

Herpetofauna: indices of abundance

Version 1.0



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

Contents

Synopsis	2
Assumptions	3
Advantages.....	3
Disadvantages.....	3
Suitability for inventory	4
Suitability for monitoring.....	5
Skills	5
Resources	5
Minimum attributes	6
Data storage	7
Analysis, interpretation and reporting.....	11
Case study A	23
Full details of technique and best practice	27
References and further reading	29
Appendix A	32

Disclaimer

This document contains supporting material for the Inventory and Monitoring Toolbox, which contains DOC's biodiversity inventory and monitoring standards. It is being made available to external groups and organisations to demonstrate current departmental best practice. DOC has used its best endeavours to ensure the accuracy of the information at the date of publication. As these standards have been prepared for the use of DOC staff, other users may require authorisation or caveats may apply. Any use by members of the public is at their own risk and DOC disclaims any liability that may arise from its use. For further information, please email biodiversitymonitoring@doc.govt.nz



Synopsis

Estimation of abundance is a vital part of many conservation programmes, as it can be used to answer questions such as how populations respond to management techniques (e.g. predator control or revegetation). As it is rarely possible to count all individuals in a population, inferences are usually made by comparing a count statistic over space and/or time (e.g. number of individuals seen within a defined time period). Count data not corrected for detectability are indices (or index counts) and assume constant detection probabilities over space, time and among individuals. Indices of abundance are one of the most commonly used estimators for monitoring herpetofaunal communities, and are based on the assumption that the sample represents a constant proportion of the population.

Indices of abundance are often sufficient to describe basic biological patterns, are generally easy to conduct, and in most cases require little statistical background. They can provide information on minimum number alive (MNA), catch per unit effort (CPUE) and density (although to a limited extent). It is important for all indices of abundance that the effort applied to capture animals and the probability of capturing an animal is equal across capture events. These data can then be used for monitoring changes in abundance among sites within a habitat type, and/or trends over time and/or with different management scenarios. Indices cannot be used to compare relative abundance among species or habitat types, because it is unrealistic to assume equal capture probabilities in these cases. It is also difficult to interpret data from very small populations, and unless there is sufficient replication of capture events, trends cannot be described with any statistical confidence. Furthermore, for most herpetological studies the direct relationship between index counts and population size is assumed and has not been tested, and it remains unknown if these indices are accurate and sufficiently sensitive to detect population trends and responses to management (see Lettink et al. 2011 for more discussion).

Indices of abundance for New Zealand herpetofauna have chiefly been estimated using systematic searches (e.g. Green & Tessier 1990; Towns 1991; Moore et al. 2010). Other techniques employed have mostly been used for lizards and include pitfall trapping (especially useful for skinks; Towns 1991; Lettink et al. 2011), funnel/G-minnow traps (particularly for cryptic species; Bell 2009) and artificial cover objects (ACOs; Lettink et al. 2011). Call counts may be used to estimate abundance of exotic frogs, but are limited to inferences based on males of the species (see Driscoll 1998 for discussion). Also of use for herpetofaunal surveys may be tracking-tunnel ink cards (Russell et al. 2010); however, the efficacy of this additional method for indices of abundance has not yet been tested for herpetofauna (as it has for rodents; Brown et al. 1996), and is therefore not recommended at this point in time.

This method covers how to design a survey and conduct analyses for indices of abundance. Techniques for gaining the data by locating/capturing herpetofauna (e.g. systematic searches, pitfall and funnel/G-minnow trapping, and ACOs) are not covered in this section. More complete technique methods are under development. As such, seeking advice from a suitably experienced person for the techniques of capture referred to in this method is recommended. Details on how to estimate population size are outside of the scope of this method, and are covered in 'Herpetofauna: population estimates' (docdm-833600).



Assumptions

- A constant fraction of individuals (direct counts) or sign (indirect counts) is available among areas at the same time, among areas over time, or within an area over time.
- All observers and capture techniques have equal ability to locate/capture animals.
- The relationship between the index (capture rates) and abundance is linear or can be linearised (see Lettink et al. 2011 for an example).
- The target species is/are catchable/observable using the technique employed.
- The probability of detecting animals does not vary over time or among sites.
- The sample site(s) is/are representative of the wider population(s).
- The population remains demographically closed throughout the survey period, i.e. it is assumed that animals are not coming into (by birth or immigration) or leaving (by death or emigration) the study area.
- Capture/sighting effort is kept constant between sampling events.
- Species of interest are truly absent from the area when none are detected.

As a range of analytical methods can be used in conjunction with indices of abundance (MNA, CPUE and sometimes density), additional assumptions may apply depending on the capture technique employed and the aims of the study.

Advantages

- Depending on the technique employed, it may be cheap and easy to conduct.
- It may be sufficient to describe basic biological patterns.
- It may be useful for comparative inference if assumptions about equal detection rates are met.
- Many factors affecting detectability can be controlled by standardisation of techniques (e.g. season, time of day, observer, species, capture technique).
- Often easily repeatable between studies and over time.
- Depending on data type being collected, or if data is collected in a standardised manner, then generally this method requires little statistical background.

As a range of analytical methods can be used in conjunction with indices of abundance (MNA, CPUE and sometimes density), additional advantages may apply depending on the capture technique employed and the aims of the study.

Disadvantages

- Does not adjust for incomplete detectability. Note: spurious results may be obtained if data are not adjusted for detectability.
- Care is required when interpreting trends derived from indices, particularly for small populations.
- If the relationship between the index of relative abundance (MNA or CPUE) and the actual abundance or density is not linear, the interpretation may be flawed (see Fig. 1 for explanation).
- Capture probabilities may vary among species. Capture probabilities within a species may vary among sites or within the same site over time. Changes in habitat and/or animal behaviour,



particularly before and after management actions, may influence capture probabilities.

(Assumptions of relative abundance methodologies are hardly ever examined, but see Lettink et al. 2011 for an example).

- Capture probabilities of herpetofauna generally vary with temperature and other variables. Although many factors affecting capture probabilities can be controlled by standardisation of techniques (e.g. observer, season, time of day) many factors (e.g. daily temperature fluctuations) cannot be standardised and will need to be incorporated into analyses. Note: this is not the same as detection probabilities *per se*, which also include other factors, such as behaviour of the individual.
- Indices of abundance can only be compared within species and habitats, because of differences in the capture probability among species and different habitats.
- If search effort is not kept constant, indices can become unreliable. For example, a change in the number of traps set or the number of trap nights could lead to incorrect interpretation of the indices. See '[Full details of technique and best practice](#)' for further explanation.
- Surveys must be designed to include sufficient replication (i.e. generate several indices in one season), otherwise only trends can be described.
- Does not allow for estimation of population parameters such as population size (with detectability etc. included in analyses), survival, migration, or reproductive rates.

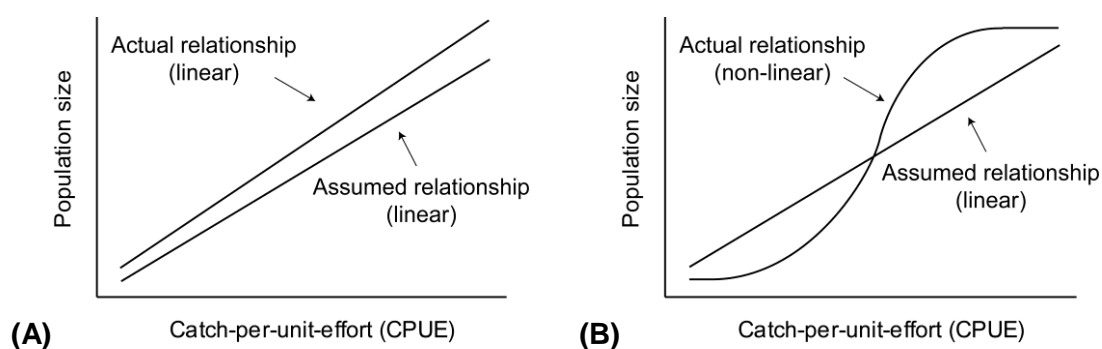


Figure 1. The relationship between the index of abundance (in this case population size) and actual abundance is assumed to be linear, but the actual relationship may or may not be. When the relationship is linear (A), changes in the index will reflect changes in actual abundance. However, when the relationship is non-linear (B), the index may not accurately reflect changes in abundance. For example, at very small population sizes, individuals may be difficult to locate, and the index of abundance would be very small unless the population exceeds a critical size. Alternately, there may be some maximum value attainable (e.g. animals in a pitfall trap may consume all bait, so that after 10 animals are trapped no lure remains).

Suitability for inventory

Indices of abundance are suitable for inventory as they provide measures of numbers for a given time or habitat type. They are particularly useful if the assumptions of the technique employed (e.g. systematic searches, pitfall trapping, etc.), particularly those of constant effort and equal detectability among trapping sessions, are met.



