Herpetofauna: pitfall trapping

Version 1.0

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

Contents

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Te Papa Atawhai

Synopsis

Pitfall trapping is a common method for surveys of terrestrial herpetofauna. Pitfall traps are containers recessed into the ground and are used to capture terrestrial fauna—animals fall into the traps and cannot get out. Pitfall traps are rarely used for frogs in New Zealand (because of issues with dehydration), and are not useful for tuatara (tuatara and frogs can be effectively surveyed using systematic searches). Pitfall trapping is most useful for capture of terrestrial lizards, especially skinks (geckos can climb out of traps; Whitaker 1982). Pitfall trapping can be used to provide data for distribution, inventory, indices of abundance, density estimates, population trends, site occupancy and estimates of survival. Pitfall trapping cannot be used for total counts as some individuals will not be caught in traps—total counts are not discussed further within this method.

Pitfall traps are relatively non-destructive to the habitat (Towns 1991). They cause more disturbance than other capture techniques (e.g. when compared with artificial cover objects or funnel/G-minnow traps), but less than hand systematic searching. Observer bias is low for pitfall trapping, and relates only to the ability of the observer to identify species and handle herpetofauna effectively (Towns 1991). Pitfall traps can take a long time to install initially, but once the traps are in place, trapping can be easily repeated over short and long periods with minimal disturbance to the habitat (traps can be closed temporarily for one night or several months or even years; note: pitfall traps should be removed if a study is not likely to be continued). Some species will not enter new traps, because they are wary of areas with recent disturbance and traps may require a placement period, or 'settling in' time (Towns 1991).

Many factors can influence the capture probability and quality of the data obtained from pitfall trapping. For example, capture probabilities vary widely among species (Lettink & Seddon 2007). Additionally, capture probabilities of one species may vary among sites and microhabitats (Lettink & Seddon 2007), and are strongly influenced by additional environmental factors such as weather, temperature and moon phase (Read & Moseby 2001). Other factors that may influence capture rate include age and sex of an individual (Jones & Bell 2010), food availability in the surrounding environment (Lettink & Seddon 2007), and previous encounters with traps, as well as behaviour of the species and individuals (Towns 1991). The total area used for trapping, sample sizes, and sampling methodology undertaken (e.g. with mark-recapture v. without mark-recapture) will depend on the aims of the project and the type of data being collected, as well as the species' behaviour and habitat type.

In some cases pitfall traps filled with preservative (originally set for capture of invertebrates) have accidentally captured herpetofauna (e.g. Hochstetter's frog, *Leiopelma hochstetteri*; Baber et al. 2006), but preservative-filled traps should not be used specifically to catch herpetofauna—only live traps should be used. Furthermore, because 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 predators and sun is necessary for all traps (see 'Full details of technique and best practice').

Pitfall 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 all depend on the aims of the study, and these aims should be well developed and thought through before pitfall traps are deployed. See 'Case study A' and 'Herpetofauna: indices of abundance' (docdm-493179) for examples of potential uses of pitfall traps.

Assumptions

- The target species is/are catchable using pitfall traps.
- All habitats are equal in their ability 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 pitfall trapping. Therefore, additional assumptions may apply depending on the capture technique employed and the aims of the study.

Advantages

- Easily repeatable between studies and over time. Traps can be closed temporarily, making it easy to repeat trapping efforts over long periods of time with minimal disturbance to the habitat. Therefore, pitfall trapping is especially useful for long-term studies.
- Relatively non-destructive to the habitat.
- Usually cheap and 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. trap type, bait type, season, time of day, observer).
- 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 pitfall trapping. Therefore, additional advantages may apply depending on the capture technique employed and the aims of the study.

Disadvantages

- Requires a large time investment to initially install a trapping grid.
- Initially may be more destructive to habitat than other methods of capture (e.g. artificial cover objects or funnel/G-minnow traps).

- 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 daily to reduce the potential for deaths (e.g. desiccation, drowning, predation, etc.).
- Capture probabilities usually decline over time within a trapping session.
- Many species will not be caught reliably in pitfall traps (e.g. arboreal species, fossorial species) and capture probabilities may vary widely among species (e.g. geckos can climb out of traps).
- Not useful for frogs and tuatara.
- 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.

