 'column variables'—i.e. arrange data from each field site on the data sheet (date, time, location, plot designation, number seen, identity, etc.) in columns, with each row representing the occasion on which a given survey plot was sampled. See Fig. 2 for an example.

If data storage is designed well at the outset, it will make the job of analysis and interpretation much easier. Before storing data, check for missing information and errors, and ensure metadata (i.e.

[2] The ARDS card is available online: http://www.doc.govt.nz/conservation/native-animals/reptiles-and-frogs/reptiles-and-frogs-distribution-information/species-sightings-and-data-management/report-a-sighting/

weather description, habitat description, date, time, search effort, etc.) are recorded (see 'Herpetofauna: indices of abundance'—docdm-493179 for further details).

Storage tools can be either manual or electronic systems (or both, preferably). They will usually be summary sheets, other physical filing systems, or electronic spreadsheets and databases. Use appropriate file formats such as .xls, .txt, .dbf or specific analysis software formats. Copy and/or backup all data, whether electronic, data sheets, metadata or site access descriptions, preferably offline if the primary storage location is part of a networked system. Store the copy at a separate location for security purposes.

date	time	line	trap	observer	bait	re-bait	O. homalonotum	O. oratum	D.pacificus	Notes
02/02/05	8:30	1	1a	j.blogg	sardine	catfood	0	1	0	
02/02/05	8:35	1	1b	j.blogg	catfood	sardine	0	0	0	
02/02/05	8:40	1	1c	j.blogg	sardine	catfood	1	0	0	female Oh1
02/02/05	8:45	1	1d	j.blogg	catfood	sardine	0	0	0	
02/02/05	8:50	1	1e	j.blogg	sardine	catfood	1	0	0	male Oh2
02/02/05	8:30	2	2a	j.doe	catfood	sardine	0	0	1	large pregnant Pacific gecko
02/02/05	8:35	2	2b	j.doe	sardine	catfood	0	0	0	
02/02/05	8:40	2	2c	j.doe	catfood	sardine	0	0	0	
02/02/05	8:45	2	2d	j.doe	sardine	catfood	0	0	0	
02/02/05	8:50	2	2e	j.doe	catfood	sardine	0	0	1	
03/02/05	8:30	1	1a	j.doe	catfood	catfood	0	0	0	
03/02/05	8:35	1	1b	j.doe	sardine	catfood	0	0	0	
03/02/05	8:40	1	1c	j.doe	catfood	sardine	1	0	0	recap Oh1
03/02/05	8:45	1	1d	j.doe	sardine	sardine	0	0	1	
03/02/05	8:50	1	1e	j.doe	catfood	catfood	0	0	0	
03/02/05	8:30	2	2a	j.blogg	sardine	catfood	0	0	1	probably same gecko as yesterday
03/02/05	8:35	2	2b	j.blogg	catfood	sardine	0	0	0	
03/02/05	8:40	2	2c	j.blogg	sardine	sardine	0	0	0	
03/02/05	8:45	2	2d	j.blogg	catfood	catfood	0	0	0	
03/02/05	8:50	2	2e	j.blogg	sardine	catfood	0	0	0	

Notes: *Funnel trapping on Great Barrier Island for capture of chevron skinks (* **Oligosoma homalonotum***)*

All lizards ***released at point of capture***

trapping from 01/02/05 to 05/02/05; checked every morning; other lizard species also recorded.

habitat characteristics (incl. grid reference etc) and weather&min/max temperatures each day are in other spread sheets

Figure 2. Example of good data entry of field data collected from funnel trapping. Note that the data are arranged in columns, the column titles have enough detail that anyone reading the spreadsheet at a later date will know what data are included, and the notes section is used to record other interesting facts. More or fewer columns can be added as required.

Analysis, interpretation and reporting

The types of analyses possible with data collected from funnel trapping include distribution, inventory, indices of abundance, density estimates, population trends, site occupancy and estimates of survival. Analytical protocols are not covered in this section. More complete analytical protocols are under development. However, as a minimum it is advisable to:

- Seek statistical advice from a biometrician or suitably experienced person prior to undertaking any analysis.

- Report results in a timely manner. This would usually be within a year of the data collection.