Suitability for monitoring

It is appropriate to use indices of abundance for monitoring. Indices of abundance are frequently used to *infer* trends in population size and can be useful for monitoring if the assumptions of the technique employed (e.g. systematic searches, pitfall trapping, etc.), particularly those of constant effort and equal detectability among trapping sessions, are met. This method is most useful when counts are repeated annually over relatively long time frames (> 10 years), when sample sizes (number of transects/sites, etc.) are high and when variation in observers, times of day, season and weather conditions are minimised. Unfortunately, controlling for observer effects and changes in environment are more demanding under long-term monitoring regimes than short-term ones. Some short-term monitoring using indices of abundance is confounded by weather but is possible given care, good design (experimental controls) and appropriate sampling effort relative to the precision required.

Skills

- Ability to design adequate sampling protocol for the survey
- Species identification
- Experience with the technique employed (e.g. systematic searches, pitfall trapping, etc.)
- Ability to write clear and thorough notes
- Proficiency using Microsoft Excel or other statistical software
- Basic understanding of statistics

The exact skills required will vary depending on the technique(s) used to capture animals; techniques (e.g. systematic searches, pitfall and G-minnow trapping, and ACOs) are not covered in this method. It is advisable to seek advice from a suitably experienced person for the techniques referred to in this method.

Resources

Standard equipment useful for calculating indices includes:

- Field datasheets
- A computer
- Microsoft Excel
- A calculator
- A statistics book (e.g. Clark & Randal 2011)
- A statistical programme (e.g. R, SPSS, etc.)

Other resources include those used in the field to get the data and these will vary largely depending on the technique(s) used to capture/locate animals. These techniques (e.g. systematic searches, pitfall and G-minnow trapping, and ACOs) are not covered in this method. As such, it is advisable to refer to these methods and/or to seek advice from a suitably experienced person for the techniques referred to in this method. More information on survey design is available under [‘Full details of technique and best practice’](#).



Minimum attributes

Consistent measurement and recording of these attributes is critical for the implementation of the method. Other attributes may be optional depending on your objective. See '[Full details of technique and best practice](#)'. However, it is recommended that novices obtain training from an expert herpetologist. 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).¹ Thorough, tidy and clear data entry is vital.

Required attributes

At a minimum, the following data should be recorded:

- Observer(s) and/or recorder
- Date and time
- Location name/grid reference
- Capture point of each individual (including a GPS position)
- Technique employed to capture/locate individuals
- Temperature data (overnight minimum and daytime maximum temperatures are essential; either collect local temperatures during the study or get weather station data after the fact)

Optional attributes

Based on the specific goals of each project and the capture technique used, other factors may be required. Additional attributes that may be useful to record while in the field, but are generally not required for indices of abundance, include:

- *Habitat characteristics*: location description, altitude, aspect, vegetation (including dominant plant species), available cover, temperature of substrate.
- *Other weather characteristics*: ambient air temperature at time of capture (shade, 1 m from ground), relative humidity, precipitation, cloud cover, wind direction and strength.
- *Individual morphological measurements*: snout-vent length (SVL; mm), mass (g) and records of natural toe-loss. For lizards and tuatara, vent-tail length (mm, include regeneration) can also be obtained.
- *Sex*: the sex of individuals is also a useful parameter to record. See the 'Full details of technique and best practice' section of 'Herpetofauna: systematic searches' (docdm-725787) for more detail.
- *Reproductive status of females*: see the 'Full details of technique and best practice' section of 'Herpetofauna: systematic searches' (docdm-725787) for more detail.

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/>



- *Infestations of external parasites*: see the 'Full details of technique and best practice' section of 'Herpetofauna: pitfall trapping' (docdm-760240) for more detail.

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).²

Collate, consolidate and store 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 area 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.

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.

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/>



	A	B	C	D	E	F	G	H	I	J	K	L	M	N	O
1	date	time start	time end	air t (C)	observer Left	observer Right	transect	length (m)	newLeft	newRight	new total	resightLeft	resightRight	resight total	total seen
2	26/10/10	21:00	23:00	11.7	AB	CD	1	25	9	5	14	0	0	0	14
3	26/10/10	21:00	00:15	11.7	EF	GH	2	30	10	11	21	0	0	0	21
4	26/10/10	23:15	01:30	10.5	CD	AB	3	20	7	8	15	0	0	0	15
5	26/10/10	00:30	02:00	10.2	GH	EF	4	30	10	11	21	0	0	0	21
6	27/10/10	21:00	23:15	9.7	EF	GH	1	25	3	2	5	5	2	7	12
7	27/10/10	21:00	23:30	9.7	AB	CD	2	30	4	2	6	5	6	11	17
8	27/10/10	23:30	00:15	9.1	GH	EF	3	20	2	1	3	4	8	12	15
9	27/10/10	23:45	02:00	9.1	CD	AB	4	30	5	2	7	5	9	14	21
10	28/10/10	21:00	23:00	10.3	CD	AB	1	25	4	2	6	2	7	9	15
11	28/10/10	21:00	00:15	10.3	GH	EF	2	30	3	1	4	7	8	15	19
12	28/10/10	23:15	01:30	9.5	AB	CD	3	20	0	2	2	6	5	11	13
13	28/10/10	00:30	02:00	9.5	EF	GH	4	30	1	1	2	10	9	19	21
14	29/10/10	21:00	23:15	12.2	GH	EF	1	25	0	1	1	9	5	14	15
15	29/10/10	21:00	23:30	12.2	CD	AB	2	30	2	3	5	10	10	20	25
16	29/10/10	23:30	00:15	11.5	EF	GH	3	20	0	0	0	8	7	15	15
17	29/10/10	23:45	02:00	11.5	AB	CD	4	30	1	0	1	9	10	19	20
18															
19															
20	NOTES:	<i>Stephens Island/Takapourewa tuatara (Sphenodon punctatus)</i>													
21		<i>Transects in Keepers Bush (5-m searchable on each side of known tracks)</i>													
22		<i>Visual systematic searches with capture and temporary marking with twink used at night</i>													
23		<i>two observers per transect (one each side; randomised side of track</i>													
24		<i>morphometric and sex data also recorded and provided on another spread sheet</i>													
25															

Figure 2. Example of metadata collected during a field systematic search. Data on individuals seen/captured are available in another worksheet (tab). 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. More or fewer columns can be added as required. These data could be used to calculate minimum number alive, catch per unit effort and crude density estimates.

Checking for missing data & errors

Missing data and large errors within datasheets can make analyses and interpretation problematic. Therefore, it is important to always do a quick check to ensure that data files are complete and appear correct.

Missing data

Missing data (empty data cells) are a problem for analyses and interpretation as it is not obvious whether the data were not recorded, not transcribed onto the spreadsheet, or not important to record for that variable (which should be recorded as 'NA'—not applicable). A simple way to find if data are missing in Excel data sheets is to use the 'Filter' function. For example, in a hypothetical example, if you are checking whether all the species names have been recorded for individuals then you would:

1. Click on 'Data' and 'Filter'.
2. Choose the 'AutoFilter' option (now each column label will have a small box with an upside down triangle visible in it; Fig. 3).
3. Click on the small triangle in the column 'grids' and a list of variables present within that column will be provided.
4. If '(Blanks)' is provided in the list then there are missing data (Fig. 4). Choose '(Blanks)' to have only the rows with blank data cells in the column 'grids' provided.



- Refer back to the field data sheet for the correct value to insert and insert the correct value. If the error is on the field data sheet then it may be appropriate to type in 'NA' so that it is obvious that the data are not missing, they were simply not recorded. Note: 'NA' is preferable over other letter combinations for analyses using the statistical programme *R*.