A range of analytical methods can be used in conjunction with pitfall 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

Pitfall trapping can confirm presence, but not absence, of herpetofauna and is suitable for inventory (e.g. Towns 1991). Pitfall trapping is relatively inexpensive, but is also strongly dependent on species' biology and habitat use, and as such is most useful for terrestrial lizards, especially skinks (see 'Full details of technique and best practice').

Suitability for monitoring

Pitfall trapping is generally suitable for monitoring of terrestrial skinks, and is relatively inexpensive. However, pitfall trapping is dependent on biology and habitat use of the species as well as weather. Pitfall 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).

Pitfall trapping requires the following skills and training:

- Species identification
- Experience with handling reptiles
- Experience with pitfall trapping
- Physical ability to use a spade and/or crowbar to install traps
- 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' for more in-depth details.

Resources

Standard equipment for all pitfall trapping techniques includes:

- One or more skilled worker(s)
- Datasheets/notebooks and pencils
- GPS
- Spade (to dig a hole for traps) and/or crowbar to lever out large rocks
- Flagging tape to mark the start of the grid
- Traps and covers
- Bait
- Moistened sponge

Useful, although not always essential items include:

- Protection from predators (e.g. square of 'chicken' mesh)
- Digital camera
- Temporary holding bags for captured animals (thin cloth bags are good for reptiles)
- Non-toxic permanent markers for individually marking reptiles
- Animal ethics permits for some techniques (e.g. PIT tagging)
- 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 recommended that novices 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; see 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 skinks of each species present in each pitfall 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 pitfall 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.

[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/

- *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.

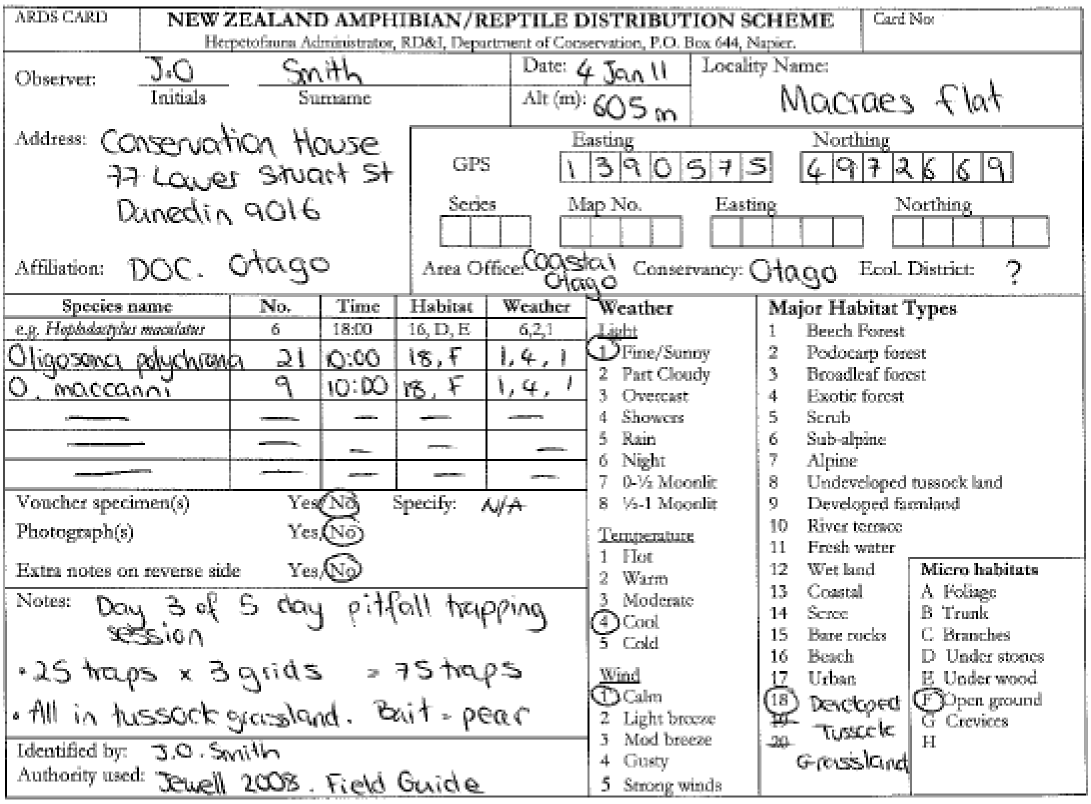


Figure 1. Example of how to fill in a New Zealand Amphibian/Reptile Distribution Scheme (ARDS) card. Note that either a GPS location or a map series number is sufficient. Also, try not to leave blank spaces—instead leave an indication that those data were not available or collected. If further notes are collected these can be included under 'Notes', and continued 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[2] should be completed and forwarded to the Herpetofauna Administrator (address shown on ARDS card; see Fig. 1).