Case study A

Case study A: trapping techniques for small-scaled skinks

Synopsis

Gebauer (2009) compared the usefulness of funnel trapping and systematic searches (coupled with capture using noosing) for capture of small-scaled skinks (*Oligosoma microlepis*). Small-scaled skinks live in small areas on greywacke rock piles and rocky screes (Whitaker 1991) and in this habitat pitfall traps are impractical because they will: a) cause large disturbance to the habitat, and/or b) result in bias to lizards living on the edges of rock piles. Site surveys were conducted at 13 sites, of which two were chosen to assess the potential use of funnel traps as capture techniques, and one for noosing. Trapping was conducted at each site using four single-funnel traps baited with either live flies or chicken-flavoured cat food. Bait type and time of day did not influence trap attractiveness, but smaller animals were more likely to be captured within traps in the afternoon than morning. Both funnel trapping and noosing were successful techniques for capturing small-scaled skinks, but noosing at easily accessible rock-piles provided more skinks in a shorter time-period. Gebauer (2009) also investigated photo-identification techniques for mark-resight and population estimates, but as these are outside the scope of this method they are not discussed further here.

Objectives

- Conduct site surveys to confirm reported sightings of small-scaled skink populations
- Assess the potential of funnel traps and systematic searches (with capture by noosing) for population studies of small-scaled skinks

Sampling design and methods

Site surveys

Between January and March 2008 an initial survey was conducted at 13 potential sites. Each site was photographed and then surveyed by two observers once (in the morning) during weather conditions that give highest detection probabilities for small-scaled skinks (Whitaker 1991, Teal 2006). Surveys were conducted using systematic searches from a distance of 2 to 10 m until small-scaled skinks were seen, or up to a maximum of 1.5 hours if no skinks were seen. When no skinks were observed, sites were searched for droppings and skins and re-visited at least one more time.

Capture using funnel traps

Two sites (Quarry and Huts) were chosen to assess the effectiveness of funnel traps and noosing for capture of small-scaled skinks. Trapping was conducted 14–18 February 2008 at Quarry Site, and 18–21 February at Huts site. The Quarry Site (2 plots; 12 m^2 and 40 m^2) was chosen as a high density site and the Huts Site (1 plot; 36 m^2) as a low density site. At the Quarry Site, two single-ended funnel traps were placed at the periphery of each plot, with the funnel opening facing towards the centre of the plot; the four traps were similarly placed at the Huts Site at a later date (note: the number of traps used here in this pilot study would be considered too small for most studies and analytical methods).

The single-ended funnel traps were made from strong wire mesh lined with fly-screen and shaped into a flat oval tube (21.5 × 17.5 × 8.5 cm; l × w × h). One side of the tube was fitted with a funnel fashioned of fly-screen (opening = 1.5 cm diameter), and the other end was covered with fly-screen mesh held in place by wire for easy access. The wire construction and flattened shape allowed easy placement in the environment, good ventilation and yet was sturdy enough to be covered with rocks to blend into the environment and provide shade (Fig. 3).

Traps were set at 7 am and baited with either five live flies or chicken-flavoured cat food. Bait type was alternated among traps to prevent position of traps from biasing results. The traps were checked at 1 pm and 7 pm, meaning that each trapping session was 6 hours in duration and spanned either the morning or afternoon.

Capture using nooses

Noosing was conducted at Plot 2 of Quarry Site from 20–21 February. Plot 2 was chosen as it has a large population of small-scaled skinks. A noose was made of fishing line and fitted to the end of 1.5-m long fishing pole. Both observers looked for skinks, and when one was spied, one observer sat next to the rock pile and carefully slipped the noose over the head of a skink, pulling the line tight. The skink was then lifted to the second observer who removed the noose and dropped the skink into a bag.

Other factors measured

For all skinks that were captured, an individual number was written on the back with a gold, non-toxic, water-based pen. Skinks also had measures of snout-vent length, tail-length and tail-regeneration length taken along with ID-photographs. Skinks were then released at the point of capture. Some site characteristics were also measured, including:

- Overall area of site and gross vegetation type surrounding sites (e.g. grazed pasture)
- Distance (m) of plots from nearest other greywacke scree

Figure 3. Single-ended funnel trap set on the edge of a greywacke scree slope. The trap is fashioned from wire mesh fly-screen and held together with wire and duct-tape. The rear of this trap (no photo available) can be opened without moving the trap, but care should be taken as lizards may dart out past the observer. The arrow points to the trap opening (photo: Konstanze Gebauer).