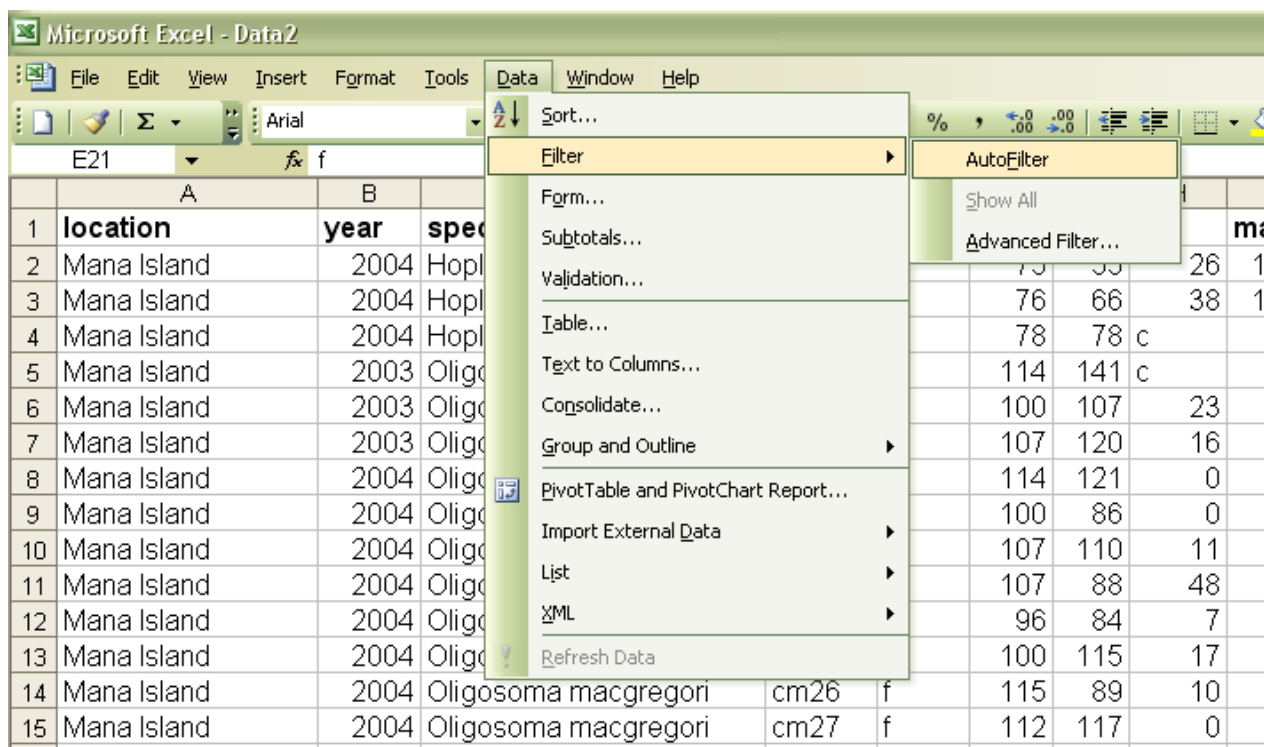


Figure 3. How to choose the 'AutoFilter' option in Microsoft Excel.

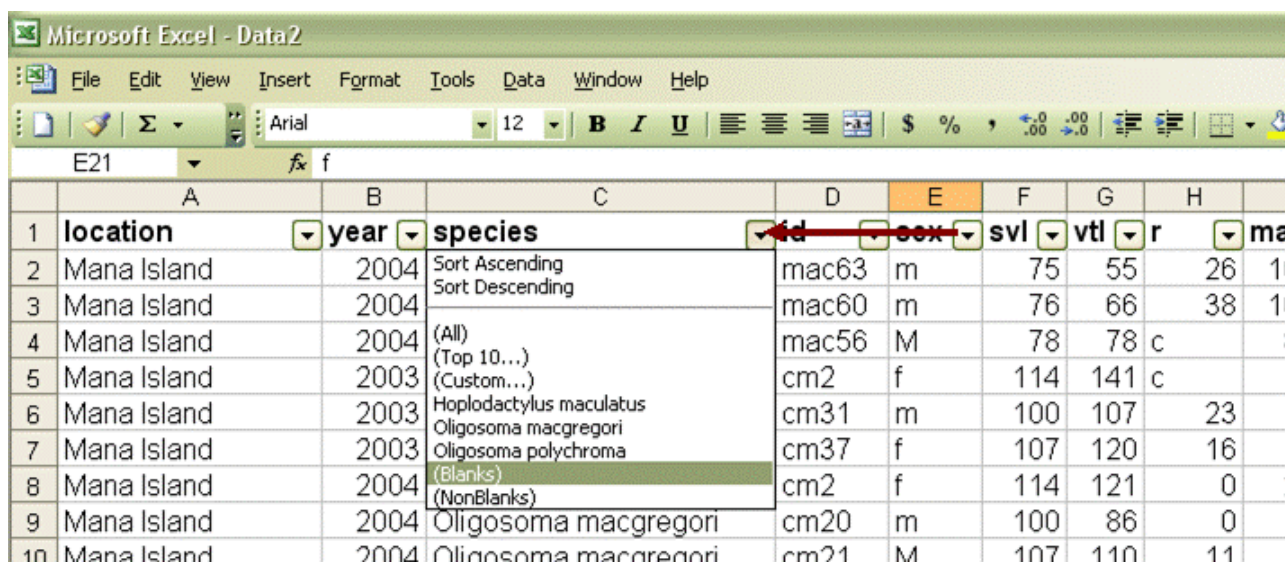


Figure 4. How to use the 'Filter' option to find missing data. The red arrow points to the 'triangle' which opens the list of data in the column. By choosing '(Blanks)' only those data cells that have no data within them will be shown. To return to having all data showing choose '(All)'.



Inconsistent notation

1. Inconsistent notation for variables can become a real problem for analyses. For example, inconsistent use of 'M' or 'm' or 'male' in a column denoting sex will result in some statistical programmes coding for three different types of sex just for males, when there should be only one. This can be easily caught in the same manner as for 'missing data' by using filters and ensuring that the list has only the relevant variables listed.

Large errors

A simple way to catch obvious errors in data sheets is to use the data sorting option. For example, if you are checking that the masses for a group of lizards are correctly recorded then you would:

- Highlight *all* the columns in the spreadsheet (if a column is missed then the data will be mis-sorted and useless); you can use 'Ctrl + A' to do this.
- Click on 'Data' and 'Sort'.
- Choose the 'Header row' option.
- Sort the data by the column variable of interest (in this case 'mass'; Fig. 5).
- Check the minimum and maximum values in the column.
- If there is an apparent error (e.g. the maximum value is 100.0 g which is much too large for a lizard) refer back to the field data sheet for the correct value.
- Insert the correct value. If the error is on the field data sheet then it may be appropriate to type in 'NA' to signify that the data are missing.

	A	B	C	D	E	F	G	H	I	J
1	location	year	species	id	sex	svl	vtl	r	mass	rege
2	Mana Island	2004	Hoplodactylus maculatus	mac56	M	78	78	c	8.986	n
3	Mana Island	2004	Hoplodactylus						66	38 10.327 y
4	Mana Island	2004	Hoplodactylus						55	26 10.489 y
5	Mana Island	2003	Oligosoma ma						07	23 16.5 y
6	Mana Island	2004	Oligosoma ma						86	0 18 y
7	Mana Island	2004	Oligosoma ma						84	7 19.2 y
8	Mana Island	2004	Oligosoma ma						10	12 202 y
9	Mana Island	2004	Oligosoma ma						15	17 20.6 y
10	Mana Island	2003	Oligosoma ma						20	16 22 y
11	Mana Island	2004	Oligosoma ma						88	48 23 y
12	Mana Island	2004	Oligosoma ma						10	11 24.5 y
13	Mana Island	2004	Oligosoma ma						89	10 26 y
14	Mana Island	2004	Oligosoma ma						17	0 27.4 y
15	Mana Island	2004	Oligosoma ma						21	0 28.57 y
16	Mana Island	2003	Oligosoma ma						41	c 32 n
17	North Brother Island	2004	Hoplodactylus maculatus	mac14					21	14 7.5 y

Figure 5. How to use the data sorting option in Microsoft Excel.



Analysis, interpretation and reporting

Standardised analysis and interpretation allows comparisons to be made among different sites and at different times. At a minimum it is recommended that you:

- 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.

The most appropriate analysis for any given dataset will depend on the goals of the project and some of the statistical properties of the data (e.g. whether the data is normally distributed with equal variance). In general, the indices of abundance that can be generated through various techniques include: MNA, CPUE, and in some cases density estimates. A biometrician or person with suitable statistical expertise should be able to recommend appropriate statistical tests. Useful statistical concepts and tests are outlined in this section, as well as some hypothetical examples for crude analyses.

Microsoft Excel can be used to conduct some basic statistical tests (e.g. ANOVA in the Analysis ToolPak). Statistical software (e.g. SPSS, SAS, or *R*) may be required for more sophisticated analyses, including the analysis of additional variables (e.g. temperature, etc.). Note: detailed analyses on population size from mark-recapture data will require specialist skills and researchers should seek advice on the best ways to analyse trends. This is covered in the 'Herpetofauna: population estimates' (docdm-833600) module.

It is critical that data be interpreted cautiously in light of the assumptions of the method and the study system. Once results are obtained, they should be reported according to any local, regional, or national requirements.

Common statistical tests and terminology

Some commonly used statistical methods and terminology referred to within the remainder of this method are outlined in more detail here in alphabetical order (not order of importance). To determine which statistical test is appropriate for your data, either consult a statistician, a statistics book (Clark & Randal 2011), or one of the many online statistical help websites (e.g. 'How to choose a statistical test'³, 'What statistical analysis should I use?'⁴; there are many more sites).

Note: this is not a complete list, but instead a starting point for those with less statistical background. It is strongly urged that statistical advice be sought from a biometrician or suitably experienced person prior to undertaking any analysis. It is also recommended that a suitable textbook such as Clark & Randal (2011) be used as a reference.

Analyses of variance (ANOVAs)—useful for comparing two, three, or more means, and provide a statistical test of whether or not the means of several groups are all equal. ANOVAs can be calculated in

³ <http://www.graphpad.com/www/book/choose.htm>

⁴ http://www.ats.ucla.edu/stat/mult_pkg/whatstat/default.htm



Excel, but are more easily and accurately calculated using statistical packages. ANOVAs are generally reported as:

$$F_{df} = \text{'a given output'}$$

Analysis of covariance (ANCOVA)—a combination of ANOVA and regression that allows for partition of variance due to certain factors after controlling for variance due to continuous variables (covariates). ANCOVAs are reported in the same manner as ANOVAs.