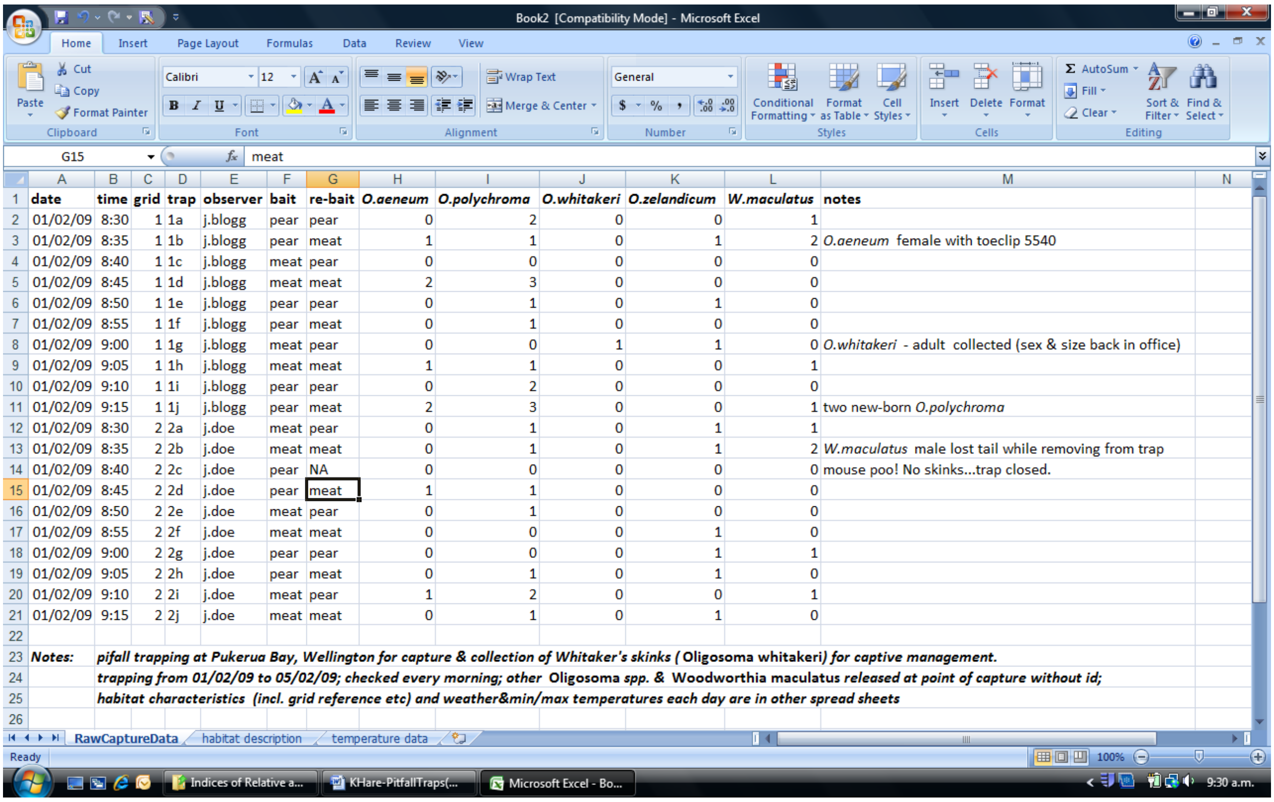
Collate, consolidate and store survey information securely, as soon as possible, and preferably immediately on return from the field. The key steps here are data entry, storage and maintenance for later analysis, followed by copying and data backup for security. Summarise the results in a spreadsheet or equivalent. Arrange data as 'column variables'—i.e. arrange data from each field site on