Results

Site surveys

Populations of small-scaled skinks were located at 10 of the 13 sites surveyed. No skinks or droppings were found at two sites. One survey at a new location provided one small-scaled skink at 1175 m a.s.l.—the most southerly and highest altitude population of small-scaled skinks found to date.

Capture by funnel traps and noosing

Analyses were conducted on data from the 6-hour trapping sessions. A total of 14 skinks (one recapture) were captured over 46 trapping sessions at the Quarry Site plots, and four skinks (one recapture) over 26 trapping sessions at the Huts Site (Table 1). Capture rate (skinks/trapping session) did not differ significantly between the Quarry and Huts Sites (0.3 and 0.2 skinks/trapping session respectively, $P > 0.05$). More skinks were captured in traps baited with flies ($n = 15$) than in traps baited with cat food ($n = 5$) ($\chi_1^2 = 4.51$, $P < 0.05$). Skinks were not more likely to be captured in the morning ($n = 8$) than the afternoon ($n = 12$) ($\chi_1^2 = 0.25$, $P > 0.05$).

Six small-scaled skinks were captured by noosing over two 2-hour periods by a two-person team (CPUE = 0.75 skinks/person-hour). Skinks at the Quarry and Huts Sites did not differ in snout-vent length (t_{17} = 0.627; P > 0.05), nor was there a difference in overall size of skinks for capture technique (trapping v. noosing; P > 0.05), or bait type (P = 0.135). However, smaller skinks were captured more frequently in the afternoon v. morning trapping sessions (t_{15} = 2.627; P = 0.02).

Table 1. Funnel-trap capture-recapture data for small-scaled skinks (*Oligosoma microlepis*) during February 2008.

Site/plot	Days of trapping	Traps	Trapping sessions (trap nights)	Skinks (recaptures)
Quarry Site—plot 1	4	2	16 (8)	5 (0)
Quarry Site—plot 2	7.5	2	30 (15)	9 (1)
Huts Site	4	4	26 (13)	4 (1)

Limitations and points to consider

Limitations:

- Due to time and financial constraints only four funnel traps were available. This resulted in very small sample sizes for skink captures, and will limit the analyses and interpretation of the data. For example, recapture rate was low; therefore population level analyses could not be undertaken. See 'Full details of technique and best practice' for discussion of trap number and placement, etc.

Points to consider:

- Although systematic searches were undertaken during good skink-catching weather, it was unclear whether the funnel traps were similarly set only during good skink-catching weather. If not, then weather variables should have been discussed and included in the analyses.
- Bait within traps was alternated among traps, and changed regularly. This allowed for the assumption that traps were equally likely to capture lizards, and also provided useful data on best bait type for capture of this species.
- Skink welfare was a high priority for this study. Therefore, the author expertly used rocks to shade the traps. The traps were also checked at 6-hour intervals in accordance with animal ethics protocol. While this provided useful data on whether time-of-day influenced capture probability, it likely increased disturbance at the site and may have influenced the behaviour of skinks.

References for case study A

Gebauer, K. 2009: Trapping and identification techniques for small-scaled skinks (*Oligosoma microlepis*). *Research & Development Series 318*. Wellington, Department of Conservation. 24 p.

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Full details of technique and best practice

Before undertaking any field work, explore whether animal ethics approvals and capture permits are required. All herpetofauna in New Zealand are fully protected by the Wildlife Act (except the introduced skink, *Lampropholis delicata*), and you may need permits and ethics approvals if you are going to manipulate a reptile or amphibian. DOC staff may capture, handle and measure herpetofauna without permits, and tail-tipping (for genetic sampling) is covered by a standard operating procedure ('Sampling avian blood and feathers, and reptilian tissue (SOP)'—docdm-531081). However, DOC staff will require animal ethics permits for other techniques (e.g. PIT tagging). A Wildlife Act Permit will be required if exporting samples overseas (e.g. for genetic analyses).

The techniques outlined below can be used for funnel trapping to locate and/or capture terrestrial and arboreal lizards, but remember that traps are less effective at capturing semi-fossorial lizards, and some species will be easier to catch in other trap types or by using other methods. Funnel traps are not presently recommended for capturing native and exotic frogs and are not useful for tuatara. If you have no experience with funnel trapping, at a minimum you should consult with someone who has experience prior to conducting field work; however, field training from an experienced person is more appropriate. Subtle details will be important for maximising captures and ensuring the well-being of captured animals. Also provided below are techniques to identify sex of individuals and determine reproductive status of adult females. The following is only an overview to provide a general idea of the practical considerations and the implementation of the techniques. Pairing up with an experienced person is critical to the success of your project.