Degrees of freedom (df)—the sample size minus the number of parameters to be estimated from the data (i.e. the number of values in the data at the final calculation that are free to vary).

Linear models (lm)—the general name for regression analyses, and can be used to calculate whether there is a difference between two or more means, and can include covariates. ANOVA, ANCOVA and *t*-tests are a type of linear model, therefore linear models are reported in the same manner as these.

Mean—also commonly called the average (but more appropriately termed the arithmetic mean), is the sum of all the numeric values divided by the number of values. In Microsoft Excel the mean can be calculated by the following formula:

$$= \text{AVERAGE('highlight cells of interest')}$$

Normality of residuals—one of the most important assumptions for linear models (including classical tests like *t*-tests and ANOVA). If the residuals are not normally distributed then linear models should not be used. Non-normal data may be able to be normalised by logging the response variable, or taking the square root if count data are used. If data cannot be normalised then other statistical tests will be required. Talk to a statistician.

Range—the difference between the minimum and the maximum values of a variable. In Microsoft Excel the range can be calculated by the following formula:

$$= \text{MAX('highlight cells of interest')} - \text{MIN('highlight cells of interest')}$$

r^2 —the coefficient of determination is used in statistical models whose main purpose is the prediction of future outcomes on the basis of other related information. It relates to the proportion of variability in a data set that is accounted for by the statistical model.

Sample size (n or N)—the number of values for a given variable. In Microsoft Excel the *n* can be calculated by the following formula:

$$= \text{COUNT('highlight cells of interest')}$$

Standard deviation (SD)—the square root of the variance in the data (where the variance is the amount of variation within the values of a variable). In Microsoft Excel the SD can be calculated by the following formula:

$$= \text{stdev('highlight cells of interest')}$$



Standard error (SE)—the standard deviation of a distribution of a sample statistic, especially when the mean is used as the statistic. In Microsoft Excel the SE can be calculated by the following formula:

$$= \text{SD}/\text{sqrt}(n)$$

Statistical significance (p-value)—the confidence one has in a given result. Statistically significant effects (differences) are unlikely to have occurred by chance. This is generally shown through use of a *p*-value ($P < 0.05$ is generally considered statistically significant). The *p*-value is the probability of obtaining a test statistic at least as extreme as the one that was actually observed, assuming that the null hypothesis is true.

t-test—one of the most simple statistical analyses available. A one-sample *t*-test is used to test whether data is different from a base-line value. Two-sample *t*-tests assess whether the means of two groups are statistically different from each other. Paired *t*-tests are used where the means are derived from data of the same sample size, and unpaired *t*-tests where different sample sizes are used. *t*-tests can be easily calculated in Microsoft Excel using the 'insert' and 'function' commands, or using a statistical programme. The *t*-test generally has the notation:

$$t_{df} = \text{'a given output'}$$

Variables—variables of interest are commonly termed predictor (independent) or response (dependent) variables, to distinguish between two types of quantities being considered; the response variables are dependent on predictor variables. In *R*, variables can be factors, integers or numeric.

Minimum number alive (MNA)

MNA indices are relatively straightforward and can be used as a rough indication of abundance. They are not as robust as CPUE indices, but are easy to calculate and can be useful to describe trends. The index is simply the cumulative number of individuals observed/trapped during a given time period. In order to generate this index, animals must be able to be individually identified so that recaptures can be noted. MNA indices must be generated over a reasonable time frame, so that the assumptions of a demographically closed population are met—births, deaths, immigration, and emigration can cause significant biases in MNA indices.

To generate an MNA index from capture data:

1. Collate all captures from within one time period.
2. Tally the number of total captures.
3. Subtract the number of recaptures from the total number of captures. This gives you the number of different individuals trapped [$\text{MNA} = \text{Captures}_{\text{Total}} - \text{Recaptures}_{\text{Total}}$].



Example MNA analyses using pitfall trapping of skinks

The following hypothetical example uses data obtained by pitfall trapping skinks over 4 nights using 20 pitfall traps. The example explains how MNA indices are generated. In Table 1 the data show that five skinks were trapped during the first night; these skinks were marked and released. Four skinks were trapped on the second night and one of these had a mark from the previous night; the new animals were marked and released along with the previously marked animal. Three skinks were caught on the third and fourth nights and on each of these nights two individuals were recaptures, and one was a new capture; the new captures were marked and all individuals were released.

By totalling the number of captures and recaptures, an MNA estimate can be generated, as shown in Table 2. The cumulative captures came to 15 skinks and 5 skinks were recaptures. Therefore, only 10 individuals were caught over 5 days and this is the MNA for the study period. If this study were repeated each year for several years, using equal effort each year (same number of traps and trap nights), MNA could be used to indicate long-term trends in population size, as shown in Figure 6. From the data over several years it appears that abundance has increased between 2002 and 2008 (Table 3). A linear regression can be used to determine whether the trends are significant, and in this case the trend of increasing abundance is significant ($P = 0.028$).

Table 1. Hypothetical capture-recapture data from pitfall trapping skinks over 4 nights.

Trap date	Traps set	Captures	Recaptures (animals with marks)
5-Mar-02	20	5	0
6-Mar-02	20	4	1
7-Mar-02	20	3	2
8-Mar-02	20	3	2

Table 2. Hypothetical capture-recapture data from pitfall trapping skinks over 4 nights and showing estimates of the minimum number alive [MNA = $\text{Captures}_{\text{Total}} - \text{Recaptures}_{\text{Total}}$].

Trap date	Traps set	Captures	Recaptures (animals with marks)	Cumulative captures	Cumulative recaptures	MNA
5-Mar-02	20	5	0	5	0	5
6-Mar-02	20	4	1	9	1	8
7-Mar-02	20	3	2	12	3	9
8-Mar-02	20	3	2	15	5	10



Table 3. Hypothetical capture-recapture data from pitfall trapping skinks over 7 years (4 nights each year) and showing estimates of the minimum number alive [$MNA = \text{Captures}_{\text{Total}} - \text{Recaptures}_{\text{Total}}$].

Year	Traps set	Trap nights	MNA
2002	20	4	10
2003	20	4	9
2004	20	4	11
2005	20	4	13
2006	20	4	15
2007	20	4	16
2008	20	4	14

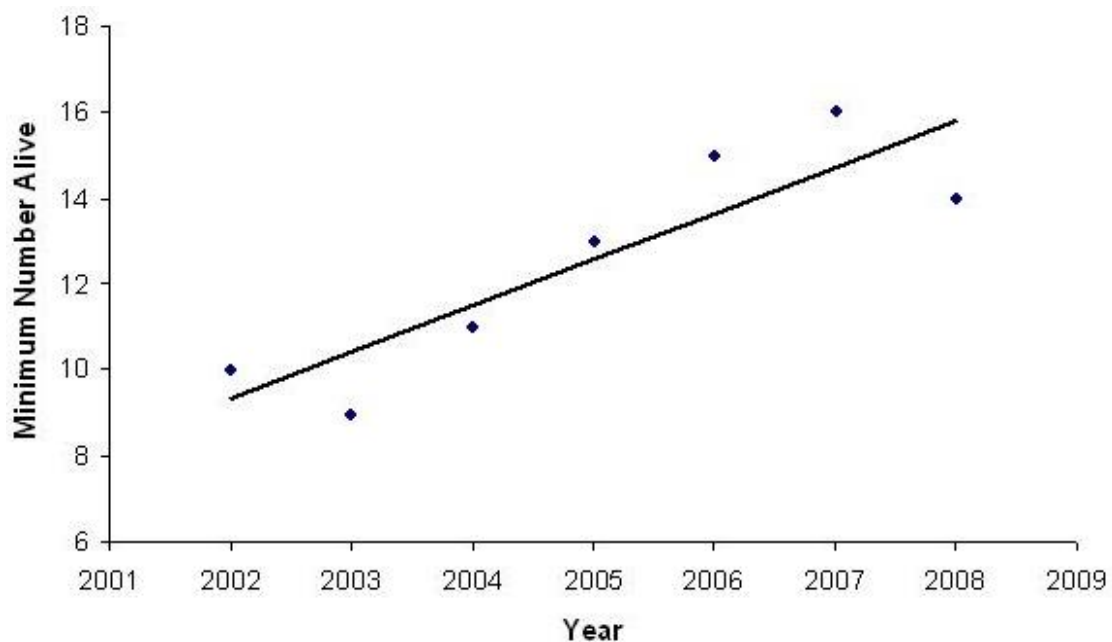


Figure 6. Hypothetical data for increase in abundance (minimum number alive) of skinks from 4 nights of pitfall trapping using 20 traps, over 7 years.

