BEST PRACTICE MANUAL

The Comparative Assessment of Arthropod and Tree Biodiversity in Old-World Rainforests

The Rainforest CRC / Earthwatch Protocol Manual

Second Edition

R. L. Kitching, S. L. Boulter, G. Vickerman,
M. J. Laidlaw, K. L. Hurley and P. S. Grimbacher

EARTHWATCH INSTITUTE
FINDING SOLUTIONS FOR A SUSTAINABLE FUTURE

Rainforest CRC
Cooperative Research Centre for Tropical Rainforest Ecology and Management

THE COMPARATIVE ASSESSMENT OF ARTHROPOD AND TREE BIODIVERSITY IN OLD-WORLD RAINFORESTS

THE RAINFOREST CRC / EARTHWATCH PROTOCOL MANUAL

Second Edition

Edited by
R. L. Kitching[1], S. L. Boulter[1], G. Vickerman,
M. Laidlaw[2], K. L. Hurley[2] and P. S. Grimbacher[1]

[1]Australian School of Environmental Studies, Griffith University
[2]School of Integrative Biology, The University of Queensland

Established and supported under the
Australian Cooperative Research Centres Program

ISBN 0 86443 740 4

Published by the Cooperative Research Centre for Tropical Rainforest Ecology and Management. Further copies may be requested from the Cooperative Research Centre for Tropical Rainforest Ecology and Management, James Cook University, PO Box 6811 Cairns, QLD, Australia 4870.

This publication should be cited as: Kitching, R. L., Boulter, S. L., Vickerman, G., Laidlaw, M., Hurley, K. L. and Grimbacher, P. S. (2005) *The Comparative Assessment of Arthropod and Tree Biodiversity in Old-World Rainforests. The Rainforest CRC / Earthwatch Protocol Manual – Second Edition*. Cooperative Research Centre for Tropical Rainforest Ecology and Management. Rainforest CRC, Cairns. (88pp.)

Cover Images ©
(Top) Terry Reis
(Centre) Roger Kitching
(Bottom) Guy Vickerman

March 2005

For copies of this document, please visit www.rainforest-crc.jcu.edu.au

PREFACE

Like many other nations, Australia is a signatory to the International Convention on Biological Diversity (Biodiversity Convention), which, among other things, commits the nation to monitoring the biological diversity within its borders. For invertebrates, and arthropods in particular, the sheer abundance and diversity of the groups involved makes this a challenging task requiring funding and expertise beyond that available even to the most prosperous of nations. Of all ecosystems, the World's rainforests are the richest and most diverse, and sampling and monitoring arthropods within the rainforest environment presents a huge challenge.

In the late 1980s and early 1990s Professor Roger Kitching, supported by numerous Earthwatch volunteers, started to develop new protocols. This work has continued with support from the Rainforest CRC since 1993. His aim was to define a set of techniques that can be used, in whole or part, in a standard fashion, allowing the assessment of the arthropod and associated vegetational diversity such that the results are comparable across sites, seasons and treatments. The goal of a 'complete' biodiversity inventory at present looks out of reach, but a rigorous comparative assessment, perhaps set against results from as large a group as possible of undisturbed sites, allows some of the more pressing problems associated with conservation and management to be answered. It has the advantage of being achievable over a modest time scale and of being open-ended and flexible in terms of just how much information may be collected from a particular site.

This manual presents a detailed account of Professor Kitching's approach, equipment, methods and handling of both specimens and numerical data. It is designed to be used by those with little formal biological training. A full survey involves a team of about fifteen enthusiastic volunteers and four or five more expert staff. Such a team can lay out a standard one-hectare plot, carry out a vegetation survey, arthropod sampling, sorting to Order and associated data entry in a two week period at a forest site. Repeat surveys at the same site will take proportionately less effort. The setting up of a simple but effective field laboratory close to the survey plot is an essential part of the process. This manual covers each stage of the process together with brief accounts of post-survey activities such as long-term specimen storage and statistical analysis of the data.

Professor Kitching and his Griffith University team have used and tested the sampling protocols at a range of sites from Lamington Park in southeast Queensland to Cape Tribulation in the Daintree Lowlands. He has also used the protocols in Malaysia, Thailand, Vietnam, China and Papua New Guinea. In doing so, he and his colleagues have trained hundreds of students and researchers in biodiversity assessment and conservation. This research has resulted in more than fifty scholarly publications in journals and books, many of which are listed in the references.

This manual is a standardised tool kit for biodiversity assessment and on-going monitoring. It is also an important step forward in helping biodiversity specialists to answer the kinds of questions that land managers, conservationists, governments and industry are asking about the changing nature of biodiversity in forested landscapes.

We commend Professor Kitching and his colleagues on their achievement.

Professor Nigel Stork
Chief Executive Officer
Rainforest CRC

Jane Gilmour AO
Chief Executive Officer
Earthwatch Institute (Australia)

CONTENTS

LIST OF TABLES

LIST OF FIGURES

ACKNOWLEDGEMENTS

The development of the survey protocol described in this manual occurred during a series of expeditions to research sites in Australia, Papua New Guinea and Borneo.

Early expeditions were supported by the Earthwatch Institute and the initial impetus for the work came from them. Subsequent expeditions were organised under the aegis of the Cooperative Research Centre for Tropical Rainforest Ecology and Management (Rainforest CRC) and attracted financial support from a variety of sources. Expeditions to Eungella, Conondale Ranges and Paluma were supported by the Australian Geographical Society, Queensland Government, Royal Geographical Society of Queensland and a host of other donors. Finally, support for our latest surveys in Papua New Guinea was derived from the Australian Centre for International Agricultural Research (ACIAR). We are deeply grateful to these agencies for the material support they have provided.

We also thank the many volunteers provided by Earthwatch who have enlivened our times in the field with their hard work and good humour. The Eungella survey was carried out by a team of students from Griffith University, and our work in Papua New Guinea was assisted by a team from the Madang Research Institute.

Pre-eminent botanical expertise on our surveys has been provided by Dr Michael Olsen (Lamington and Eungella), Mr Andrew Small (Atherton and Brunei), Dr Wong Khoong Meng (Brunei), Mr Robert Kiprianis (Papua New Guinea) and Mr Kipiro Damas (Papua New Guinea). Additional technical and research assistance have been most ably provided by Ms Heather Mitchell, Ms Tara Martin, Ms Jane Skrandies-Martin, Mr Terry Reis and Mr Rod Eastwood.

Our surveys have always required a base and the providers of support and sustenance at these bases have played an essential part in making our work possible: at Lamington – the O'Reilly family and the staff of the Green Mountains Guesthouse; at Atherton – Marie and Kevin Livingstone and the staff of the Lake Eacham Hotel; at Eungella – the staff of the Broken River Resort and the Pearson family; in Brunei – Ms Masnah bte Hj. Mirassan and the staff of the Kuala Belalong Field Studies Centre of the University of Brunei; in Papua New Guinea – the manager and staff of the Jais Aben Resort at Madang; and in Vietnam – the management and staff at Cat Tien Reserve Forest Park.

We are grateful to the Queensland Parks and Wildlife Service, Queensland Forest Service, the University of Brunei, the Brunei Museum and the Government of Papua New Guinea for providing appropriate permits for our research.

Professor Roger Kitching is grateful to Harvard University who provided him with the Bullard Fellowship during which a draft of the first edition of this technical report was completed.

Associate Professor Myron Zalucki and Professor Jonathon Majer provided constructive comments on an earlier draft of this Manual. This final version is much improved in consequence.

INTRODUCTION

SCIENTIFIC BACKGROUND

Entomologists have long been aware of the huge numbers of both individuals and species of insects that occur in natural ecosystems. Scientific and public attention was re-focused on this fact as recently as 1982 when Terry Erwin announced to the World his results from studies of the canopy faunas of a single species of rainforest tree in Panama. On the basis of his findings in Panama, Erwin suggested that the World's rainforests may hold up to thirty million species of insects and their relatives (the Arthropoda). Although much debate had occurred about this number, with more conservative answers around the eight to ten million range becoming more generally accepted, there has been little disagreement with the basic premise that the arthropods are a hugely diverse group and that most of that diversity is to be found in rainforests.

There has also been no disagreement with the observation that most of these species are formally undescribed and that the World's entomologists are too few to cope with the sheer volume of material in a 'traditional' manner of occasional collecting expeditions, the gradual accumulation of material in museums, and the subsequent production of monographs containing formal species' descriptions. Of course such activities should and will continue, but we need more rapid methods of assessment of biodiversity to fill the gap between these traditional activities and the demands of governments and others for near 'instant' information. This is especially the case for ecosystems such as rainforests, which are being cleared at such a rate that the opportunity to ever know much of their arthropod biodiversity threatens to also disappear.

The assessment of arthropod biodiversity is not merely a scholarly task of interest only to science. As has been clearly explained recently by Constanza and his colleagues (1998), the World's biodiversity provides humankind with an immense wealth of free service – from the maintenance of soil and water quality and the bases for pharmaceuticals, through to the raw material for highly profitable tourism operations. Constanza *et al.* (1998) estimate the value of these services ranges from US$16 to $54 trillion ($10^{12}$) per year. These astonishing estimates come on top of the establishment and general acceptance of the International Biodiversity Convention (1992) by most nations of the World. This convention requires that nations make every effort to prevent any further loss of biodiversity from within their borders, and that they develop inventory and monitoring programs to ensure that these efforts are successful.

When dealing with the species' level of biodiversity, the arthropods dominate life on Earth and make up a very large majority of all known and predicted species (Parker 1982). Accordingly, the development of practical procedures for estimating arthropod biodiversity is a high priority.