[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/

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

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	grid	trap	observer	bait	re-bait	*O.aeneum*	*O.polychroma*	*O.whitakeri*	*O.zelandicum*	*W.maculatus*	notes
01/02/09	8:30	1	1a	j.blogg	pear	pear	0	2	0	0	1	
01/02/09	8:35	1	1b	j.blogg	pear	meat	1	1	0	1	2	*O.aeneum* female with toeclip 5540
01/02/09	8:40	1	1c	j.blogg	meat	pear	0	0	0	0	0	
01/02/09	8:45	1	1d	j.blogg	meat	meat	2	3	0	0	0	
01/02/09	8:50	1	1e	j.blogg	pear	pear	0	1	0	1	0	
01/02/09	8:55	1	1f	j.blogg	pear	meat	0	1	0	0	0	
01/02/09	9:00	1	1g	j.blogg	meat	pear	0	0	1	1	0	*O.whitakeri* - adult collected (sex & size back in office)
01/02/09	9:05	1	1h	j.blogg	meat	meat	1	1	0	0	1	
01/02/09	9:10	1	1i	j.blogg	pear	pear	0	2	0	0	0	
01/02/09	9:15	1	1j	j.blogg	pear	meat	2	3	0	0	1	two new-born *O.polychroma*
01/02/09	8:30	2	2a	j.doe	meat	pear	0	1	0	1	1	
01/02/09	8:35	2	2b	j.doe	meat	meat	0	1	0	1	2	*W.maculatus* male lost tail while removing from trap
01/02/09	8:40	2	2c	j.doe	pear	NA	0	0	0	0	0	mouse poo! No skinks...trap closed.
01/02/09	8:45	2	2d	j.doe	pear	meat	1	1	0	0	0	
01/02/09	8:50	2	2e	j.doe	meat	pear	0	1	0	0	0	
01/02/09	8:55	2	2f	j.doe	meat	meat	0	0	0	1	0	
01/02/09	9:00	2	2g	j.doe	pear	pear	0	0	0	1	1	
01/02/09	9:05	2	2h	j.doe	pear	meat	0	1	0	1	0	
01/02/09	9:10	2	2i	j.doe	meat	pear	1	2	0	0	1	
01/02/09	9:15	2	2j	j.doe	meat	meat	0	1	0	1	0	

Notes: *pifall trapping at Pukerua Bay, Wellington for capture & collection of Whitaker's skinks (* **Oligosoma whitakeri** *) for captive management.*
trapping from 01/02/09 to 05/02/09; checked every morning; other **Oligosoma spp.** *&* **Woodworthia maculatus** *released at point of capture without id;*
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 pitfall 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 pitfall 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: influence of microhabitat on capture of lizards

Synopsis

Lettink & Seddon (2007) used pitfall trapping to investigate whether trap placement influences capture rate of four terrestrial lizard species. The influence of trap placement on capture rate of herpetofauna has not been explored in great detail, but is important because optimising trapping techniques can increase the detectability and capture rates of species, making conclusions gained from data more robust. Lettink & Seddon (2007) placed 30 pitfall trap grids of 16 pitfall traps within three habitat types on Kaitorete Spit (South Island) and obtained data for 4800 trap days. Five microhabitat factors were measured and tested for their ability to influence capture of the two most commonly captured species—McCann's skink (*Oligosoma maccanni*) and the common skink (*O. polychroma*). Lettink & Seddon (2007) showed that capture rate was higher in traps set closer to (v. further from) cover. Other weaker correlations with amount of vegetation and vegetation type were also found. The data show that trap placement and microhabitat effects can strongly influence capture rate.

Objectives

- To investigate the influence of microhabitat factors on pitfall trap capture rates of terrestrial lizards in a coastal environment.

Sampling design and methods

Pitfall trapping

Lizards were sampled at 30 locations along Kaitorete Spit during November and December 2003. The species sampled included McCann's skink, common skinks, spotted skinks (*O. lineoocellatum*) and common geckos (*Woodworthia brunnei*, as *Hoplodactlus maculatus* in Lettink & Seddon 2007). Sampling sites were randomly allocated within three habitat types: duneland (*n* = 20), shrubland (*n* = 5; remnant surrounding Birdling's Flat) and farmland (*n* = 5) using GIS technology. Twenty sites were assigned to coastal duneland due to the high conservation value of this habitat, and these were situated between Birdling's Flat and Taumutu.

Each sampling site consisted of 16 pitfall traps placed 15 m apart in a 4 x 4 grid. Pitfall traps were 4.5-L square, white plastic buckets or 'space-savers' from Containment Solutions and were dug into the ground leaving their rims flush with the surface. Holes were drilled into the bottom for drainage. To prevent desiccation and predation of captured lizards, plywood covers with spacers glued to each corner were secured above each trap using steel pegs, leaving a 1–2 cm gap for entry by lizards. Traps were baited with c. 1 cm^3 pieces of tinned pear and checked daily for 5 consecutive days each month; bait was replaced every second day. When traps were not in use, sticks were left in them to allow lizards to climb out.