Catch per unit effort (CPUE)

CPUE indices are generated by dividing the number of animals captured by the effort expended. CPUE data are typically presented as number of captures per standard unit of effort. For example, pitfall trapping data are generally reported as $n/100$ trap nights and data from systematic searches as $n/\text{person}/\text{hour}$. The required data for CPUE are minimal (number of animals caught per capture event), but several optional variables may greatly aid in the interpretation of the results. Further, CPUE estimates assume that individuals are not removed from the population either by harvest or by death due to stress or injury from handling. Generally, multiple capture sessions will take place during each sampling period.



Where additional factors have been collected that are likely to influence CPUE estimates (e.g. changes in temperature during a trapping period), CPUE estimates can be corrected (see '[Example CPUE analyses corrected for temperature](#)' below).

Example CPUE analyses using pitfall trapping of skinks

The following hypothetical example uses data obtained by pitfall trapping skinks over a sampling period of 10 trap nights at Site A, where 50 pitfall traps were set each night (Table 4). Remember, CPUE for pitfall trapping is generally presented as number of skinks caught per 100 trap nights, i.e. for night one 50 traps are set and 20 skinks are caught. Therefore, the CPUE would be 40 skinks/100 trap nights as calculated by: $CPUE = (20 \text{ skinks}/50 \text{ traps}) \times 100 \text{ trap nights}$.

Note: If 20 traps were set, and eight skinks captured, CPUE would also be 40 skinks/100 trap nights.

Table 4. Hypothetical pitfall trap capture data over 10 trapping events in 1989 with catch per unit effort (skinks/100 trap nights) calculated.

Trap date	Traps set	Skinks caught	CPUE
06-Jan-89	50	20	40
07-Jan-89	50	15	30
08-Jan-89	50	23	46
09-Jan-89	50	18	34
10-Jan-89	50	26	52
11-Jan-89	50	12	24
12-Jan-89	50	14	28
13-Jan-89	50	21	42
14-Jan-89	50	28	56
15-Jan-89	50	19	38

Table 4 shows trapping data over the course of the 10 days, with the CPUE estimates ranging from 28–56 skinks/100 trap nights. The variability of CPUE over the 10 days can be used to give an average estimate of CPUE. The mean CPUE in January 1989 was 39.0 ± 3.3 SE (i.e. the average was 39.0 skinks/100 trap nights and the standard error around the average is 3.3). We can use this average as a more meaningful estimate to understand how the population size changed from 1989 to 1999 at Site A (Table 5). It is vital that trapping be undertaken in a similar manner, i.e. at the same site with the same distribution and density of traps and in the same season. In Table 5 hypothetical data are provided for pitfall trapping at Site A over 10 nights in 1999.



Table 5. Hypothetical pitfall trap capture data over 10 trapping events in 1999 with catch per unit effort (skinks/100 trap nights) calculated.

Trap date	Traps set	Skinks caught	CPUE
19-Jan-99	50	22	44
20-Jan-99	50	26	52
21-Jan-99	50	27	54
22-Jan-99	50	32	64
23-Jan-99	50	19	38
24-Jan-99	50	33	66
25-Jan-99	50	29	58
26-Jan-99	50	28	56
27-Jan-99	50	30	60
28-Jan-99	50	31	62

In January 1999, the mean CPUE was 55.4 ± 2.8 SE. As CPUE is available from both years and we know the variance of those estimates, we can say that significantly more animals were captured in 1999 than 1989 ($t_{18} = 3.802$, $P = 0.001$). Because the same number of traps were set each night in 1989 and 1999, the assumption of equal effort is met. The habitat did not change at the site over time, so we will assume that capture probabilities have not changed. However, differences in weather variables (e.g. temperature) between the two trap periods may influence CPUE indices, and these were not corrected for in these analyses, instead an assumption was made that weather was constant. We also assume that the relationship between CPUE and actual abundance is linear. If these assumptions of CPUE are met, then we can interpret this as an increase in population size over the 10-year period.

Example CPUE analyses corrected for temperature

Following on from the same hypothetical example as above, if temperature data were also collected during both trapping sessions in 1989 and 1999 (Table 6), then they may be included in analyses using linear models, and be used to 'correct' the data gained. For simplicity, this hypothetical example includes nocturnal skinks and therefore minimum overnight temperature is the most likely variable to influence capture rate (Table 6).

If we plot these data we can see that CPUE increases as temperature increases (Figure 7), and analyses confirm this relationship ($t_{16} = 10.002$, $P < 0.001$), with c. 93% of the variation in the data explained by temperature (Figure 7). As reported for the uncorrected CPUE estimates above, capture rates for the temperature-corrected CPUE estimates are also higher in 1999 than 1989 ($t_{17} = 2.543$, $P = 0.021$). Because temperature has such a large influence on the capture rate, we may wish to report a CPUE for each year that has been corrected for temperature. This can be done easily using a statistical programme such as *R* (ask a statistician for help). However, we can also do this in Excel, which helps to explain more fully the principles behind correcting data.



First, plot all the data of interest as a scatter graph as shown in Figure 7. Add a trendline and the equation that explains the line:

1. Right click on a data point and choose 'Add trendline...'
2. Under the tab 'Type' click on 'Linear'.
3. Under the tab 'Options' click on 'Display equation on chart' and 'Display R2 value on chart' (Figure 8).
4. Select 'ok'.

You now have an equation that can be used to calculate CPUE values adjusted for a given temperature. The equation shown on Figure 7 is as follows:

$$y = 0.1106x - 6.3179 \quad (\text{from the equation } y = mx + c)$$

where:

y = the temperature value

0.1106 = the slope of the line (m)

x = the CPUE value

-6.3179 = the y -intercept (c)

Therefore, for 6 January 1989, when the minimum overnight temperature was 10.5°C, the temperature-adjusted CPUE is:

$$x = (10.5 - 6.3179) / 0.1106$$

$$x = 37.8$$

The remainder of the CPUE can be adjusted in the same manner, and new means and SE calculated (Table 6). By correcting the data for temperature we can now report that the temperature-corrected mean CPUE in 1989 was 39.9 ± 3.6 and in 1999 it was 54.5 ± 3.3 (v. uncorrected mean CPUE (skinks/100 traps) of 39.0 ± 3.3 and 55.4 ± 2.8 in 1999).



Table 6. Hypothetical pitfall trap capture data from 20 trapping events in 1989 and 1999 with catch per unit effort (skinks/100 trap nights) calculated. Minimum overnight temperature is also provided (°C).

Trap date	Traps set	Skinks caught	CPUE	Min. temp	Adj. CPUE
06-Jan-89	50	20	40	10.5	37.8
07-Jan-89	50	15	30	9.8	31.5
08-Jan-89	50	23	46	12.0	51.4
09-Jan-89	50	18	34	10.0	33.3
10-Jan-89	50	26	52	11.8	49.6
11-Jan-89	50	12	24	9.0	24.3
12-Jan-89	50	14	28	9.2	26.1
13-Jan-89	50	21	42	11.0	42.3
14-Jan-89	50	28	56	12.9	59.5
15-Jan-89	50	19	38	11.1	43.2
19-Jan-99	50	22	44	10.8	40.5
20-Jan-99	50	26	52	11.8	49.6
21-Jan-99	50	27	54	12.9	59.5
22-Jan-99	50	32	64	13.1	61.3
23-Jan-99	50	19	38	9.9	32.4
24-Jan-99	50	33	66	13.0	60.4
25-Jan-99	50	29	58	12.6	56.8
26-Jan-99	50	28	56	12.8	58.6
27-Jan-99	50	30	60	13.2	62.2
28-Jan-99	50	31	62	13.4	64.0



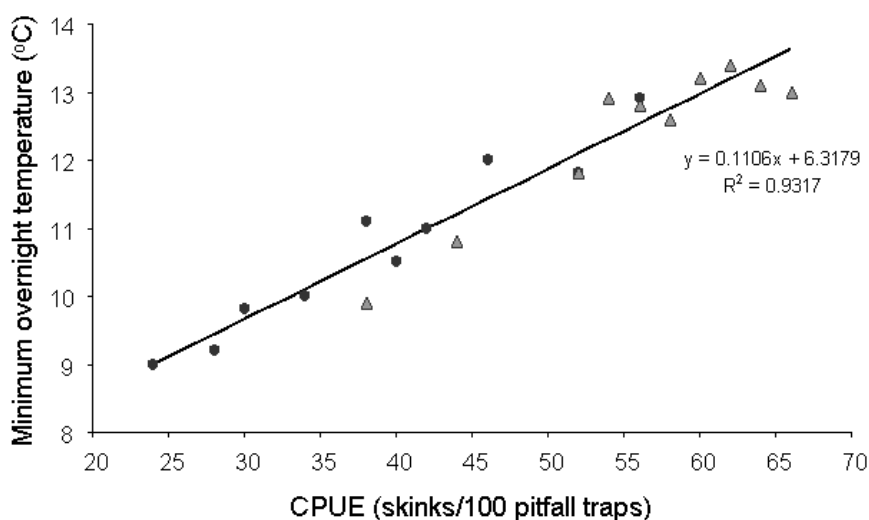


Figure 7. Hypothetical data for increase in catch per unit effort (CPUE) of skinks with an increase in overnight temperature. Data are from 20 trapping events during 1989 and 1999 using 50 pitfall traps for each event. The line on the graph shows the linear relationship between temperature and CPUE, and can be used to predict a CPUE given a certain temperature. The fit of the data to the line is commonly referred to as the r^2 , and shows that temperature accounts for c. 93% of the variation in the data. The equation explains the line. A residual is the difference between a given value predicted by the model (on the line) and the true value (i.e. the distance of each data point from the line).