Since 1988 we have been involved with biodiversity estimation in rainforests, principally in Australia but also in Brunei in northern Borneo. Over the last fourteen years we have expanded early work on the canopy alone (see Kitching *et al.* 1993, 1998) to produce a protocol that samples arthropods from many components of the forest within a designated one-hectare plot. Our basic premise in designing our sampling protocol was that a complete inventory of any particular site was, practically speaking, impossible. We have focused, therefore, on producing a comparative procedure which, when applied in an identical fashion to different sites, or the same sites in different seasons, will allow statistical comparisons among data sets. The effort involved in application of our protocol can be increased or decreased in response to available resources or to tackle particular, more focused, ecological questions. For example, even though we advocate the simultaneous use of eight

different arthropod sampling devices, any subset of these, applied in the fashion we advocate, will produce data sets comparable across sites or times. Similarly, we are only too aware that additional sampling methods would target segments of fauna that are currently under-represented in our collections. Addition of extra activities within the overall protocol, or substituting for sampling methods less relevant to the particular ecological question, are of course a matter of choice by particular researchers. We do strongly advocate, however, that any overlap with the methods we have chosen be carried out in such a fashion that comparable results are obtained.

We have also chosen to carry out simultaneous tree surveys at our reference sites. We have done this because we find it difficult to think about the insects in particular without reference to the co-occurring set of plants. It is now well established that many of the key evolutionary radiations within the Insecta that have led to their immense success have occurred simultaneously with angiosperm radiations (Farrell 1998, Scoble 1992). Accordingly any attempt to explain why particular insect groups are as rich as they are at particular sites requires, in our view, simultaneous information on the plants. We have opted to survey all trees within our one-hectare plots, which at 1.3 metres have a diameter greater than five centimetres. Of course, this also generates valuable information on plant biodiversity. Other users of our protocol may choose to exclude trees from their surveys or choose a lower (but more demanding) size cut-off in their surveys.

We have entitled our set of methods the *Rainforest CRC / Earthwatch Protocol* because it has been developed from within Griffith University, Brisbane, a node of the Cooperative Research Centre for Tropical Rainforest Ecology and Management (Rainforest CRC) with major support from the Earthwatch organisation.

THE PLANNING PROCESS

As with most research, attention to detail in the planning stage, the preparation of equipment and on-site flexibility are vital to a successful outcome. Rainforests can be very demanding places to work especially when surveys are carried out in the wet season. We plan our procedures with the worst possible conditions – driving rain, low visibility and mud underfoot – in mind. In addition, a team of twenty people can do untold long-term damage to a site if proper precautions are not taken. Pre-survey visits to a site, the careful selection of the exact location for the plots, and the mental dexterity to imagine field conditions while preparing for the survey, are essential.

When employing volunteer labour, careful and extensive briefing is required before a team is taken into the field. It has been our experience that a full but non-technical explanation of the scientific reasons for the survey itself, the various methodological decisions made in planning, and any technical problems discovered along the way should be presented to a work team. Frequently answers to problems of a technical nature have come from our volunteer field workers themselves, based often on specialist knowledge of other fields – from engineering to dentistry – of which we biologists have little if any experience.

Surveys are team efforts and a team is more than the sum of its parts. This simple truism is the best guide to effective planning and leadership.

The most important part of each day of a field survey, accordingly, is the *Morning Briefing Session* in which jobs are assigned for the day and progress from previous days reviewed. The team leader must have a keen appreciation of the *whole* task of the survey team, ensuring that particular jobs start on particular days and that progress is made at an appropriate rate, and when required juggling working groups to suit skills, personalities and inclinations. This is not always easy.

PRE-FIELD PREPARATIONS

Equipment

One of the most time consuming parts of preparing for our surveys is planning and collating equipment. Many of our field sites are remote and a forgotten essential item of equipment can cause considerable frustration, friction and delay, especially when overseas and unfamiliar with local suppliers. In planning the survey, equipment should be organised as early as possible to allow for the manufacture of items, delivery of materials and transport of equipment. This also provides an opportunity to identify damaged equipment, missing parts and in the case of mechanical equipment, to have them serviced in preparation.

Where a survey is to be conducted overseas, it is also important to establish what can and can't be shipped to the site and similarly what can and can't be sourced in the host country. Items that are likely to cause shipping or purchasing problems include batteries and chemicals. We find that packing gear by trapping method is by far the best method. This means that a team responsible for a trapping method can be readily briefed on equipment use and prepared for the field on arrival.

Details of equipment for every method accompany the descriptions in the text of this manual. In addition to the equipment for each method, we also take spares of many things, the equipment to fix breakdowns and general equipment for a lab set-up. A complete equipment list is provided in Appendix 4.

Labels

Careful and accurate labelling of samples is absolutely imperative. The failure to label a sample in the field will render that sample useless once back in the lab. We pre-print all our labels in the office before a trip. Labels are printed on a laser printer. This ink will stay fast in ethanol and water. Writing labels in the field when wet is impossible, so pre-writing dates on labels at the laboratory facilities is important. Always use pencil to write labels as any other ink will run or dissolve in water and ethanol.

Two types of label are generally used. Firstly, a 'sample' label is placed with a collected sample in the field. This label includes the plot location, latitude and longitude of the site, trapping method, the trap number, the collection date and the researcher's name (Figure 1). These labels are approximately 30 mm x 15 mm in size, and about 96 labels will fit on an A4 sheet.

18° 59.30S 146° 11.00E
Paluma
Pitfall Trap – Small
PFS-… Jan…… 1999
R. L. Kitching

Figure 1: Example of a sample label, in this case to be put into collected Pitfall trap vials.

The second type of label is that produced to identify the taxa in sorted vials or for sorted and pinned moths. These labels include plot location, latitude and longitude of the site, trapping method, the trap number, the collection date and the researcher's name as per the sample label. In addition the taxa that has been sorted into an individual vial is added (Figure 2) or in the case of moths, a morphospecies number. One of these labels would be used for each taxa sorted from each sample. These labels are approximately 25 mm x 15 mm and about two hundred labels will fit on an A4 sheet.

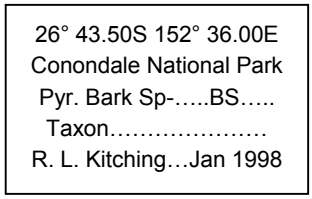


Figure 2: Example of a taxon label, in this case collected from a bark spray sample.

Volunteers

Determine the number of volunteers needed to complete the survey and the number that can be accommodated at the field site. We have worked in small teams of eight to ten people as well as larger groups of about twenty people when surveys where completed as part of a training course. Volunteers should be fully briefed prior to the trip on what equipment they need to bring with them. Ensure any volunteer paperwork is completed for insurance purposes and that any pertinent medical conditions have been identified, especially allergic reactions to stings or medication.

SAFETY

The need to consider safety in the field is a given for most experienced field researchers. The use of a volunteer workforce requires some additional planning and consideration, but general rules of bushwalking can be applied, particularly if the field site is remote to vehicle access.

- Always travel at the pace of the slowest team member.
- Carry sufficient first aid supplies for the worst case scenario and spread these among volunteers.
- Carry the best communications equipment you can access. Options include radios, mobile phones, satellite phones and EPRB (Emergency Personal Radio Beacon). Check the coverage in the area you are working.
- It is also advisable to keep volunteers working in pairs.

Different sites will require different levels of planning and equipment. For example, a site that requires six kilometres of walking to a mountain ridge in storm season requires some weather watching and planning on how to keep a big group of volunteers safe in the event of an electrical storm. Whereas a site close to a fully equipped resort and less than two hundred metres to a support vehicle might warrant a small first aid kit and a larger one kept in the car. Common sense and good planning is the key.

Keeping volunteers comfortable is also important. Ensure that all participants have been briefed on the need to carry wet weather gear, cool weather clothes, sufficient water, sunscreen, hats, sturdy shoes, insect repellent and, if stinging plants are a feature of the site, long pants. You may need to emphasise this each day and keep up to date on weather forecasts if possible. Watch for developing injuries and discomforts and change duties to suit. The most common injuries we have encountered are leech bites, tree stings, general scrapes and scratches, bruises, strains and blisters. Based on that, we would include in the first aid kit antiseptic cream, bandaids, waxing strips (for removal of stinging tree hairs) and if walking a long distance some blister pads. Blisters can cripple the most enthusiastic volunteer.

GENERAL RATIONALE AND SITE SELECTION

THE GENERAL DESIGN

The goal of our research program is the establishment of a series of more or less permanent one-hectare plots in rainforests along a latitudinal gradient from south-east Queensland (28°S) to the equatorial tropics of Borneo (4°N). To date (February 2004) we have set up ten such plots: six in Australia, two in Papua New Guinea, one in Vietnam and one in Brunei (Table 1). In addition, we completed a modified sampling regime in the Nothofagus forest within Lamington National Park, south-east Queensland. We are aware of the substantial potential for expanding this network further. Other networks of permanent rainforest sites have been established around the World for the study of tree diversity and phenology. Our sites complement these.

Table 1: The location and methods used in the Kitching one-hectare surveys.