Data recorded for each trap included: site, trap number, species, mass, snout-vent length, individual ID, sex and pregnancy status (of adult females only). All captured lizards were toe-clipped[3] to allow for individual identification, and released within 1 m of the trap they were caught in, within 24 h of capture.

Microhabitat factors

For each pitfall trap (n = 480) the authors recorded:

- Amount of vegetation cover within 1-m radius of trap
- Distance (m) to nearest vegetation large enough to act as a potential refuge for lizards (described in Martín & Lopez 1995)
- Habitat type (duneland, shrubland, or farmland)
- Presence/absence of divaricating shrubs and/or vine *Muehlenbeckia complexa* within 5 m.

At each trapping site (n = 30), the authors also obtained an index of invertebrate abundance by counting the number of small invertebrates (1–5 mm long) collected in ten 80-mL lethal pitfall traps filled 1/3 with dilute antifreeze (modified from Green 2000). Small pitfall traps were used to prevent lizards from drowning in these traps. Invertebrate traps were placed in pairs c. 5 cm apart at five positions within each lizard trapping site. Invertebrate pitfalls were also covered with plywood covers and were open only during the lizard trapping. A site-specific index of invertebrate abundance was assigned to lizard pitfall traps for use in analyses.

Capture rate

Capture rate was calculated as the number of lizards per trap per 10 days and was used for all analyses.

Results

In total, 536 captures of 401 individuals were made over 4800 trap days. Most captures were of McCann's skinks (c. 61%) and common skinks (c. 34%), and analyses focused on these two species. Analyses for spotted skinks and common geckos did not have enough power to make strong predictions; spotted skinks were at very low numbers at the sites and pitfall trapping is not so useful for capture of geckos. Details of size and sex-ratio were provided for all four species. Capture rates were lower in November compared with December, but as data for a given trap were pooled this effect was unimportant for the analyses. Over half the traps failed to capture any lizard during the 10-day survey.

[3] Note that DOC no longer considers toe-clipping an ethical method of marking herpetofauna. Alternatives should be sought if at all possible.

Capture rate increased with an increase in vegetation cover around the traps and declined as the distance between the trap and nearest cover increased (these variables were correlated). Having divaricating shrubs and/or *M. complexa* within 5 m of a given trap also improved capture rate. Capture rate varied with habitat type for both species, being highest for McCann's skinks in duneland and highest for common skinks in farmland. Fewer common skinks were captured where invertebrate abundance was high, but this relationship was weak; invertebrate abundance had no influence on capture rates of McCann's skinks.

Limitations and points to consider

Limitations:

- Capture rates are strongly influenced by density and detectability of species and individuals. Therefore, the authors assumed density to be relatively uniform within sites given the small size of their trapping grids (0.023 ha) and relative homogeneity of the habitat at each site, and used this within analyses. However, the assumption of uniform density is probably violated given that capture rate was strongly influenced by microhabitat variables within the sites. Another option was for the authors to state that both detectability and density could not be included in the analyses.
- As stated by the authors, some overlap in microhabitat factors could not be avoided (e.g. proximity to cover and amount of cover were inversely related), making it difficult to determine which (if any) are more important.
- The statistical modelling necessary for this complex observational study means that only the two species for which enough captures were obtained (McCann's and common skinks) could be used in analyses. The capture rate of spotted skinks and common geckos could not be used in analyses.

Points to consider:

- Traps were all baited with pear and this was changed regularly allowing for the assumption that traps were equally likely to capture lizards based on bait type.
- All traps were set flush to the ground and had drainage holes included, which is excellent practice for pitfall trapping studies. However, whether there were differences in capture rate between the two trap types used was not discussed. This raises important questions that may or may not be relevant to capture rate of the lizards, including: How many of each trap type were used? Were the two trap types evenly distributed among and within sites and microhabitats? Was one trap type more effective for capturing lizards?
- Robust statistical models were developed to account for the many variables included in this ambitious project design. For example, including site in statistical models vastly improved the fit of the model to the data, and was an important variable.
- Some important details alluded to in the introduction are missing from the methods, and may be an artefact of the requirement for brevity in published journal articles. For example, no details of time-of-day of trap checks, length of time required to visit the traps, etc. were included, which may influence capture rates among traps. Could all 30 sites and 480 traps be visited within one day? If the answer to this question is yes, this may be why it took up to 24 h to release the

animals as they were measured elsewhere, meaning that traps were in fact checked quickly and efficiently. If the answer is no, and traps within the sites were open on alternate weeks within the 2 months, then details of weather and temperature factors would also need to be included as factors in analyses as these will influence capture rates.