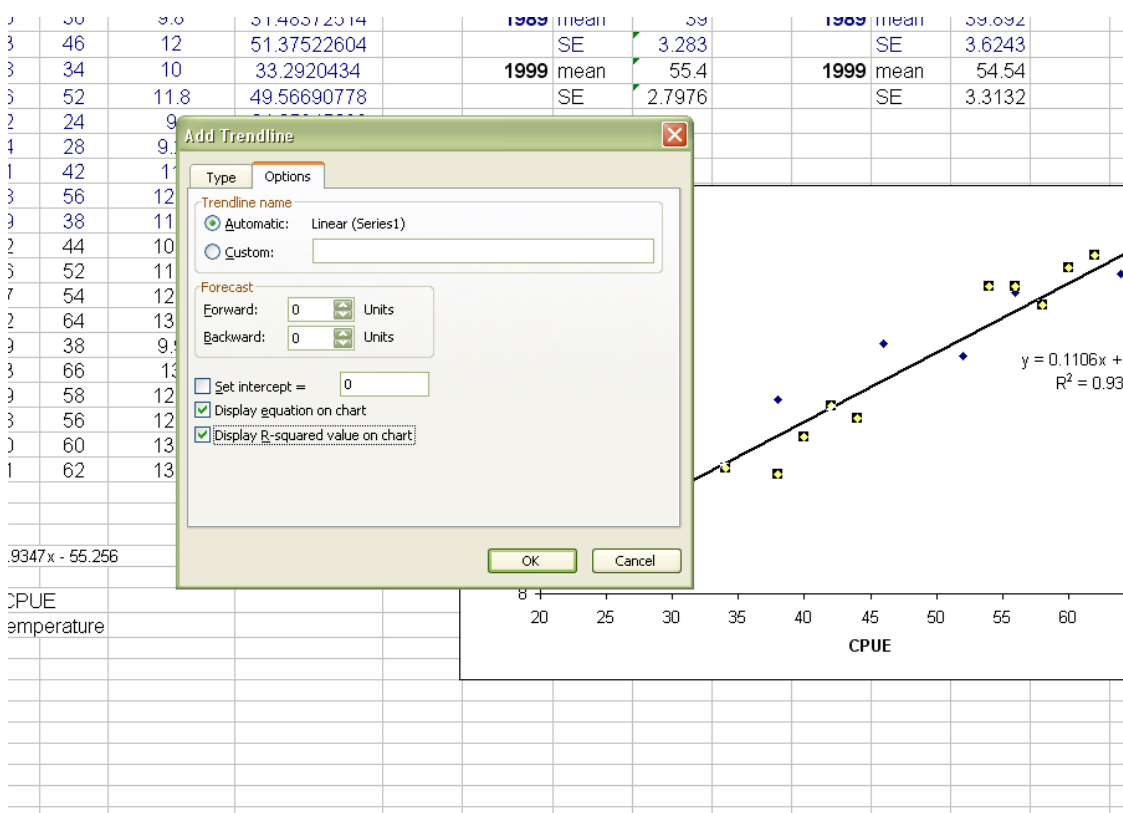


Figure 8. How to display an r^2 and line-equation using Microsoft Excel.

Density estimates

A crude density index can be generated by dividing the number of animals captured by the area searched. Although not the most robust method for generating density, as it does not account for detectability, it can be useful to have these data in some cases such as when broad differences in skink density among different habitats is required (e.g. East et al. 1995). It is *not* appropriate to extrapolate to much larger areas (e.g. an entire island) because density of animals varies among habitats; it is important to remember that density estimates should *only* be used to extrapolate within the same habitat. These indices are also based on the (risky) assumption that the sample represents a constant proportion of the population. Density estimates are generally calculated as number of individuals/hectare. Only use these density estimates where a rough and general guide is required. For more accurate density estimates, where detectability is accounted for, see 'Herpetofauna: population estimates' (docdm-833600).

Example density estimates using systematic searches of geckos

The following hypothetical example uses data obtained by visual systematic searching for geckos using five transects within a small section of low scrub on a 20 ha island (Table 7). It is assumed that the same search effort is used for each transect. Density is generally presented as number of individuals per hectare. In Table 7, 25 geckos are seen along transect 1, a 40-m long transect of bush that can only be effectively searched within 2 m of *each* side of the transect. This means the total area searched is 160 m² (40 m × 4 m) or 0.016 ha. Therefore, density of geckos within this habitat (assuming transect 1 is representative of the habitat) is c. 500 geckos/ha (density = 8 geckos/0.016 ha). Note: a more rigorous study would use data collected from multiple days of searching the same transects.

Table 7. Hypothetical data from systematic searching of five transects with density (geckos/ha) calculated.

Search date	Transect	Transect length (m)	Area searched (m ²)	Geckos sighted	Density (geckos/ha)
20-Feb-00	1	40	160	8	500.0
20-Feb-00	2	51	204	11	539.2
20-Feb-00	3	32	128	6	468.8
20-Feb-00	4	16	64	4	625.0
20-Feb-00	5	52	208	10	480.8

The variability of density among transects can be used to give an average estimate of the density within the low scrub habitat. The mean density for the five transects is 522.7 ± 28.2 SE (i.e. the average was 522.7 geckos/ha in scrub habitat and the standard error around the average is 28.2 geckos/ha).

Assuming that geckos are only found on low scrub on the island, we can use this average as a more meaningful estimate of the number of geckos on the island. That is, if 2 ha of the 20 ha island is covered in low scrub, and there are 522.7 geckos/ha of scrub, then there should be c. 1045.4 geckos on the island.



Density estimates can also be used to compare trends in abundance over time and with management. For example, if the island is undergoing revegetation (planting an additional 5 ha of low scrub) then managers may wish to know whether geckos are using the new habitat, and whether the total number of geckos has increased. It is important that the same search effort be used so that the assumption of equal effort is met. In Table 8 hypothetical data are presented 10 years after the first survey in 2010. The original five transects are used and five new transects created in the revegetated low scrub areas. At first glance it appears that fewer geckos are captured in the revegetated sites ($n = 23$) than the original sites ($n = 39$). However, by calculating the density of geckos for each transect, and averaging these within each site it becomes apparent that both sites of low scrub on the island have similar densities. The mean density of geckos in the original site is 504.2 ± 21.6 and for the revegetated site is 504.6 ± 31.6 SE, and these densities are not significantly different ($t_8 = 0.011$, $P = 0.992$). Similarly, we can say that density has not changed over time by comparing the mean density data from the original site in 2000 (522.7 ± 28.2 geckos/ha) and 2010 (504.2 ± 21.6 geckos/ha) which, despite having a lower density estimate in 2010, are also not significantly different ($t_8 = 0.521$, $P = 0.617$). Finally, the data collected in 2010 can be used to create a new estimate of number of geckos present in the 7 ha of low scrub on the island (c. 3531 geckos).

The habitat at the original site did not change over time, so we will assume that capture probabilities have not changed. However, differences in weather variables (e.g. temperature) between the two trap periods may influence density indices. We also assume that the relationship between density and actual abundance is linear. If these assumptions of density are met, then we can interpret this as a stable population over the 10-year period, which has increased in size as more habitat has become available. Finally, remember that density indices are based on the assumption that a fixed amount of searching effort will always locate a fixed proportion of the population and implies that the index is proportional to the density.

Table 8. Hypothetical data from systematic searching of ten transects with density (geckos/ha) calculated. Transects 6–10 are in revegetated low scrub and transect 1–5 are in original low scrub.

Search date	Transect	Site	Transect length (m)	Area searched (m ²)	Geckos sighted	Density (geckos/ha)
19-Feb-10	1	Original	40	160	7	437.5
19-Feb-10	2	Original	51	204	11	539.2
19-Feb-10	3	Original	32	128	7	546.9
19-Feb-10	4	Original	16	64	3	468.8
19-Feb-10	5	Original	52	208	11	528.9
19-Feb-10	6	Revegetated	21	84	4	476.2
19-Feb-10	7	Revegetated	15	60	3	500
19-Feb-10	8	Revegetated	26	104	5	480.8
19-Feb-10	9	Revegetated	17	68	3	441.2
19-Feb-10	10	Revegetated	32	128	8	625.0



Case study A

Case study A: long-term trends in a lizard community

Synopsis

Hoare et al. (2007) used CPUE to assess long-term trends in skink and gecko captures at the Pukerua Bay Scientific Reserve. The example demonstrates the utility of habitat data for interpretation of relative abundance indices, and the difficulty of interpreting CPUE data for species with low detectability.