Date	Location	Lats and Longs	Altitude (m)	Mean Annual Rainfall (mm)	Annual Max	Annual Min	Trapping Methods	Quarter Hectare	Forest Type	No. stems > 5cm	No. tree species
January, 1995	Lamington National Park, South-east Queensland	28ºS 153ºE	600	1622	31.2	2.8	YP, SPF, MAL, FOG, LL, BS, LGHT		Complex notophyll vine forest	1278	75
August, 1995	Kuala Belong Field Centre, Brunei	4ºN 115ºE	30	3900	22.3	31.8	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		Lowland Dipterocarp forest	1036	279
January, 1996	Robson Creek, Far North Queensland	17ºS145ºE	686	1394	29.9	7.6	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		Complex notophyll vine forest	1163	195
July, 1996	Lamington National Park, South-east Queensland	28ºS 153ºE	600	1622	31.2	2.8	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		Complex notophyll vine forest	1278	75
January, 1997	Eungella National Park, Central Queensland	22ºS 148E	720	1699	34.8	6.4	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		Simple/complex notophyll vine forest	1983	49
Jun/Jul 1997	Kuala Belong Field Centre, Brunei	4ºN 115ºE	30	3900	22.3	31.8	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		Lowland Dipterocarp forest	1036	279
January, 1998	Connondales National Park, South East Queensland	26°43'50"S 152°36'00"E	550	1345	34.2	3.7	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT	Yes (LL, YP, Veg)	Complex notophyll vine forest	1380	50
Jun/Jul 1999	Baitabag, Papua New Guinea	5°08'31"S 145°46'37"E	60	1972	30	23.1	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT	Yes (Veg, YP)	Complex mesophyll vine forest	1039	152
July, 1999	Lamington National Park, South-east Queensland	28ºS 153ºE	600	1622	31.2	2.8		Yes (LL)	Complex notophyll vine forest	1278	75
March, 2000	Cape Tribulation, Far North Queensland	16º07'30"S 146º26'30"	81	2500	28	22	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT	Yes (Veg, YP, LL)	Complex mesophyll vine forest	1538	135
July, 2000	Oomsis, Papua New Guinea	6º40'30"S 146º4'00"E	65	1979	32.3	21.6	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		Medium crowned lowland hill forest	1020	121
January, 1999	Paluma National Park, North Queensland	18º57S 146º11E	1000	2532	19.1	29	YP, LPF, SPF, MAL, FOG, LL, BS, LGHT		simple notophyll vine forest	2093	78
July, 2002	Cat Tien National Park, Vietnam	11º26'32"N 107º20'17"E	200				YP, LPF, SPF, MAL, FOG, LL, BS, LGHT, FIT	Yes (Veg)	lowland tropical forest	1123	78
January, 2004	Lamingon National Park, Sth East Qld	28º 15' 59.6" S 53º 10' 25.9" E	1165	1622	31.2	2.8	YP, LPF, FIT, FOG, LL, BS	No	*Nothofagus* forest	N/A	N/A

YP = Yellow Pan, LPF = Large pitfall, SPF = Small Pitfall, MAL = Malaise traps, FIT = Flight Intercept, FOG = Canopy knockdown, LL = Leaf litter, BS = Barkspray

We chose sites in remnants of mainly undisturbed forest as much as possible. We view our current data sets as baselines for future comparative work where we will examine the impacts of various natural and human-induced disturbance on biodiversity. The sites act as resources for more detailed studies of particular ecological processes, for which a well mensurated area of forest, with trees located and identified, increases the options for future research substantially.

At each site we have carried out at least one wet-season survey of both the vegetation and the arthropod fauna. The arthropod survey uses up to eight different trapping or sampling methods, each one of which is replicated between three and forty times within the one-hectare plot. Sampling locations within a plot are randomised (as far as is practicable). Our arthropod samples, then, give us measures of mean numbers of individuals and taxa with a statistically valid measure of variability about the mean. This allows us to compare one site with another using a variety of standard statistical procedures. It does not, however, allow us

to generalise about biodiversity at a particular latitude – this would require a series of plots matched for forest type within the same region. Of course our protocol would be ideal for the establishment of such sets of replicate plots but, to date, we have not had sufficient resources available to carry out this highly desirable work.

At each site we carry out a complete survey of all trees greater than five centimetres diameter at 1.3 metres height. This is a complete count rather than a sample of the trees within the plot. Accordingly it does not generate information that can be compared *statistically* across plots at different locations – although simple side-by-side comparisons are often very illuminating. In order to obtain some measure of variance within the chosen site that will allow such statistical comparisons on our plant data, we have begun surveying two additional one-quarter hectare plots in the vicinity of our main plot at each location. These quarter hectare plots are chosen to be of the same forest type as the main plot and are located within five to ten kilometres of that plot. Tree data from these smaller plots can then be combined with that from a quarter hectare subset of the main plot to allow the calculations of means and densities of some vegetation measures. We have carried out limited arthropod surveys in these quarter hectares also.

The survey has the following components:

1. Plot establishment and marking.
2. Vegetation survey.
3. Insect trapping.
4. Data sorting and analysis.

SITE SELECTION

As indicated we have chosen sites that more or less represent undisturbed forest. This decision is based on the documented history of a site (e.g. logging records) and also on local knowledge, particularly when working in areas where land is owned and managed by Indigenous people. Added to this are some general principles involved in site selection that are important whenever the Protocol is applied.

Accessibility

The biodiversity survey of a forest plot involves a substantial amount of equipment, none of which is very large, but some of which can be awkward to carry long distances. The awkwardness is exacerbated if travel involves traversing steep country, crossing major rivers or is through dense secondary vegetation. Accordingly, we look for sites where there is access along a four-wheel drive road or by boat to within approximately a kilometre of the plot. We use existing tracks wherever possible to bring us to the plot or establish a single well-marked track, following land contours where feasible. In the case of our Nothofagus survey, there was no way to avoid walking at least seven kilometres to our site. In this case extremely good planning was essential.

Adjacency to Laboratory Facilities

We have used our field teams to sort all material collected, to the level of Order in most cases, while the team is in the field. Accordingly we have set up simple laboratories within easy walking or driving distance of a plot. Sometimes we have had the luxury of using existing field stations set up for research purposes. At other times we have moved trestles and microscopes into the lounges of hotels, unused storage sheds, or cabins on campsites and established our laboratory there. We do not advocate trying to set up such a facility

under canvas, although tents for living accommodation can surround some more permanent structure used as a laboratory. Electricity is essential for such a laboratory, either through attachment to a public grid, through connection to a hotel or field station generator, or *in extremis,* by use of smaller portable generators. This last option is to be avoided as much as possible, involving as it does the need for generator upkeep, fuelling and so forth. Whatever the arrangement, we advocate that the laboratory be no more than an hour's travel time (by whatever means) to the field site.

Environmental Uniformity

Any hectare of forest will contain light gaps, patches of old secondary forest, minor ridge tops and drainage channels with or without streams in them. These variations are both inevitable and part of the forest being studied. In general though, we have avoided plots that contain, for example, major soil boundaries within them, or that span waterways or forest pools. This can usually be foreseen by prior examination of topographical and pedological maps (where these exist).

Topography

The ideal site is flat, but this is seldom available. We have used sites in Borneo in which less than half the hectare was uniformly flat, the remainder becoming a series of deep gully systems. This made the survey of vegetation, in particular, much more difficult and increased the risk of undesirable environmental impact on the landscape. Nevertheless, where there is choice we advocate selection of the flatter of any available sites.

PLOT ESTABLISHMENT

CARTESIAN COORDINATE PLOT

The standard plot we use is a 100 m x 100 m square. We begin our surveys by marking the corners and the centre, then laying out a grid of pegs at ten metre intervals. We designate the bottom left hand corner of the plot as our origin (00,00) point. This imposes on the hectare a Cartesian coordinate system and allows any point within the plot to be specified as a four to six digit code (e.g. 30,60 which is thirty metres from the origin on the horizontal (x) axis and sixty metres from the origin along the vertical (y) axis) (see Figure 3). In this way, each trap can be positioned as close as practical to a position dictated by randomly generated coordinates. Marking the plot involves placing 121 pegs regularly across the plot and is the first priority in beginning a survey. Indeed, if this can be done (with a smaller team of people) before the main survey commences, this is a major advantage and allows vegetation and arthropod surveying to begin on Day 1.

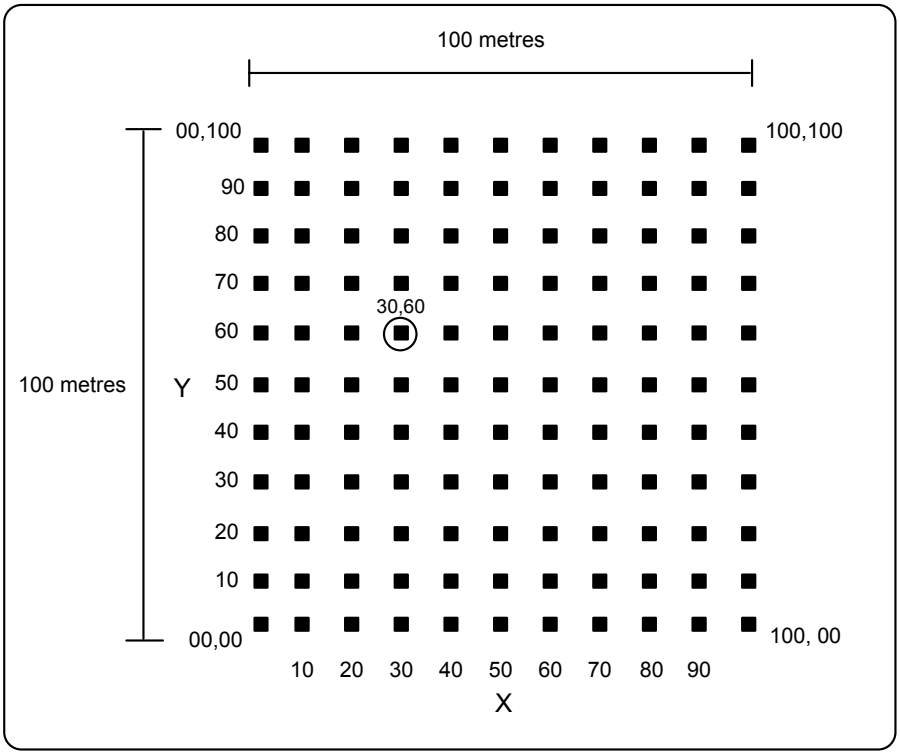


Figure 3: Final layout of 100 m x 100 m plot showing Cartesian coordinate system.