- The study demonstrates nicely the differences in capture rates among species and within habitats, and how only within-species comparisons can be used because of differences in the capture probability among species and different habitats.
- Many fewer sites were trapped for lizards in shrubland (only one area of remnant shrubland is suitable for trapping on Kaitotere Spit) and farmland compared with duneland and, despite the excellent statistical analyses undertaken, may have an influence on the results due to unbalanced site numbers.

References for case study A

Green, C. 2000: Pitfall trapping for long-term monitoring of invertebrates. *Ecological Management 8*: 73–89.

Lettink, M.; Seddon, P.J. 2007: Influence of microhabitat factors on capture rates of lizards in a coastal New Zealand environment. *Journal of Herpetology 41*: 187–196.

Martín, J.; Lopez, P. 1995: Influence of habitat structure on the escape tactics of the lizard *Psammodromus algirus*. *Canadian Journal of Zoology 73*: 129–132.

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 pitfall trapping to locate and/or capture terrestrial lizards; traps are less effective at capturing geckos (they can climb out of traps). Pitfall traps are not presently recommended for capturing native and exotic frogs and are not useful for tuatara. If you have no experience with pitfall trapping you should consult with someone who has experience prior to conducting field work. 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

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, pitfall 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, individual traps are used as the sampling unit (Lettink & Seddon 2007), but in this case the captures per trap for a given time period are calculated. At a minimum, three grids/lines should be employed, but five or more is better for statistical analyses. 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. Depending on the goals of the study, traps may be set at distances of 1 m up to 10–20 m apart (e.g. 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, pitfall 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. Lettink et al. 2011). This length of time is a trade-off between gaining enough data for analyses and the fact that capture rates are normally highest the first night traps are set and decline steadily over time (Moseby & Read 2001). Traps can be closed temporarily by clearing out all trap contents and placing sticks or rocks inside the trap up to the lip. For long-term closure (e.g. months/years) the trap lid should also be sealed and heavy sticks or rocks placed over the top. Although a tight seal should prevent animals from entering traps while closed, placing sticks or rocks inside the trap will ensure that any animals that may enter the trap are able to climb back out.

Traps should be checked at least every 24 h, 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 pitfall trap

Trap type

Generally, 2-L to 4-L paint buckets are typically used for herpetofauna in New Zealand; however, any size trap can be used as long as it is deep enough that the largest individuals of the target species cannot climb out (Whitaker 1994). Also, sometimes overly deep traps reduce the likelihood of lizards falling in (see Whitaker 1994), and small pitfall traps appear to be as effective as larger ones (10 L) suggesting that the additional effort and costs involved in installing larger pitfall traps is not rewarded by improved capture rates (Maritz et al. 2007).It is preferable to have the same trap types for a study and grid in case trap type influences capture probabilities. Plastic is preferable to metal for long-term monitoring as metal buckets rust, making it easy for animals to climb out of traps, although by using lacquered tins, rusting can be minimised. Also, metal traps can easily heat in the sun and cook a trapped lizard. Dark-coloured traps are better than light-coloured ones (Crawford & Kurta 2000), probably because the trap looks more like a refuge if it is dark. A thin layer of soil or other debris sprinkled on the bottom can be used to darken the bottom of light-coloured traps.

Important trap modifications and principles for lizard safety

Traps should have small holes drilled through the bottom so that if it rains, water can drain out of the trap. It is important to check the drainage holes periodically (especially for long-term traps) to ensure they do not get clogged. Although most lizards can swim, drowning is a serious threat for lizards trapped within pitfalls (Whitaker 1994). In areas close to water or of a boggy nature it is also advisable to check that trap bottoms are not set below the water table.