Sampling design and effort

When trapping is conducted in a standardised way, it will improve the robustness of the data and reduce errors, especially when sampling is conducted over different years by different researchers. Therefore, sampling effort and design for funnel trapping should be similar to that of pitfall trapping. In some cases, funnel traps are used simply as an inventory tool; trap placement will still be important to maximise detection, but sampling effort and design is less crucial than for monitoring.

For good trapping design, trapping areas must be defined prior to setting out traps. The size and shape of the trapping area will depend on the goals of the study, the habitat, and the target species. However, traps are generally set out in grids or lines within a study area. Because the entire grid/line (rather than an individual trap) is usually the sampling unit, there must be enough grids/lines for useful statistical analyses to be undertaken. Sometimes, when placed far enough apart to be considered true replicates, individual traps are used as the sampling unit (e.g. pitfall trapping; Lettink & Seddon 2007) with the captures per trap for a given time period calculated. At a minimum, three grids/lines should be employed where calculating relative abundance over time is the goal, but five or more is better for statistical analyses. When the goal is to compare relative abundance between sites (e.g. two or more habitat types or two or more management sites), at least two grids/lines per habitat/treatment are required. The design of the grid will depend on the goals of the study, and as many traps per grid/line that are feasible with the given finances, habitat, time and number of observers should be employed. A pilot study, followed by an appropriate power analysis (e.g. Taylor & Gerrodette 1993) will assist with determining the design trade-offs and number of traps required. Depending on the goals of the study, traps may be set at distances of 1 m up to 10–20-m apart (e.g. pitfall trapping; Towns 1975; Spurr & Powlesland 2000; Lettink et al. 2011). For example, inventory may have traps 20-m apart, whereas population monitoring may need spacing of 2 m to improve recapture rates (traps must be within the individual’s home range). For most types of data, trapping at each site should be conducted over multiple capture occasions, and one capture occasion is a trap night. Further, funnel trapping of herpetofauna is typically presented as number of captures/100 trap nights for comparisons over time and as captures/trap/time-period for comparisons among sites.

The number of trap nights will depend on the objective of the study, but trapping sessions are generally 5–10 days in length (e.g. pitfall trapping; Lettink et al. 2011). It is unknown whether capture rates with funnel trapping follow a similar pattern to pitfall trapping, i.e. are highest for the first night traps are set and decline steadily over time (Moseby & Read 2001).

Traps may require a placement period (‘settling in’ time), as some species are wary of areas with recent disturbance and will not enter newly set traps. Traps should be checked at least every 24 hours, or in accordance with ethics approval. Frequent checking will prevent accidental mortality from desiccation or predators (if present). When comparing sites/habitats, traps should ultimately be checked in the same time frame (i.e. within the same day), or at the very least within weeks of each other with potential confounding variables such as weather recorded for analyses and interpretation. Weather variables should be recorded for each trap night, as certain species are more active under certain weather conditions. Take photographs and draw a map of the site with grids and traps marked clearly.

Characteristics of a funnel trap

Trap type

Generally, modified double-ended gee-minnow (g-minnow) traps are used for funnel trapping of herpetofauna in New Zealand (see Fig. 4). However, g-minnow funnel traps are relatively expensive to initially purchase (Bell 2010), which can result in a small number of traps being available for a given study, unless these can be borrowed from freshwater fish teams. Funnel traps also can be fashioned from wire mesh (e.g. Fitch 1951; Gebauer 2009; Fig. 3) which will reduce costs of materials, but this may be offset by increased labour costs. If traps are home-made, it is important to remember that the mesh must be small enough so as to trap the smallest lizards, and that the opening should be large enough to admit the largest individual of the target species (but small enough to omit larger animals). Another option to reduce costs is to split the g-minnow trap in half (so that two single-ended traps are available), then the traps are effectively doubled in number. Regardless, it is preferable to have the same trap types for a study and within a plot in case trap type influences capture probabilities. Some form of cover within the trap (e.g. a layer of dead leaves) will give trapped lizards a place to hide.

Figure 4. Setting a double-ended g-minnow funnel trap on the forest floor. Note: if a lizard was present in the trap then it may easily escape when the trap is opened. To reduce the potential for losses of lizards, it may be wise to open the trap within or over a large bag (photo: Jo Hoare).