Equipment Design and Preparation

In previous surveys we have used wooden garden stakes as our pegs. We paint one surface of the top of each peg white as a base for labelling and each peg has its x and y coordinates written upon this white surface with a waterproof marker. In the humid, wet conditions of the rainforest, pegs quickly decay and on return visits several years later many of the pegs may have been lost, fallen over or badly decayed. One of the authors (M. Laidlaw), who continues to visit a number of the plots to perform further survey work, has devised an alternative style of peg. Wooden pegs are replaced by one-metre lengths of PVC conduit into which a small hole is drilled at one end. We then wire aluminium tree tags through this hole. The tags are

pre-punched with coordinates (e.g. 30,60). Putting the post into the ground is done using a modified post rammer.

In the Field

In laying out a plot, the first task is pegging out the baseline – the x-axis of the plot. Conventionally, we run this west to east from the origin (00,00). Making sure this line is straight is both important and difficult. The surveyors' method of laying the line bare over the whole one hundred metres simply does not work in forests. Gullies, patches of thorns, and even large trees get in the way. We work with a fifty-metre tape and align points by eye, dropping pegs into position as their location is established.

Once the first fifty metres of the baseline are established, the centre of the plot can be marked using the 3:4:5 rule of geometry. This is easier and more accurate than using a sighting-compass, Dumpy level or theodolite. We have tried these more sophisticated methods and find any increase in accuracy they may give is out-weighed by the disadvantages of weight, positioning them on wet slopes, or relative complexity of use. Once the baseline and the centre point are established and marked, the remaining fixed points within that quarter of the plot can be added by aligning them with existing known markers. The remaining quarters of the plot are marked out in similar fashion – first by identifying the corners from the established baseline and then by infilling the points row by row. We have found that orienting all the pegs so that the white face or the aluminium tag is parallel with the x-axis is a major aid to anyone becoming disoriented in the forest.

The typical characteristics of rainforest topography inevitably mean that every 10 m x 10 m section of the plot is unlikely to be exact in squareness or dimensions, but the amount of error this introduces into the results is not great. Once the marking out of the grid is completed, any point within the plot can be located to about a ± one-metre error. We mark all posts on the boundary lines (i.e. x = 0, y = 0, x = 100, y = 100) with yellow flagging tape and we mark the fifty metre lines, horizontally and vertically, in a similar fashion. This, along with the actual coordinates marked on the posts/tags and the north/south orientation of the pegs, aids rapid and accurate navigation within the plot.

Quarter hectare plots are set up in a similar fashion.

The corners of the plot should be located using a Global Positioning System so that it can be indicated on a standard map. Where forest canopy obscures a clear GPS reading, then some nearby point where readings can be obtained should be established and the plot's (00,00) point located with respect to that point by distance and bearing.

The setting up of a plot in this fashion will take a team of about four people up to two days. Steep topography increases the difficulties considerably. Once a number of 10 m x 10 m squares are clearly marked, the vegetation survey can begin. The vegetation survey and the later stages of marking out of a plot can go on simultaneously. Arthropod sampling, however, cannot begin until any randomly generated (x,y) point on the plot can be located confidently and therefore must await the completion of the marking out process.

Equipment List

Using timber stakes:

- 121 CCA treated 1500 mm x 25 mm x 25 mm timber stakes; one end painted white
- white paint (lab use)
- 20 permanent marking pens (e.g. 'nikko pens')
- mallet
- compasses
- 50 m tape measures
- 5 rolls yellow flagging tape

Using PVC stakes:

- 121 x 1 m lengths of electrical conduit with a hole drilled in one end
- 121 x tree tags and wires
- a set of punches
- hammer (for punches)
- post rammer
- compasses
- 50 m tape measures
- 5 rolls yellow flagging tape

POLAR COORDINATE PLOT

Our recent survey in the *Nothofagus* forest of Lamington National Park presented logistical problems we had previously avoided. The need to walk seven kilometres to the plot, up a steady grade, rendered the carrying of 121 stakes into the field undesirable and difficult. We also had some time restrictions, a limited volunteer pool and this was essentially a pilot study of the *Nothofagus* environment. We chose in this case to use a circular plot and use polar coordinates to locate our traps. The setting up process of permanently staking the plot was avoided and traps where positioned using randomly generated polar coordinates. So for example a randomly generated point might be 13º, 25 metres. One team member using the compass directs the second person trailing a tape measure along a transect at 13º. Once the second person is 25 metres from the centre point the trap is put into place. While there is definitely some drift from the exact degree while clambering around and between vegetation, we found this technique more than adequate.

THE VEGETATION SURVEY

GENERAL

The vegetation survey is the most time-consuming element of the field survey. We have routinely used a team of at least four people to survey the trees. This team tackles each of the 10 m x 10 m quadrats in turn until all one hundred quadrats are completed. Multiple teams can speed the process up by working simultaneously in different parts of the one-hectare plot. Effective communication between these teams is, of course, vital.

Equipment Design and Preparation

The field equipment for the vegetation survey is fairly simple and the list follows after this section. We have found it useful to use waterproof paper for pre-printed data sheets. In torrential rain complete sheets can be ruined. A proforma data sheet is provided in Appendix 2.

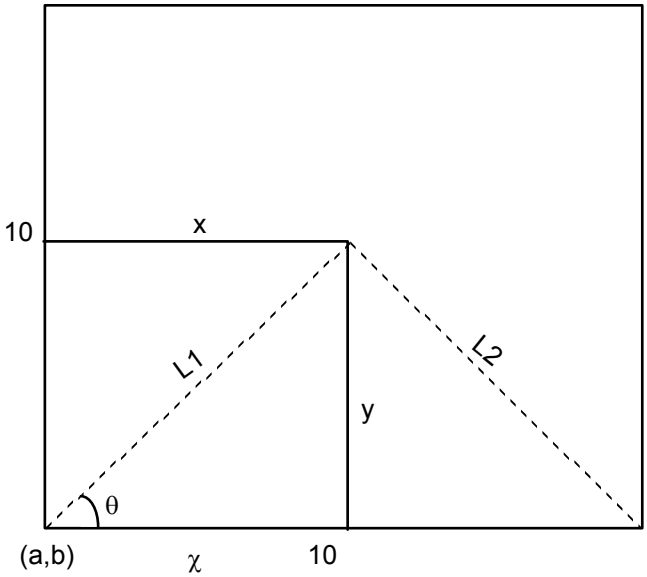
In the Field

Volunteers are organised into two teams. The first, 'the survey team' is responsible for attaching a temporary label of flagging tape on each tree, for measuring the diameter of the stems, estimating stem height and obtaining the position of each stem within each quadrat using x and y coordinates for every stem equal to or greater than five centimetres diameter at breast height (dbh; 1.3 metres from the ground on the uphill side of the bole) within each 10 m x 10 m quadrat. The second team is made up of a consulting botanist and an assistant. They are responsible for the identification of the stems to species or morphospecies. The botanist's assistant's role is to write things down, look for fallen leaves, throw branches at trees to knock fruit down, or merely to act as a discussant for the various identification points involved!

The following data is therefore collected within each 10 m x 10 m quadrat as follows:

- Each stem ≥ 5 cm dbh should initially be labelled with flagging tape. With a permanent marker, each stem is marked following a convention of (x, y) 1, (x, y) 2, (x, y) 3, etc., where x and y are the coordinates on the bottom left hand corner post of each quadrat. If the plot is to be resurveyed at a later date, permanent botanical alloy tags can replace these temporary labels. If the survey is to occur only once, the flagging tape should be removed once the trees have been identified.
- We treat any coppice stems ≥ 5 cm dbh as separate individuals. Only those stems rooted within a quadrat are recorded for that quadrat. Where a stem straddles two adjacent quadrats, the data may be recorded for either quadrat.
- The diameter at breast height (1.3 metres from the ground on the uphill side of the bole) of all stems ≥ 5 cm in diameter should be measured using either a diameter tape or a cloth tailor's tape (the measurements in the latter case must subsequently be converted from girth to diameter). Any deformities in the trunk at 1.3 metres from the ground should be avoided by passing the tape just above or below them. The tape should be passed gently underneath any vines of epiphytes on the stem. Where large buttress roots are present, the stem diameter should be taken directly above the buttresses by way of a ladder or other climbing equipment.
- The height of each stem can be estimated by eye. More accurate measurements can be taken using Abney levels or a clinometer to sight the top of each tree from a known distance. These methods are very time consuming and often impose demands of visibility that are not easily met. We have found that visual estimates give reliable and analysable results.

- The x and y coordinates of each stem can be obtained by measuring the distances from each stem to each of the bottom corners of the quadrat (Figure 4).



$$\text{Cos } \theta = \chi / L1$$

$$\text{and Cos } \theta = \frac{L_i^{\,2} + 10^2 - L_2^{\,2}}{2L110}$$

$$\text{so: } \chi = \frac{L_i^{\,2} + 10^2 - L_2^{\,2}}{20}$$

$$\text{y} = \sqrt{(L1)^2 - x^2}$$

Figure 4: The calculation of x and y coordinates within a 10 m x 10 m quadrat.

Special Notes

To maintain consistency in data recording, a single team member should be designated as 'scribe' and numerical results called out to that person. These should be recorded on pre-printed data sheets (Appendix 2), preferably on waterproof paper as mentioned above. If waterproof paper is not available for the datasheets, the scribe should protect themselves and the datasheets as best as possible from any rain or general moisture whilst recording data. An all-enveloping waterproof cover or umbrella may help in this regard. Once back in the laboratory such records should be transcribed as soon as possible (we enter them directly onto a laptop computer) while the field-group's memory of details persists. Damp data sheets need to be carefully dried and stored.

Equipment List

Equipment provided to each survey team includes:

- permanent marker pens
- yellow flagging tape
- log for recording x,y coordinates, species and dbh of each tree (≥5cm dbh) made from pre-printed waterproof paper is recommended (see Appendix 2). One page per 10 m x 10 m quadrat
- 2 x tape measures at least 10 m long
- 2 x seamstress tapes (to measure dbh)
- pencils
- clipboards

In the lab we also have a plant press and newspaper on hand for collected specimens.