All traps should have a cover. The cover will protect lizards from the sun and predators such as birds and tuatara. The bucket lid can be used as a cover, but other lids may be more suitable in different habitats (e.g. large flat rocks provide good cover for traps on boulder beaches). The cover should be set so that a gap of 1–2 cm is present for the lizard to enter the trap (Whitaker 1994). Covers should be held in place with either a rock/heavy stick or pinned to the ground with wire. If a rock/heavy stick is used, be careful that this will not fall into the trap and crush lizards—this is especially important when checking the traps.

In some habitats traps will need to have added protection from potential predators (e.g. crabs, mammals, tuatara). Wire mesh can be placed over traps (but under lids) to exclude large crabs from entering traps placed on boulder beaches (crabs will happily dissect lizards trapped with them). Wire mesh can also be put on the bottom of the trap to protect lizards from mammals (Whitaker 1994). However, mice are a threat that cannot be effectively excluded from traps (see Towns 1992), and some level of predation is to be expected at sites with mice. Any predation is an ethical dilemma and will also bias results due to fewer animals in traps than expected. Therefore, if mouse faeces or gnawed lizard carcasses are found within a trap it should be closed. If more than one trap in a grid has mouse sign, consider stopping the trapping operation. Similarly, in some locations possums, cats, dogs, etc. can disturb traps and dislodge lids. In this case, traps should be closed and ending the trapping session should be considered until mammal control is implemented (larger mammals will learn to check traps).

Installing and setting pitfall traps

Trap installation

Set out pitfall traps in the pre-determined configuration (see earlier section 'Sampling design and effort'). Label each trap by trap number (both near the lip on the inside of the trap and on the underside of the lid). 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.

Using the spade, dig a hole deep enough for the trap. If setting traps on a rocky shore, a spade may be useless and instead a crowbar can be used to lever rocks from the ground (be sure to set traps above the tide mark). If water enters the hole it is not a suitable location for a trap. Set the trap so that it sits flush with the ground and pack substrate around the rim of the container—the animal should not have to climb over a lip to fall into the trap (Fig. 3).

Figure 3. Pitfall traps should lie flush with the ground, so that animals do not have to climb over a lip to fall into the trap. This trap has the lid used as a cover with a rock on top to hold the lid in place (photo: Kelly Hare).

Setting traps

After all traps are placed into the ground, they can be set. Remove lids and add some debris and a moistened sponge as a hiding space in the bottom of the trap. A piece of wet sponge will help reduce desiccation and can provide cover in the trap (Fig. 4). 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). Some studies have tried other baits (including none) with limited success (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 flat rock or a 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 reduce the variables needed in analyses.

Use sticks or other items to raise the cover and create a 1–2 cm space for the animals to pass through. Place the cover so that it fully shades the inside of the trap, and secure it so it will not blow away in wind and/or rain.

Figure 4. Skinks and geckos within a set pitfall trap. The trap has a large moistened sponge to provide moisture and some debris in the bottom for cover. The bait has been eaten (photo: Jo Hoare).

What to do with lizards in a trap

Lizards can be removed one at a time from the trap for 'processing', or alternatively placed in soft, cloth bags, tied tightly with cord. 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 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. Remove more aggressive species and larger specimens from the trap first and do not hold them in bags with smaller species that could be potential prey for larger lizards (Whitaker 1994). 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, 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 and released alive beside the trap in which they were caught. Other useful attributes can also be recorded, including sex (see 'Sex identification of lizards' below), common size variables of mass (in grams), snout-vent length and vent-tail length (in mm), and 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 (see Fig. 5).