The Pukerua Bay Scientific Reserve contains one of three remaining natural populations, and the sole mainland population, of Whitaker's skink (*Oligosoma whitakeri*), in c. 1 ha of habitat. Management of this reserve is targeted at the protection of Whitaker's skink (Department of Conservation 1996). Therefore, grazing stock were removed from the reserve in 1987 to reduce habitat destruction, and pitfall trapping was conducted between 1984 and 2006 to monitor trends in population sizes of five lizard species, particularly Whitaker's skink (Hoare et al. 2007). Copper skinks (*O. aeneum*), an ecologically similar species, were used as an indicator species for Whitaker's skinks, because capture rates of Whitaker's skinks were low (Towns & Elliott 1996).

Objectives

The study aimed to determine how lizard populations responded to the removal of grazing stock from the Pukerua Bay Scientific Reserve.

Two alternate hypotheses, raised soon after the removal of grazing mammals (Towns & Elliott 1996), were tested:

- The removal of grazing stock improves habitat so that species sensitive to predation and disturbance (Whitaker's and copper skinks and common geckos (*Hoplodactylus maculatus*)) increase in abundance over time.
- The removal of grazing stock increases the proliferation of seeding grasses, resulting in the periodic irruptions of mice. These irruptions lead to increased predation on lizards by mammals, and lizards decrease in abundance over time.

Sampling design and methods

Pitfall trapping

Pitfall trapping was conducted annually from 1984–2006 (excluding 1989, 1990, 1998, 1999, and 2003) in a 336 m² grid at the Pukerua Bay Scientific Reserve. Trapping intensified over the period of the study (Table 9), but always took place in summer, between January and March.



Weather and habitat

Weather data were obtained for each 24-hour trapping period (maximum and minimum air temperature, rainfall (mm), and sunshine hours). Mean annual weather data were also obtained from the National Climate Database to assess potential trends over time. Additionally, habitat in the trapping grid was surveyed in 1986 and 2006 to assess changes in vegetation structure.

Analyses

CPUE indices were generated for each year of monitoring and for each species over the 23-year monitoring period.

Results

In the 23-year monitoring period (1984–2006), 1693 lizard captures were made over 7597 trap nights (Table 9). Significant changes in CPUE were seen for copper skinks, Whitaker's skinks and common geckos over this period. Capture frequency of both Whitaker's skinks and copper skinks decreased between 1984 and 2006 ($P < 0.001$ for both species, ANOVA), but the capture frequency of common geckos increased ($P < 0.001$, ANOVA). The capture frequencies of common skinks and brown skinks showed no linear trend over time ($P = 0.24$ and $P = 0.65$, respectively). Local weather conditions during a trap night influenced the capture rates of some species, but there were no temporal trends in mean annual weather.

Substrate and vegetation were largely similar between 1986 and 2006, but subtle changes were found. Creeping vegetation increased from 0% to 4.13% on the surface and from 1% to 11.0% on the subsurface. The overall proportion of bare ground decreased from 11.5% in 1986 to 3.63% in 2006. Introduced veld grass (*Ehrhata erecta*), which was present elsewhere at the Pukerua Bay Scientific Reserve in 1986 (but not then at the study site), increased in cover to become the spatially dominant adventive species (58% of cover by adventive species).



Table 9: Pitfall trapping effort and lizard captures at the Pukerua Bay Scientific Reserve between 1984 and 2006 (modified from Hoare et al. 2007).

Year	Trap nights	Number of lizard captures				
		Copper	Whitaker's	Common	Brown	Gecko Common
1984	259	26	3	48	21	11
1985	333	11	2	38	3	3
1986	259	39	1	24	12	4
1987	222	49	6	97	31	1
1988	185	9	1	23	2	1
1989	0	-	-	-	-	-
1990	0	-	-	-	-	-
1991	36	3	2	13	3	0
1992	18	0	0	5	1	0
1993	18	3	1	11	1	0
1994	54	2	1	11	1	0
1995	107	3	0	36	1	1
1996	162	4	1	51	5	9
1997	150	2	0	13	3	6
1998	0	-	-	-	-	-
1999	0	-	-	-	-	-
2000	286	13	0	115	20	8
2001	479	3	1	103	9	13
2002	1596	10	0	28	15	51
2003	0	-	-	-	-	-
2004	1679	0	0	64	23	44
2005	1099	6	1	69	37	202
2006	655	0	0	131	29	39
Total	7597	183	20	393	880	217

Interpretation

CPUE of Whitaker's skinks and its indicator species, copper skinks, declined over the 23-year period from 1984 to 2006. Although CPUE of common geckos increased, the authors suggest this may have been due to changes in the times that the traps were checked. The results did not support the hypothesis that the removal of grazing stock increased the reserve's capacity to support Whitaker's skinks (Towns & Elliott 1996). Combined with the data on vegetation structure, CPUE indices support the hypothesis that removal of grazing stock increases the extent of introduced grasses and decreased abundance of Whitaker's skinks. Increases in the extent of introduced grasses lead to periodic irruptions of rodents (Newman 1994), and Hoare et al. (2007) propose that this led to increased predation rates on Whitaker's and copper skinks.

Limitations and points to consider

The study spanned 23 years, meaning that various factors changed and provided limitations that could potentially lead to both flawed results and interpretation. However, Hoare et al. 2007 identified these



limitations and subsequently considered many factors during the analysis and interpretation of their data, providing robust analyses and interpretation and pointing out any remaining limitations. These include:

- In long-term studies, equalising effort is often complicated, but critical to interpretation of the data. Different observers would have checked pitfall traps each year (or possibly even within a year) and may have had different competencies in identifying lizards. Therefore, Hoare et al. 2007 included observer as a factor in their analyses.
- The same trapping grid was used each year, but additional traps were added to the 336 m² grid in some years. Therefore, Hoare et al. 2007 removed the additional traps from the dataset for analyses, so that only captures in traps that were set every year were analysed. Whilst the number of trap nights varied between years, the number of traps set per night were equivalent. One estimate of CPUE for each trap night, and values across multiple nights in a year, were used for statistical analyses.
- Despite trapping effort being applied equally among species, the CPUE indices cannot be used to directly compare species. For example, it is not possible to say that common skinks are more abundant than brown skinks at Pukerua Bay, even though CPUE is up to four times greater. It is possible that capture probabilities for common skinks are much higher than brown skinks, so that a greater proportion of the common skink population is trapped.
- The capture rate of Whitaker's skink was too low to make robust assessments of change in population size (only two Whitaker's skinks were captured between 2000 and 2006; Table 9). When population sizes are very small and/or capture rates are extremely low (i.e. < 1/100 trap nights), it is difficult to detect and interpret changes in CPUE. Copper skinks are ecologically similar to Whitaker's skinks, and were used as an *indicator* species, but they cannot be used to *predict* what may happen with Whitaker's skink (see previous comment about comparing among species); only CPUE of copper skinks can be compared over multiple years.
- Although changes in vegetation were not very pronounced, the subtle changes in the amount of creeping vegetation and the change in composition of adventive species may influence microhabitat and therefore capture probabilities (e.g. Lettink & Seddon 2007). A limitation of CPUE is that it doesn't include capture probability in analyses (although variables that may influence capture rates such as temperature can be included); therefore, these should be considered and discussed when interpreting the trends in CPUE.

References for case study A

Department of Conservation. 1996: Conservation management strategy for Wellington 1996–2005. Department of Conservation, Wellington.

Hoare, J.M.; Adams, L.K.; Bull, L.S.; Towns, D.R. 2007: Attempting to manage complex predator-prey interactions fails to avert imminent extinction of a threatened New Zealand skink population. *Journal of Wildlife Management* 71: 1576–1584.

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.



Newman, D.G. 1994. Effects of a mouse, *Mus musculus*, eradication programme and habitat change on lizard populations of Mana Island, New Zealand, with special reference to McGregor's skink, *Cyclodina macgregori*. *New Zealand Journal of Zoology* 21: 443–456.

Towns, D.R.; Elliott, G.P. 1996: Effects of habitat structure on distribution and abundance of lizards at Pukerua Bay, Wellington, New Zealand. *New Zealand Journal of Ecology* 20: 191–206.

Full details of technique and best practice

Details of technique and best practice for designing and implementing surveys resulting in data for indices of abundance are explained here. It is important to remember that the *exact* methods will vary depending on the technique(s) used to capture/locate animals.

Planning a study

Choosing the study location(s)

Care should be taken to ensure that the survey location(s) (commonly called 'sites') are representative of the study site and/or habitats of interest. Use a random, probability-based sampling design (random sampling, systematic sampling, stratified sampling, etc.) to maximise inference and provide accurate variance estimates. It is advisable to think about the potential for long-term monitoring even if funding does not permit this in the short-term. Many useful papers are based on long-term data from multiple surveys (e.g. Hoare et al. 2007; Moore et al. 2007). If possible, sites (and grids within) should be permanently marked so the same locations can be revisited at each survey. At a minimum, take GPS locations and good photographs of the site.