ARTHROPOD SURVEY

GENERAL

We have selected a set of eight trapping methods (Table 2) that target a wide range of forest components from the leaf litter to the high canopy. This is not an exhaustive set. We do not target the fauna of any streams or other aquatic situations, the soil fauna, the fauna of dead and dying wood, the fauna of fungi or the fauna attracted to particular baits such as dung, carrion, pheromones or fruit. Techniques do exist for sampling all of these and other segments of the fauna, and surveys with particular target faunas may be expanded to include any them. Southwood's (1978) textbook describes the many ways in which this can be done.

Of course additional sampling methods will add to the field effort and to the sorting demands. In the accounts that follow we provide a few introductory references for each trapping method, mostly drawn from Southwood's (1978) account. We have tried to include reference to the earliest use of the particular method, plus the more accessible papers that describe the advantages and disadvantages of the particular method. Most of the methods are widely used and each has a large associated literature. The references we give should provide an entry point to this wider literature.

Table 2: Summary of trapping methods, number of replicates and days of trap operation.

Method	Number of traps/samples		Number of days
	Ground	***Canopy***	
Leaf litter extraction	10	Nil	single collection
Pitfall traps - small	4 arrays of 9	Nil	4
Pitfall traps - large	4 arrays of 9	Nil	4
Yellow pans	10	Nil	4
Flight intercept traps	10	Nil	4
Malaise traps	3	3	4
Light traps	3	3	5
Canopy knockdown	Nil	3 (20 hoops each)	

CANOPY SAMPLING

For three of our sampling procedures – malaise trapping, light trapping and canopy knockdown – trapping devices need to be placed in the canopy. We have investigated many ways of doing this but have settled on the use of a bow and arrow as the most efficient and reliable way of placing ropes into the canopy. Other methods of placing lines in the canopy include the use of slingshots, cross bows, or even naval style line-throwing guns. A forty to fifty pound compound bow is used with a fishing reel attachment fastened to the front just below the arrow ledge. We modify standard heavy arrows by attaching a length of fishing 'leader' along the shaft. Fishing line is then attached to this leader using a simple swivel such that when the arrow is fired the line slides down the arrow and ends up being pulled into the canopy. Attaching the fishing line to the front of the arrow would of course, simply cause it to tumble out of control. Finally we add a rubber stopper to the tip of the arrow such that if it strikes a branch or other obstruction it will bounce off rather than remain stuck in the canopy. With a little practice lines can be dropped over almost any required branch. The fishing line is used to pull heavier line into the canopy (we use 'sash cord' for this purpose) and then this thicker line is used to pull a rope of an appropriate weight over the branch. When joining one

line to another we usually smooth over the knot with plastic tape so that it is less likely to become jammed while passing through foliage. Of course there is not always an appropriate near-horizontal branch at the randomly selected (x,y) point where we wish to place a particular trap. In this case we choose the nearest appropriate branch to the selected point, noting the new (x,y) coordinates for our records afterwards.

LEAF LITTER EXTRACTION

There are a variety of devices invented by soil zoologists by which the animals present in a volume of soil or leaf litter can be extracted, more or less efficiently, for subsequent study. Most of these are based on the observation that animals will move away from a heat source when this is applied above a mass of soil or litter. We have used the simplest of these devices, the Tüllgren funnel. Invented in Sweden by Tüllgren in the early twentieth century, it has been much improved over the years. Modern designs are generally based upon those of Macfadyen (1953, 1955). This author has written the definitive comparative accounts of the various methods of sampling soil and litter animals (see Macfadyen 1955, 1962). Ford (1937) was the first to use close packed arrays of Tüllgren funnels for extracting the fauna of many samples simultaneously. Paris and Pitelka (1962) discuss the many factors affecting the efficiency of use of Tüllgren funnels in describing their surveys of isopods.

Equipment Design and Preparation

Our array of funnels is contained within a custom built insect-proof box with a hinged lid and removable metal legs at each corner (Figure 5). Our funnel equipment was constructed by our colleague Denis Rodgers and has been designed to be light for portability whilst being robust enough to withstand field conditions. Within this box ten forty-centimetre plastic funnels are inserted with their stems emerging from holes in the base of the boxes. Each funnel contains a coarse mesh disk close to the top of the stem, inside the funnel itself. This prevents excessive amounts of litter fragments contaminating the samples of extracted arthropods. Above each funnel, suspended from the box lid is a forty-watt light bulb connected to a power source. Outside the box the funnel stems are attached to removable vials. Ordinary seventy millilitre vials or sample jars are used and the lid of one such vial is held on the end of the funnel stem by a tight fitting rubber ring. The base of a vial can then be screwed into the lid and effectively sitting under the funnels' stem. The funnels can be taken out and packed into the cabinet along with its legs for portability.

In the Field

Samples are collected from ten random points in the one-hectare plot (and five within each quarter hectare plot). Each comprises a litre of moist leaf litter scraped up from around the selected point using a plastic one litre container. Wear gloves for this job. We restrict our samples to the litter itself and avoid, as far as possible, including any soil. Larger branches and wood fragments are discarded. These samples are placed into sealable plastic bags in the field with an accompanying sample label which records the sample number and (x,y) coordinate.

On return to the field laboratory the leaf litter is emptied into the tops of the funnels. The cabinet is prepared by filling the vials beneath the funnels with 80% ethanol. A sample label, identical to that used in the field (ensuring the correct sample number is recorded) is placed in the vial beneath the funnel. The leaf litter is spread out on the gauze platform in the funnel and the original sample label placed in the funnel with the litter. Extraction occurs over four to six days during which time the lights are on continually over each funnel in the array. It is important to check the funnels regularly as both bulbs can blow and ethanol will evaporate. In the latter case, the ethanol should be simply kept topped up. Animals moving away from the heat source (the light bulb) pass down the stem of the funnel and are collected in the vial

beneath. At the end of the extraction period the now-dry leaf litter is re-bagged along with its sample label for weighing. By weighing the leaf litter, counts from these samples can be standardised on a per-unit-weight basis.

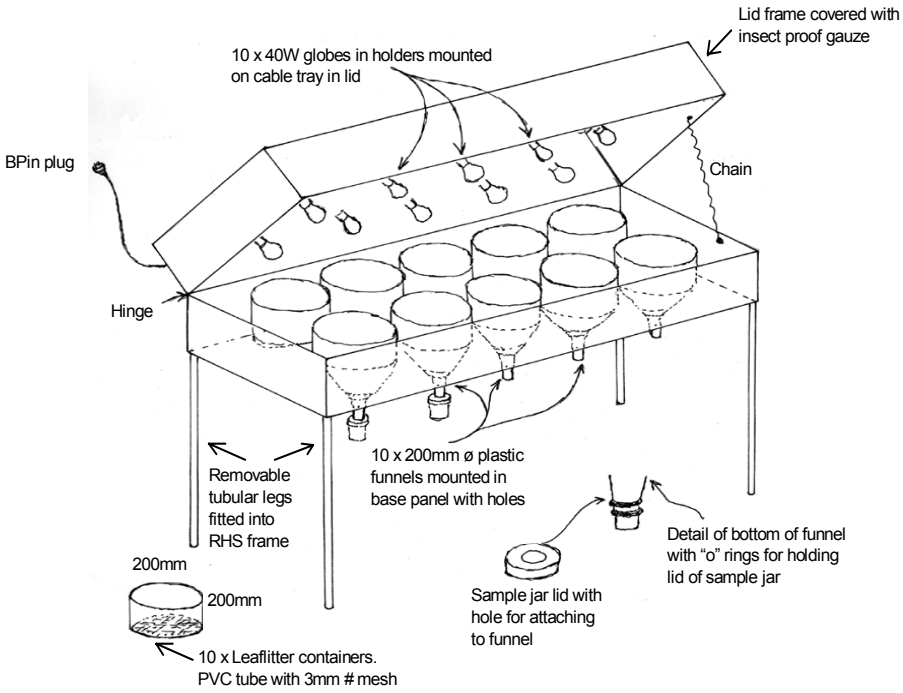


Figure 5: Schematic diagram of the portable leaf litter extractor.

Figure 6: Photograph of the portable leaf litter extractor.

Special Notes

Samples extracted from leaf litter in this fashion are very rich especially in Collembola and mites (see Figure 7 for sample result). They take significantly longer to sort than most other samples due to the high number of arthropods present and the large amount of fine sediment in each sample.

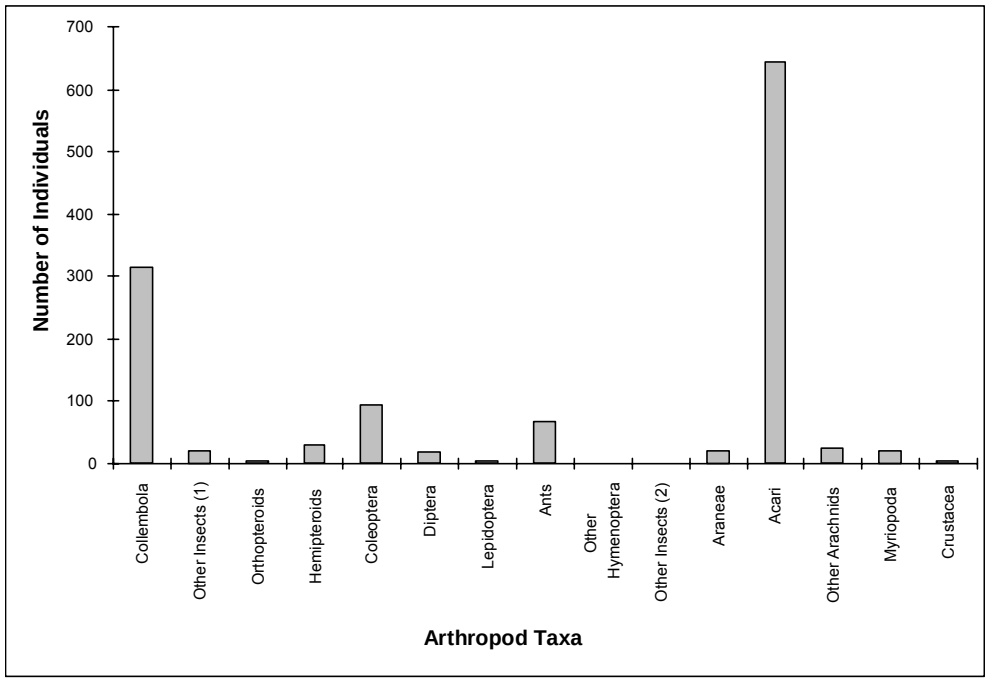


Figure 7: Total number of individuals collected from a set of ten litter samples from Baitabag Village, Madang, Papua New Guinea.