Important trap modifications and principles for lizard safety

Funnel traps are generally made of metal, which can easily heat in the sun and may cook a trapped lizard. Therefore, metal traps set in open habitats should have some form of cover which does not impede the air-flow. Trap covers could include small rocks, nestling the trap within vegetation, or custom-made sheets/planks tied down using wire or pegs. Traps in open areas which are exposed to warmer temperatures will also need a dampened sponge to provide moisture. Funnel traps have also been constructed of plastic, but these were less effective at capturing herpetofauna than their mesh counterparts (Maritz et al. 2007).

Although most lizards can swim, drowning is a serious threat for trapped lizards. Therefore, in areas close to water it is advisable to check that traps are not set where they will be inundated (e.g. by rising tides or flooding). If your traps are set in river beds or stream dams, check the weather forecast daily to anticipate potential flooding and remove the traps.

Funnel traps are also effective at trapping non-target species (Fitch 1951), and as such care should be taken in some environments. For example, native centipedes (*Cormocephalus rubriceps*) have been shown to prey on trapped chevron skinks (*O. homalonotum*; H. Jamieson, pers. comm.). Small mammals can also easily enter the traps, and will prey on trapped lizards (Fitch 1951). For example, kiore (*Rattus exulans*) prey on trapped chevron (B. Barr, pers. comm.). Therefore, in areas of high mammal abundance funnel traps may not be appropriate.

Ensure that all traps are removed at the end of the study period. A trap left out in the field will continue to attract (by means of dead animals) and therefore trap animals.

Installing and setting funnel traps

Trap installation

Set out traps in the pre-determined configuration. Label each trap by trap number. It is a good idea to use a label that also includes the grid number as this will reduce confusion at a later date. For example, trap '1c' would be trap 3 in grid 1, whereas trap '3g' would be trap 7 in grid 3. Trap numbers written on with marker pen may need re-writing periodically as they tend to rub off—melting the number in can be more long-term. Do not rely only on flagging tape to label traps and grids, as this degrades with UV light and is not reliable as a long-term marking method. Take a GPS fix of the site so that traps can be found again. As stated above, it is vital that all traps are removed at the end of a study.

Trap placement is important as it will influence your capture rate. Pilot studies should be used to determine best trapping microhabitats for each species. In general, funnel traps set on the ground are generally set a little into the substrate. For example, on the forest floor the leaf litter may be cleared away to provide a small indent and then pushed up around the trap. For traps set within rocky areas, the trap opening is generally set so that it is below some rocks (Fig. 3). Funnel traps may also be set high-up on vegetation to capture arboreal species, and in this case need to be secured firmly so they do not fall or get blown out of the bush/tree. For chevron skinks, microhabitat preferences are known (Jamieson & Neilson 2007; Barr 2009) and traps are set within debris dams. This means that the distance between traps is generally not uniform, and may be clustered (Barr 2009). For chevron skinks, traps are a third-filled with debris dam material, baited, tied to a tree, and covered in vegetation (Barr 2009).

Setting traps

After the trap is in place it can be set. Open the trap and add some debris as hiding space in the base of the trap. A piece of wet sponge will help reduce desiccation in warm and open areas. A squeeze-bottle with water can be used to remoisten dried sponges.

Bait is often used to attract animals to traps. Commonly used and especially attractive baits are tinned pear and fish-based cat food (Whitaker 1994). Chevron skinks are known to prefer banana and raspberry lures (Jamieson & Neilson 2007). Baits (other than pear and cat food) used for other species (including none) have had limited success (e.g. Perrott et al. 2011). Some species are more attracted to certain baits, but other considerations (e.g. the presence of mammalian predators, large numbers of ants, etc.) may influence your choice of bait. Placing the bait on a small piece of paper can aid with quick bait replacement, and reduce the potential for rotting fruit and meat contaminating the trap. Bait should be replaced every day to keep the attractant fresh and unspoiled, and to reduce the variables needed in analyses. Used bait should be removed from the trapping area and treated as normal rubbish/compost.

What to do with lizards in a trap

Lizards will need to all be removed *en masse* once the trap is opened. The easiest way to do this, without losing any lizards, is to open the trap over a large bag (such as a pillowcase), gently removing the lizard and placing it in another bag. The lizards can then be removed one at a time from the bag for processing. If there are lots of captives, remove more aggressive species and larger specimens first and do not hold them in bags with smaller species that could be potential prey for larger lizards (Whitaker 1994); perhaps split them up among a few smaller bags.