How many counts are enough?

For statistical purposes, the number of counts (days/sites/samples) should be as high as possible, which is not always practical or financially feasible. For example, how many pitfall traps can one person visit within a day? How many transect lines can be deployed given the total search area and terrain? How many funnel/G-minnow traps or field workers can the budget cover? Below are some basic guidelines to assist survey designers, but in all cases the habitat, species and resources available may mean that you will need to be able to adapt. A pilot study is also useful before embarking on any surveys. Further, it is advisable for the inexperienced survey designer to talk with experts.

Survey days. At a minimum, for all indices of abundance at least 3 days of effort under optimal conditions should be used (see Hoare et al. 2010 for discussion). However, most studies aim to use more with 5–9 days being the usual number of survey days (e.g. Hoare et al. 2010; Moore et al. 2010).

Sites. Again, at least three sites (for each habitat type) are recommended in order to provide some statistical soundness; however, use common sense—if the site is a small island, for example, then three sites will not be possible.



Samples. For statistical analyses to have strength then sampling effort within each site should be, at an absolute minimum, five replicates (e.g. five transects), but more samples are strongly urged. Generally, for a given number of samples, it is better to have more points visited less frequently than fewer points visited more frequently (i.e. assuming that greater precision will result if variation between samples contributes substantially to the total variance).

Recommendation. 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 counts required. These are usually contingent upon local factors and chosen analysis methods. When planning a study, researchers should be aware that small changes are more difficult to detect than large changes.

Conducting the count

Collecting data

Indices of abundance all require similar methods for counting individuals, regardless of the capture technique(s) employed.

When collecting data for MNA and density analyses each individual needs to be uniquely identified or marked when first observed. These marks may be permanent over the life of an individual (e.g. PIT tags or photo-ID) or temporary, but lasting over the observation period (e.g. non-toxic markers). Marks need not be individual-specific, but individual marks are preferred if feasible as they can provide additional biological data (e.g. trap-happy/shyness, home range estimates, etc.). MNA indices must be generated over a short time frame (generally within a month, outside of the period in which females give birth/eggs hatch), so that the assumptions of a demographically closed population are met—births, deaths, immigration, and emigration can cause significant biases in MNA indices.

Recording data

Observations should be recorded on standard forms either at the time or transcribed at the end of each day onto the forms from field notebooks. Record all minimum attributes. Include any additional information that is deemed useful for future researchers so they can repeat the study using exactly the same methods.

Reducing variability

A large amount of variation in the number of herpetofauna observed/captured is created through various factors, many of which can be controlled. Therefore, a good survey will attempt to control for as many potential factors of variation as possible as this variation can obscure the real changes in herpetofauna numbers that the study is designed to detect. Make every effort to reduce the variability between counts by paying attention to the factors described below. Collect information on the factors that may influence variability, including weather, time of day, and observers. Statistical models may be used during the analysis to distinguish sources of variation in counts (e.g. [Case study A](#)).



Standardising conditions

For all counts, effort, time of day and season should be the same. Where possible, observers should all be of similar experience; habitat should also be similar. Weather can obviously not be controlled, but should be recorded in a sufficient manner.

Effort. The effort used to survey each site should be the same. For example, the number of person-hours or pitfall traps used within a given area should be identical. This is especially important where repeat surveys are being undertaken; the same locations should be explored with the same effort (e.g. person/hours, pitfall traps, etc.).

Observers. All observers should have experience with the species, habitat and/or method being employed. Observers will differ in their ability to see, catch, handle and identify herpetofauna and good training will help reduce some of this variability. Researchers should be aware that observer's abilities may increase or decrease with time and among habitat types.

Time of day. Counts should be undertaken at appropriate times of the day. For example, systematic searches for a diurnal, sun-loving skink should be done early in the morning or late in the afternoon (Whitaker 1994). If the best time of day is not known, then at a minimum surveys should be undertaken within the same time periods each day.

Season. Emergence of herpetofauna differs among seasons (e.g. frogs; Newman 1990) so counts should be done in the same seasons and within a short time period (e.g. 4 weeks maximum). This is especially important when repeat surveys are done over multiple years.

Weather. Counts should only take place on optimal catching days, which will differ depending on the species. For example, warm, wet nights (v. warm dry and windy nights) are good for systematic searches for frogs (Cree 1985). If the capture period is set, and weather conditions are not optimal, it is important to document this fully so that the results can be interpreted correctly. Counts should not be made during strong winds or heavy rain because these conditions affect both the behaviour of herpetofauna and the ability of observers to detect them.

References and further reading

- Bell, T.P. 2009: A novel technique for monitoring highly cryptic lizard species in forests. *Herpetological Conservation and Biology* 4: 415–425.
- Brown, N.P.; Moller, H.; Innes, J.; Alterio, N. 1996: Calibration of tunnel tracking rates to estimate relative abundance of ship rats (*Rattus rattus*) and mice (*Mus musculus*) in a New Zealand forest. *New Zealand Journal of Ecology* 20: 271–275.
- Clark, M.J.; Randal, J.A. 2011: A first course in applied statistics: with applications in biology, business and social sciences. Pearson Education, New Zealand.
- Cree, A. 1985: Water balance of New Zealand's native frogs (Anura: Leiopelmatidae). Pp. 361–371 in Grigg, G.; Shine, R.; Ehmann, H.; (Eds): *Biology of Australasian Frogs and Reptiles*. Surrey



- Department of Conservation. 1996: Conservation Management Strategy for Wellington 1996–2005. Department of Conservation, Wellington. Beatty, Chipping Norton, New South Wales.
- Driscoll, P. 1998: Counts of calling males as estimates of population size in the endangered frogs *Geocrinia alba* and *G. vitellina*. *Journal of Herpetology* 32: 475–481.
- East, K.T.; East, M.R.; Daugherty, C.H. 1995: Ecological restoration and habitat relationships of reptiles on Stephens Island, New Zealand. *New Zealand Journal of Zoology* 22: 249–261.
- Green, D.M.; Tessier, C. 1990: Distribution and abundance of Hochstetter's frog, *Leiopelma hochstetteri*. *Journal of the Royal Society of New Zealand* 20: 261–268.
- Hoare, J.M.; Adams, L.K.; Bull, L.S.; Towns, D.R. 2007: Attempting to manage complex predator-prey interactions fails to avert imminent extinction of a threatened New Zealand skink population. *Journal of Wildlife Management* 71: 1576–1584.
- Hoare, J.M.; O'Donnell, C.F.J.; Westbrooke, I.; Hodapp, D.; Lettink, M. 2010: Optimising the sampling of skinks using artificial retreats based on weather conditions and time of day. *Applied Herpetology* 6: 379–390.
- Lettink, M.; O'Donnell, C.F.J.; Hoare, J.M. 2011: Accuracy and precision of skink counts from artificial retreats. *New Zealand Journal of Ecology* 35: 236–246.
- 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.
- Moore, J.A.; Grant, T.; Brown, D.; Keall, S.N.; Nelson, N.J. 2010: Mark-recapture accurately estimates census for tuatara, a burrowing reptile. *Journal of Wildlife Management* 74: 897–901.
- Moore, J.A.; Hoare, J.M.; Daugherty, C.H.; Nelson, N.J. 2007: Waiting reveals waning weight: monitoring over 54 years shows a decline in body condition of a long-lived reptile (tuatara, *Sphenodon punctatus*). *Biological Conservation* 135: 181–188.
- Newman, D.G. 1990: Activity, dispersion, and population densities of Hamilton's frog (*Leiopelma hochstetteri*) on Maud and Stephens Islands, New Zealand. *Herpetologica* 46: 319–330.
- Newman, D.G. 1994: Effects of a mouse, *Mus musculus*, eradication programme and habitat change on lizard populations of Mana Island, New Zealand, with special reference to McGregor's skink, *Cyclodina macgregori*. *New Zealand Journal of Zoology* 21: 443–456.
- Russell, J.C.; Klette, R.; Chen, C-Y. 2010: Tracking small artists. *Arts and Technology* 30: 165–172.
- Taylor, B.L.; Gerrodette, T. 1993: The uses of statistical power in conservation biology: the vaquita and northern spotted owl. *Conservation Biology* 7: 489–500.



Towns, D.R. 1991: Response of lizard assemblages in the Mercury Islands, New Zealand, to removal of an introduced rodent: the kiore (*Rattus exulans*). *Journal of the Royal Society of New Zealand* 21: 119–136.

Towns, D.R.; Elliott, G.P. 1996: Effects of habitat structure on distribution and abundance of lizards at Pukerua Bay, Wellington, New Zealand. *New Zealand Journal of Ecology* 20: 191–206.

Whitaker, T. 1994: Survey methods for lizards. *Ecological Management* 2: 8–16.



Appendix A

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

docdm-760240	Herpetofauna: pitfall trapping
docdm-833600	Herpetofauna: population estimates
docdm-725787	Herpetofauna: systematic searches
docdm-146272	Standard inventory and monitoring project plan