We generally place yellow pan traps at the same locations from which we collect litter samples. The same team of people can readily handle both tasks in one morning.

Particular points to note in litter sampling are:

- Ensure there is no space around the neck of the funnel and the collecting vial (this can be blocked with cotton wool if necessary) as the lights and the ethanol attract tiny insects directly, which contaminate the sample.
- Ensure that the tops of the funnels are also protected from tiny flying insects with gauze. It is preferable to set the funnels up inside, but if that is not feasible we have used a single fitted bed sheet over the top of the cabinet as a useful insect excluder.
- Guard against fire by checking all electrical connections frequently.

Equipment List

In the field (to collect ten samples):

- 10 x A4 zip lock bags
- 10 x sample labels
- 10 x randomly generated coordinates
- pencils
- 1 x one litre plastic container
- thick gardening gloves

In the field laboratory:

- 1 x complete Tüllgren extraction cabinet to hold 10 funnels
- extension lead/powerboard
- spare 40W light bulbs
- 10 x 70 ml sample vials
- 1 litre of 80% ethanol
- 10 x sample labels

PITFALL TRAPS

Pitfall traps are one of the oldest devices known to humans, whether used to trap ants or elephants. Their use in arthropod survey is widespread particularly when the target species are free-living ground dwelling groups. They have been used extensively for studies of spiders, springtails, myriapods, ants and beetles. Many studies have been reported in which the capture efficiency of pitfall traps are related to factors such as weather (Mitchell 1963), available food supply (Briggs 1961), details of the placement and construction materials of the traps (Greenslade 1973), and in response to various baits (Greenslade and Greenslade 1971). Luff (1975) provides an important overview of these factors.

In designing pitfalls for general survey, we took into account the material used, the ease with which they can be removed from the ground, serviced and replaced, the need to avoid swamping either by overland water flow or direct rainfall, and the necessity of using a killing agent within them to prevent larger captures eating or pulverising smaller ones.

Equipment Design and Preparation

We use two sizes of traps, one based around 25 mm diameter glass test-tubes (see Figure 8 and Plate 2), the other using 50 mm plastic tubes. The latter are 120 ml screw top vials (50 mm x 300 mm) used without their lids (Figure 8 and Plate 2). Each has smooth sides preventing animals from escaping once they are caught. Both tubes are slipped into close fitting sleeves made from tough plastic electrical conduit cut into lengths sufficient to house the tubes. The bottom of the conduit tube is chamfered to allow easy insertion into the hole in the ground. For the 120 ml vials, a ring of plastic is slipped over the top of the vial above the thread to fill a gap left between the top of the vial and sleeve. A plastic roof is fitted over each pitfall trap to protect the catches from the rain. This consists simply of a square of rigid plastic (approximately 150 mm x 150 mm square), with holes drilled in each corner. The hood is suspended above the trap by four large nails pushed through the holes. A specially made metal spike (one for each trap size) is used to make the hole in the ground for each trap, and an extractor tool (or garden trowel) for retrieving the outer tube.

We use both trap sizes as an on-going experiment, testing the effects of trap size on catch composition and body size. If only one size is to be used we advocate the more convenient 25 mm glass tubes.

There has been some debate over the use of killing agents and preservatives in pitfall traps. We know that ethanol will attract some insects, particularly drosophilid flies – from some distance around the trap. Ethanol is, however readily available. Without such a killing agent the larger insects in the catches will either eat the smaller or pulverise them to the point where they cannot be identified. Accordingly we regard the attractiveness to drosophilids as simply one of the characteristics of our traps – any trapping method will carry biases and one reason for using a set of complementary traps is that the biases will be different.

In the Field

Nine traps are arranged in a 5 x 5 cross with half a metre between each trap as a trapping ‘unit’ (Figure 10) and we combine the catches from each such set of nine each day. We place four sets of nine of each size of trap (a total of 72 individual traps) centred on randomly assigned points within the hectare. Nine holes are made by hammering the appropriately sized metal spike into the ground to the depth that the hard plastic sleeve will be flush with the soil surface. It is essential that the top of the trap is flush with the ground to operate effectively, the principal of the trap being that animals walking on the soil surface will fall into the trap. The tube is then inserted into the sleeve (for our large pitfall traps this includes the plastic ring to ensure the trap top is level) and filled one-third full with 80% ethanol. The rain

hoods are then placed over the traps. Once an array of nine traps is established, we place a small marker of flagging tape on a wire peg next to each tube and tape off the whole area of the trap array, to prevent accidental trampling by others who are working on different tasks within the hectare.

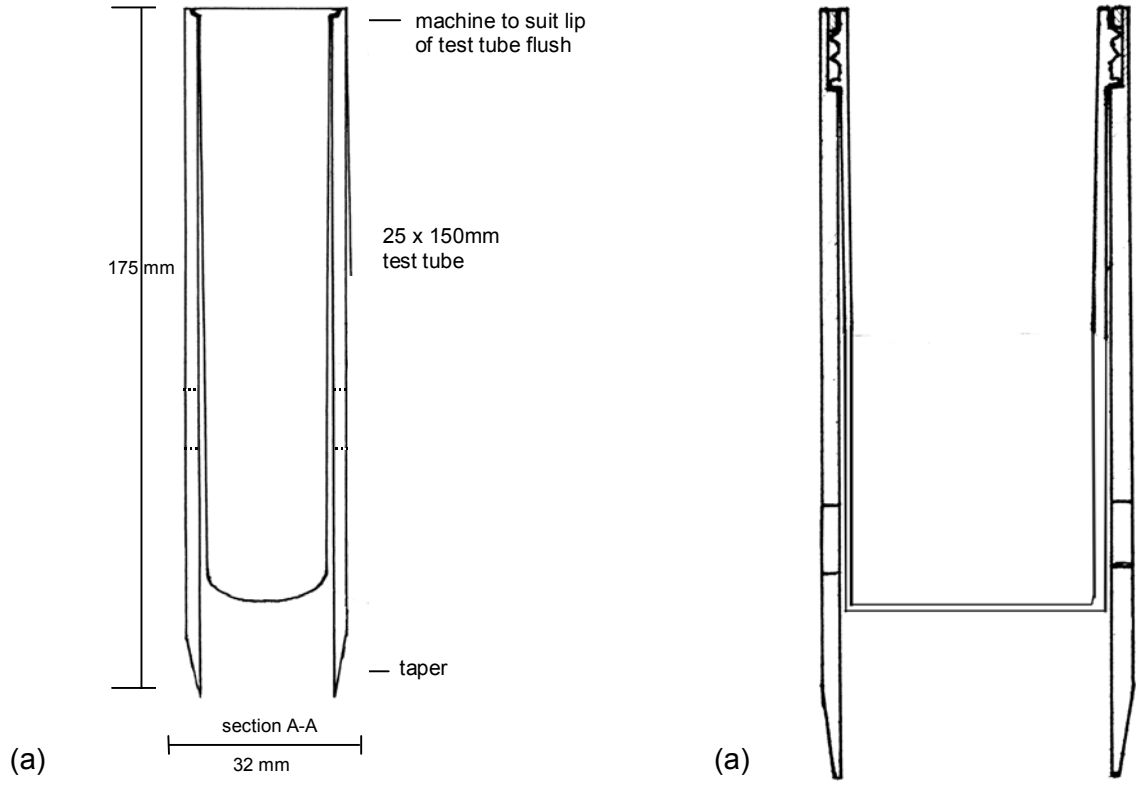


Figure 8: Schematic of the (a) small pitfall trap and (b) large pitfall trap (not to scale).

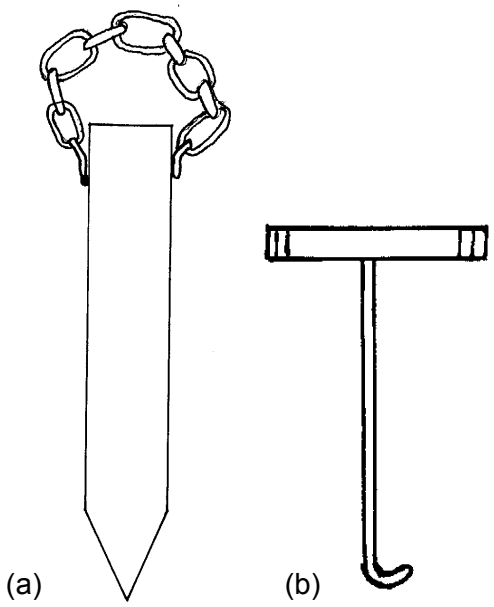


Figure 9: (a) Pitfall trap insert tool or 'spike' is hammered into the ground to create a perfect sized hole into which the trap can be inserted, and (b) the pitfall trap removal tool is basically a steel rod with a handle and assists in removal of the pitfall trap.