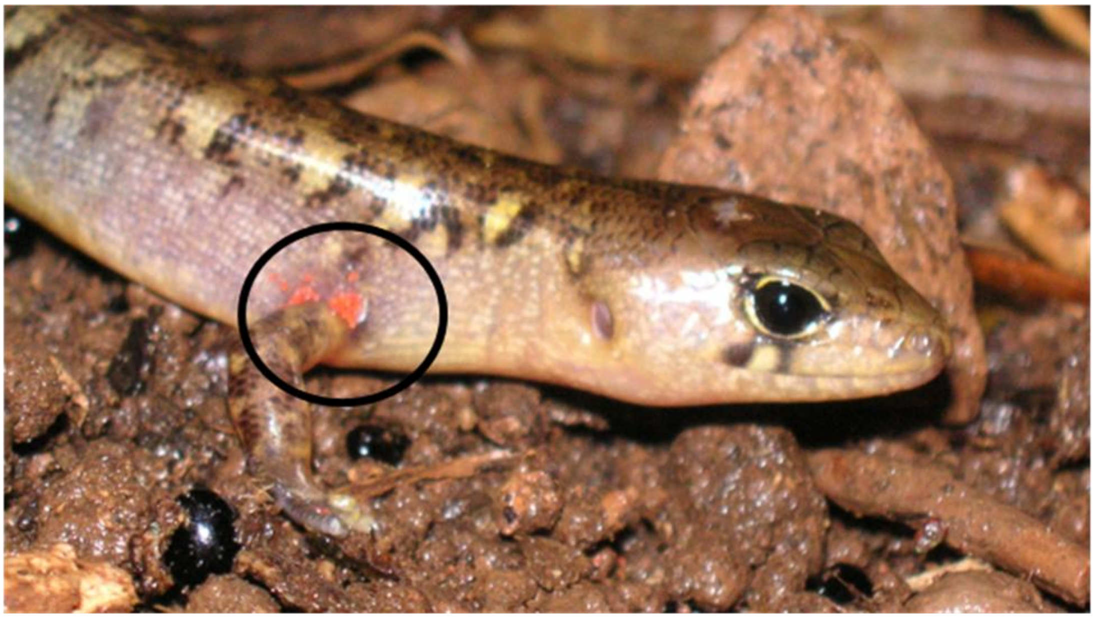


Figure 5. A robust skink (*Oligosoma alani*) with ectoparasitic chigger mites (*Odontacarus lygosomae*) clearly visible around the join of the front leg and body. These mites are most frequently located around leg joins, eyes, and ears. Another type of ectoparasitic mite, the scale mite (*O. scincorum*), 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 external sexual colour-dimorphism (see Jewell 2008 with corrections in Chapple & Hitchmough 2009 for details). Sex of all mature geckos can be easily identified—males have externally visible hemipenial sacs and femoral pores; 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 requires training from an expert as incorrect technique can damage the lizard.

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.

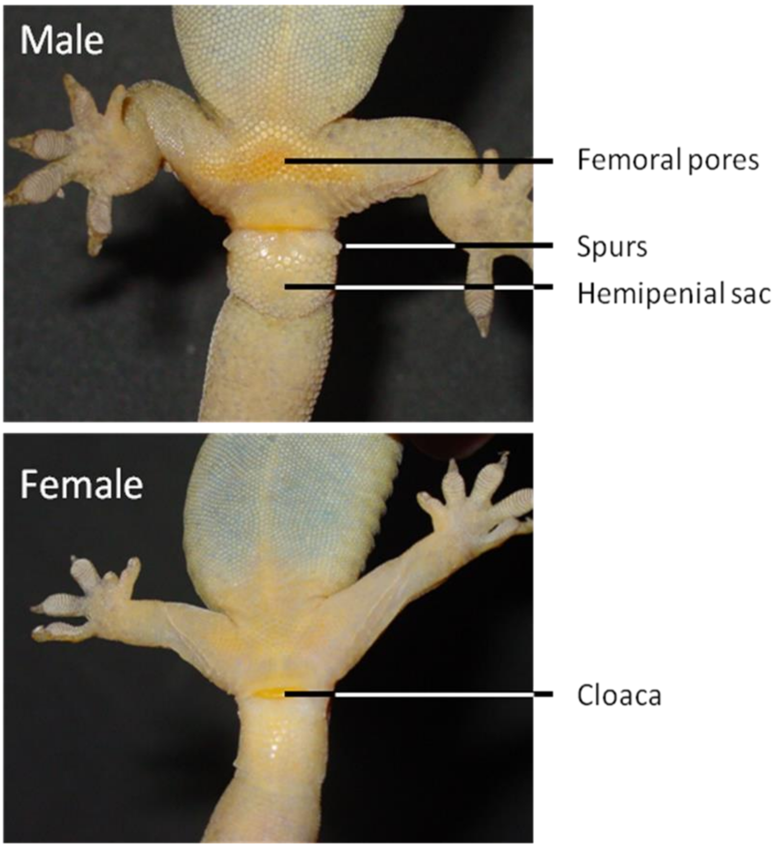


Figure 6. Cloacal region of mature male and female common geckos (*Woodworthia maculatus*). For simplification the cloaca is shown on the female only. The male shows the femoral pores, spurs and hemipenial sac, which are not present in females (photos: Jo Hoare).

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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