Hold lizards firmly (but don't be too rough) as they are relatively robust and the animal will settle more quickly in your hands if it's made to feel secure (Whitaker 1994). Do not hold lizards in your hands for too long, or leave them in the sun, as they can develop heat stress and die quickly. Be careful to avoid tail loss during capture and handling by taking care to always hold lizards by the body and never by the tail alone, especially when lizards are cold as cold lizards lose their tails more readily (Whitaker 1994). If you have never held a lizard before then seek appropriate training from an expert. Although lizards can re-grow their tails the tail is important for escaping predators, fat storage, behavioural interactions (including mating) and having healthy offspring (Chapple et al. 2002; Chapple & Swain 2002a,b).

At a minimum, lizards should be identified to species level and released alive beside the trap in which they were caught. However, other useful attributes can also be recorded including sex (see 'Sex identification of lizards' below), and the common size variables of mass (in grams), snout-vent length and vent-tail length (in mm) as well as any regeneration of the tail. For the measures of size, a Pesola balance or mini-portable scales and a ruler will be required. If feasible, you may also be able to record pregnancy status of females (see 'Determining reproductive status of female lizards' below) and count ectoparasites (Fig. 5).

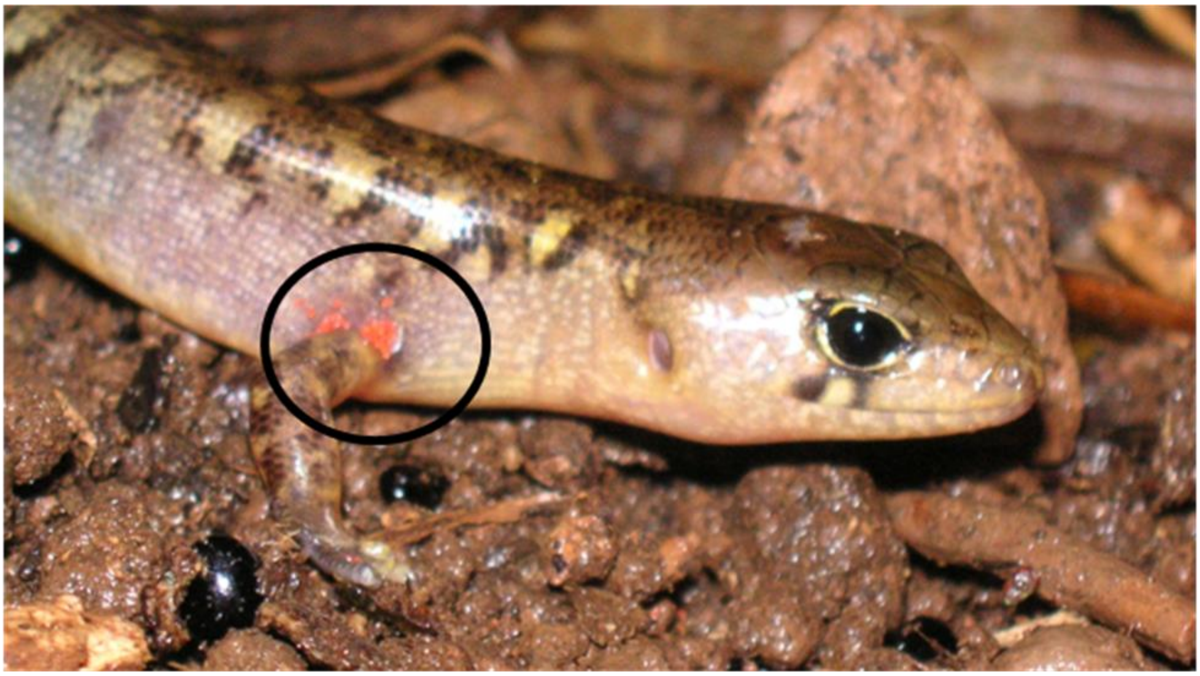

Figure 5. A robust skink (*Oligosoma alani*) with ectoparasitic chigger mites (*Odontacarus lygosomae*) clearly visible around the join of the front leg and body. These mites are most frequently located around leg joins, eyes, and ears. Another type of ectoparasitic mite, the scale mite (*O. scincorum*), has nymphs frequently found beneath the scales (particularly the tail scales) and larger adults mostly crammed into the ears. Ectoparasites are generally seasonal in abundance (Oliver 1989) (photo: Kim Miller).