Traps are emptied daily for four mornings during a single survey event. Final collection and retrieval of the traps is completed on the fourth morning. When traps are emptied we use a small sieve (of the kitchen variety) lined with a folded piece of very fine nylon gauze (1028 microns in size or similar) to concentrate the catches from all nine tubes in an array together. To empty the traps, the inner tube can be slipped out of the hard plastic sleeve. The contents are then tipped into the sieve or funnel, the tube rinsed into the sieve or funnel using a wash bottle containing 80% ethanol. After emptying the tubes are again filled to one third with 80% ethanol and returned to their sleeve still in the ground. Once all nine tubes of an array have been emptied thus, the catch is washed from the gauze liner into a vial that contains a sample label for that array and day, again by using 80% ethanol in a wash bottle.

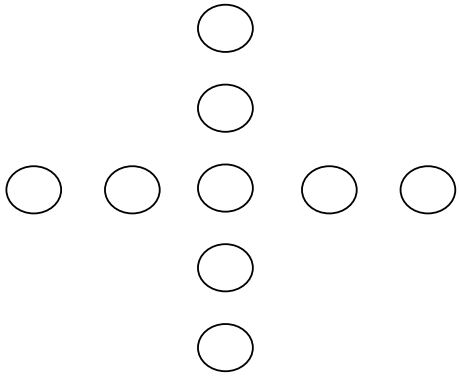


Figure 10: Layout of pitfall traps. Space each tube 500 mm apart.

Special Notes

It cannot be stressed too strongly that the effectiveness of a pitfall trap will reflect how well it is positioned initially. Any discrepancy between the lip of the trap and the ground surface such that the lip protrudes will reduce significantly the number of captures. We empty pitfalls on a daily basis and again it is necessary to check that when replaced in the ground the original perfect alignment of lip and ground is maintained.

We found that on one particular wet season trip in Borneo the overland flow and consequent micro-erosion of soil was such that pitfall traps simply were not effective. However well they were re-seated each time, the loose soil around their rims eroded and made them ineffective. This was, however, a period of exceptional wetness even for a humid tropical rainforest.

We add the following tips on pitfall trapping:

- Always carry spare tubes as both glass and plastic varieties occasionally break during insertion, handling and removal.
- Attempt to keep the samples as ‘clean’ as possible by preventing soil particles dropping into them during removal.
- A rubber bung with a screw inserted part way into its top can aid in removing recalcitrant glass tubes from their sleeves.
- Clean all the pitfall equipment carefully after use. It tends to attract the most mud and dirt of any of the equipment used and this is often more readily cleaned at the field laboratory shortly after use, than weeks later.

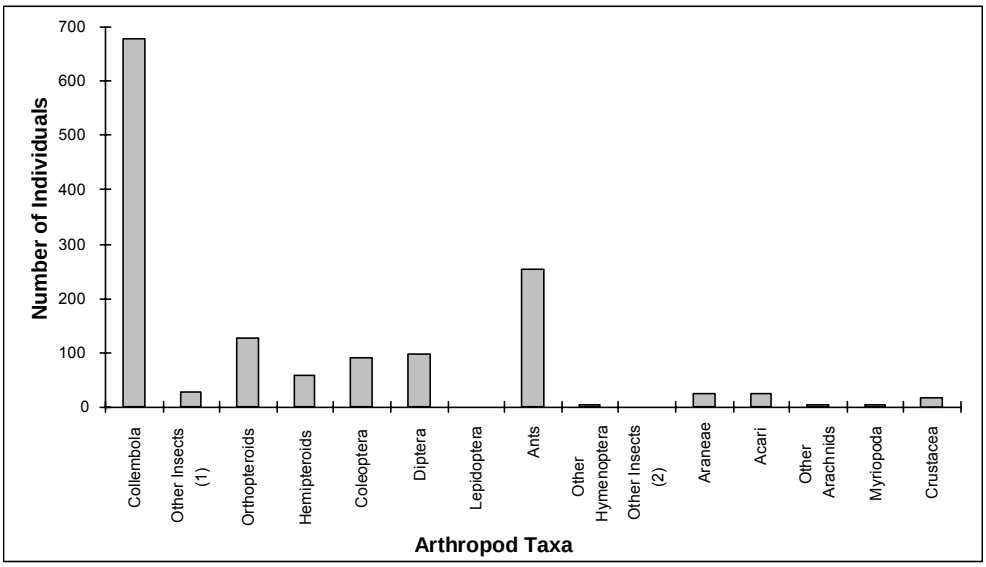


Figure 11: Total number of individuals derived from four days of pitfall trapping in tropical rainforest in Baitabag Village, Madang, Papua New Guinea.

Equipment List

Setting up small pitfall traps (4 arrays of 9):

- 36 x small pitfall traps (glass tube, plastic sleeve to fit glass tube)
- 36 x rain hoods
- 144 x nails (100 mm)
- small spike
- mallet
- 2 litres 80% ethanol
- flagging tape
- 4 x pegs
- permanent marking pens
- 4 x randomly generated coordinates

Setting up large pitfall traps (4 arrays of 9):

- 36 x large pitfall traps (120ml vial, plastic sleeve, plastic ring)
- 36 x rain hoods
- 144 x nails (100mm)
- large spike
- mallet
- 2 litres 80% ethanol
- flagging tape
- 4 x pegs
- permanent marking pens
- 4 x randomly generated coordinates

Collection (both sizes for 4 days):

- sieve or funnel
- fine gauze (1028 microns)
- 4 litres 80% ethanol
- 32 x large plastic vials
- 32 x samples labels
- wash bottle
- pencils
- large sealable bags

YELLOW PAN TRAPS

Yellow pan traps are one variety of water trap, widely used to collect small airborne arthropods that make up the aerial 'plankton'. They will also collect insects that jump from the forest floor (such as Collembola) and larger insects attracted to the highly reflective water surface either because it represents a light source, or because aquatic insects search out small water bodies in this fashion. They were first used by Moericke (1951) to study aphids. Harper and Story (1962) compared water traps of a variety of colours in their studies of sugar beet fly. Yellow painted traps were particularly successful followed, in order, by white, black, red, blue and green traps.

Equipment Design and Preparation

Our yellow pan traps are simply plastic take-away food containers (1675 mm x 120 mm x 40 mm), which we paint yellow using an aerosol can of yellow enamel paint (Plate 4). Each container is placed in a flat location and filled two-thirds full with water to which a few drops of detergent has been added. This causes any arthropods that alight on the water surface to sink. Harper and Story (1962) showed that, without the detergent, catch sizes were halved.

In the Field

Team members responsible for yellow pans are also responsible for collecting leaf litter at the same location. This is simply a matter of convenience and leaf litter could be collected elsewhere by another team. We put out ten yellow pans within the one-hectare plot (five in the quarter hectare plots) using randomly generated coordinates. We label each yellow pan with a number and a length of flagging tape is tied to adjacent trees to effectively rope off the area and as an aid to finding the container again (they are small and at ground level). We empty the yellow pan traps daily for a four-day period within a single survey, collecting the traps on the fourth day. As with the pitfall traps, we pass the catches through a gauze filter (1028 microns in size or similar) in a funnel on site to concentrate the catches. The traps are rinsed out with ethanol and the whole catch washed with ethanol into a vial containing a prepared label. The yellow pan is then refilled with water and detergent and replaced in position.

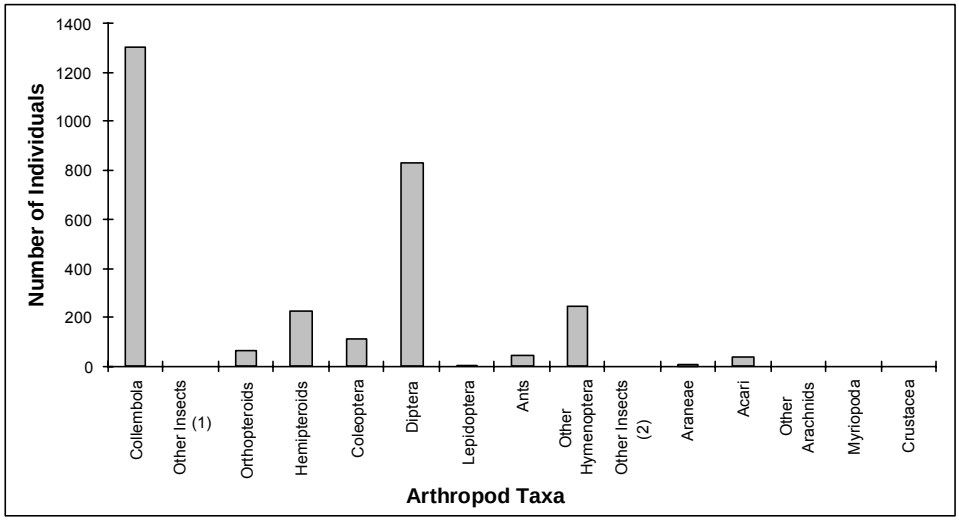


Figure 12: Example of the composition of a yellow pan catch from our surveys. Total number of individuals by order, collected from yellow pan trapping, in tropical rainforest at Baitabag Village, Madang, Papua New Guinea.

A team of two people can set out ten yellow pan traps *and* collect ten litter samples from the same sites in about three hours. It will take two people about two hours on site to empty them daily.

Special Notes

Points to remember in using yellow pan traps include the following:

- Take care when lifting a water-filled container from the ground: they are not particularly rigid and flex easily, spilling water and catch in the process.
- Be aware that large mammals and birds sometimes take an interest in these water-filled dishes and occasional daily catches may be lost in this fashion. Such losses should be recorded so that totals can be adjusted in terms of actual trapping effort later.
- Carry spare containers so that those that get damaged – by animals, branches or passing feet – can be replaced.