Sex identification of lizards

Geckos

Some species of geckos (or populations of species) have sexual colour-dimorphism (see Jewell 2008 with corrections in Chapple & Hitchmough 2009 for details). Sex of all mature geckos can be easily identified as males have externally visible hemipenial sacs and femoral pores, whereas females have no visible sacs or femoral pores (Fig. 6). The sex of immature geckos cannot be easily identified.

Skinks

Accurate sex identification of mature *Oligosoma* skinks is relatively straightforward as hemipenes (the paired intromittent organs) are easily everted in males, particularly during the mating season in late summer/early autumn (Molinia et al. 2010). Pregnant/gravid adult females are also easily diagnosed via abdominal palpation (see 'Determining reproductive status of female lizards' below; Holmes & Cree 2006) coupled with negative hemipenial eversion. However, for most juvenile *Oligosoma* skinks, hemipenial eversion is not a reliable method of sex identification (Hare & Cree 2010, but see Hare et al. 2002). Hemipenial eversion techniques require training from an expert.

Determining reproductive status of female lizards

This section is only relevant during the breeding season when females are pregnant/gravid. Gentle palpation of the abdomen of females can provide an accurate assessment of reproductive status and often the number of embryos/eggs (e.g. skinks, Hare et al. 2010; geckos, Cree & Guillette 1995). However, palpation should *only* be used after training has been received from an expert due to the potential to rupture eggs/embryos (Gartrell et al. 2002). Always seek appropriate training and expert help.

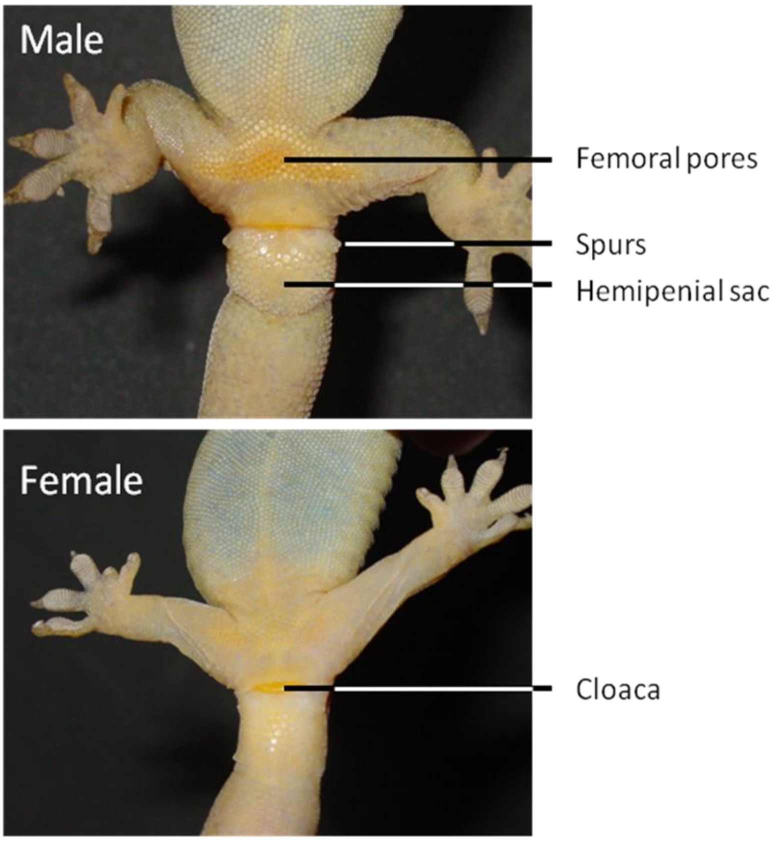

Figure 6. Cloacal region of mature male and female common geckos (*Woodworthia maculatus*). For simplification the cloaca is shown on the female only. The male shows the femoral pores, spurs and hemipenial sac, which are not present in females (photos: Jo Hoare).

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Appendix A

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

docdm-493179	Herpetofauna: indices of abundance
docdm-531081	Sampling avian blood and feathers, and reptilian tissue (SOP)
docdm-146272	Standard inventory and monitoring project plan