Equipment List

For setting up yellow pans:

- 10 x plastic take-away food containers (1675 x 120 x 40 mm) painted yellow
- 2 litres of water
- detergent
- 2 x rolls flagging tape
- permanent marker pens
- 10 x randomly generated coordinates

For collection (4 days, 10 traps):

- sieve or funnel
- fine gauze (1028 microns)
- 3 litres 80% ethanol
- 40 x 70 ml plastic vials
- 40 x samples labels
- wash bottle
- pencils
- 2 litres of water
- detergent
- large sealable bags (useful to carry out samples)

FLIGHT INTERCEPT TRAPS (FIT)

Flight intercept traps are those that intercept animals as they move through the air. Traps such as flight intercept or window traps target flying insects that fall hitting an obstacle during flight. Our ground-based flight intercept traps were developed by students Alex Creagh and Peter Grimbacher, based on the design of ground intercept traps used by Grove (2000).

Equipment Design and Preparation

The flight intercept traps consist of a clear polypropylene interception surface (150 µm thick, 400 mm wide, and 500 mm tall) tied between trees or stakes, set above a 5 litre polypropylene container (340 mm long, 160 mm wide, 120 mm high). The top edge of the screen is folded over a length of wire to give the screen stability and cord is tied to loops formed in the end of the wire. The bottom corners are strengthened with washers taped to the screen and lengths of cord are tied through these. A rain hood (clear polypropylene 150 µm thick) is also tied to surrounding vegetation to prevent rainwater filling and overflowing the catch container. Again washers are used to strengthen tie points (see Figure 13). The catch container is a readily available food container (available from catering suppliers or plastic manufacturers).

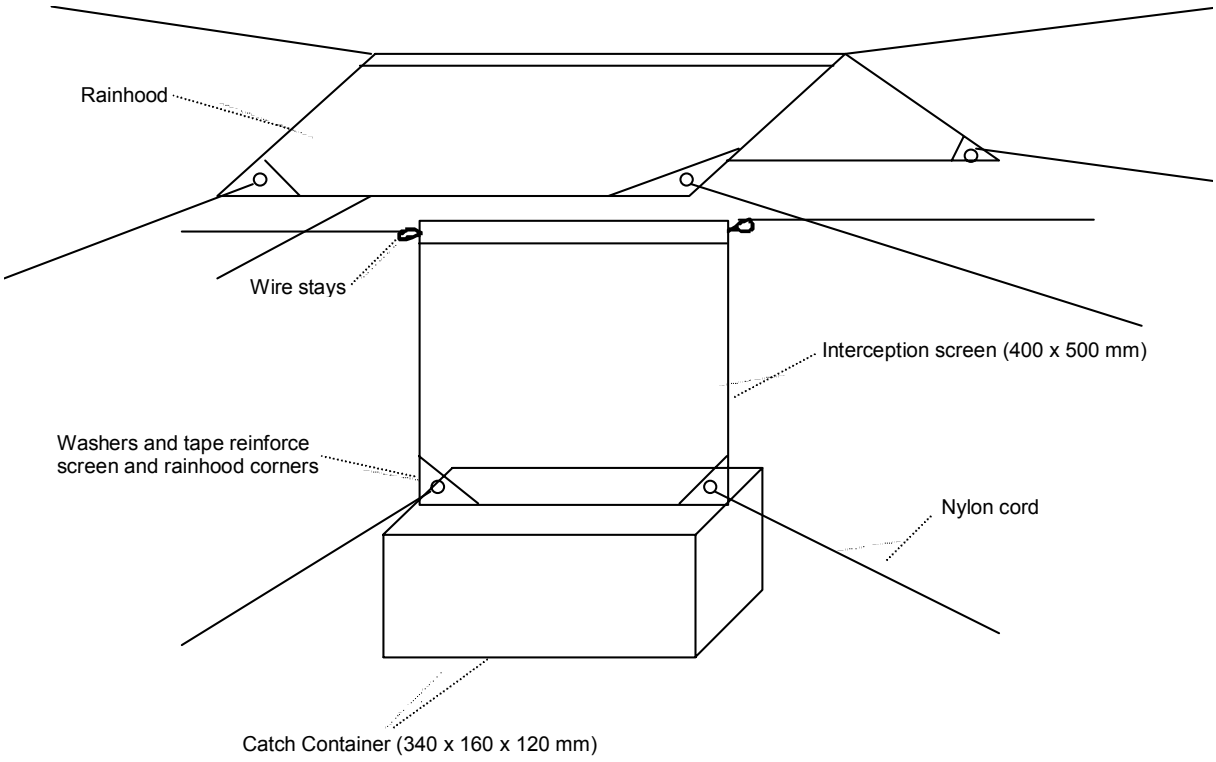


Figure 13: Design of the ground flight intercept traps used in one-hectare surveys.

In the Field

Successful trapping using the interception method requires following a straightforward set-up procedure to ensure the correct working. First, tie the interception screen into position a little above the catch container. Then the bottom edge of the interception surface is made flush with the polypropylene container by pegging down the corners tightly. The container is also

pegged down by pushing wire pegs into the ground with the hook of the peg over the lip of the container. The number of pegs will depend on the exposure of the site. The catch container is then filled to a depth of about 40 mm with a solution of water and detergent. Where traps are to be left for a period of days (up to two weeks), two litres of propylene glycol (33% solution) can be used in the container to act as a killing and preserving solution. The rain hood is erected above the trap by again stringing this between trees and ensuring the bottom edge does not come below the top of the catching screen and reduce the catch area.

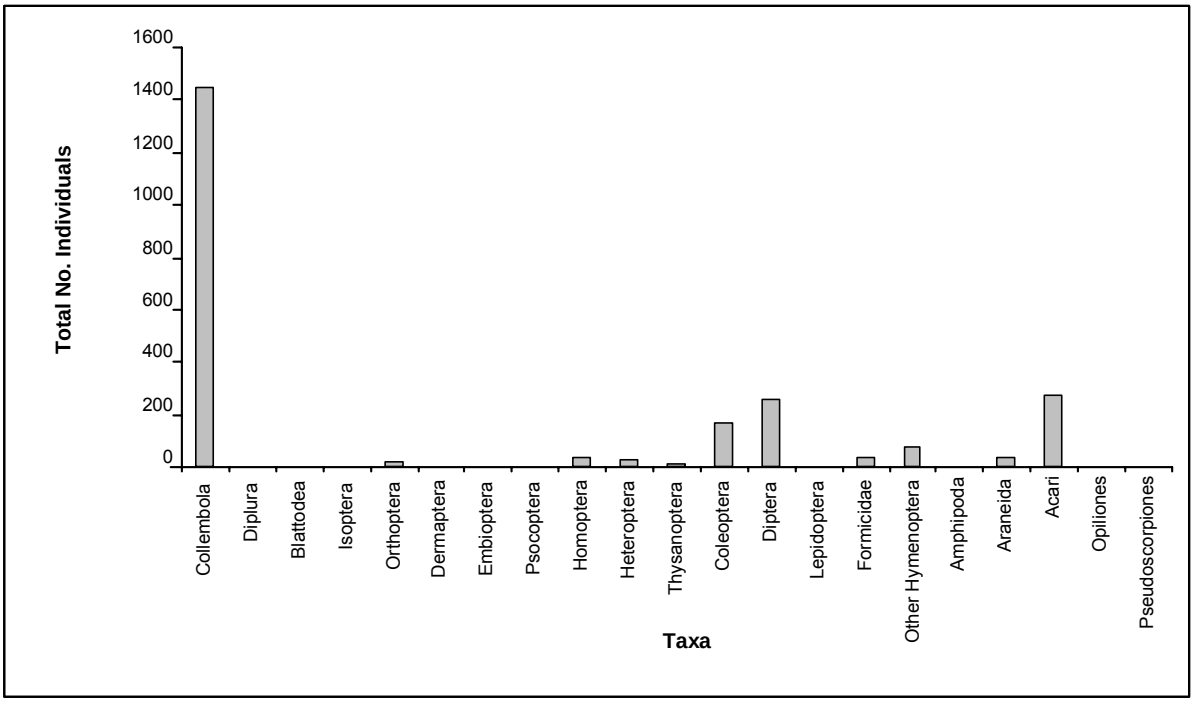


Figure 14: An example of total capture for flight intercept traps, here used in Cat Tien National Park, Vietnam, July 2002.

Equipment List

Set up:

- 10 x catch containers
- 10 x intercept screens
- 10 x rain hoods
- approximately 60 tent pegs
- hammer
- 10 litres of water and detergent solution
- 2 x rolls flagging tape
- permanent marking pens
- spare cord

Collection:

- funnel
- fine gauze filter
- 10 x 70 ml plastic vials
- water and detergent container
- ethanol wash bottle
- 80% ethanol
- 10 x pre-prepared labels
- pencils

MALAISE TRAPS

The fabric tent trap which has become known as the Malaise trap was first described by Malaise (1937) and subsequently modified by a number of authors including Gressitt and Gressitt (1962) and Townes (1962). It targets free flying insects and is particularly successful in catching Diptera of which many thousands may accrue over four days of trapping. Juillet (1963) has suggested that Malaise traps are unbiased for Diptera but less so for Coleoptera and Hemiptera. Roberts (1970) disagreed with that assessment, pointing out that even catches of Diptera were influenced by both the shape and colour of the trap. To this we would add that the positioning of the trap is a crucial factor.

Equipment Design and Preparation

We use commercially manufactured Malaise traps, which have a collector at one end of the trap only (see Figure 15, Figure 18 and Plates 6 & 7). We have added to the basic design a lightweight rectilinear frame within which the trap can be erected before it is hauled into the canopy (Figure 17). Using gloves and long forceps, we place a small block of Dichlorvos™ impregnated plastic within the collecting jar, but otherwise samples accumulate 'dry'. We deploy six traps within the hectare, three at ground level and three hauled into the canopy on ropes.

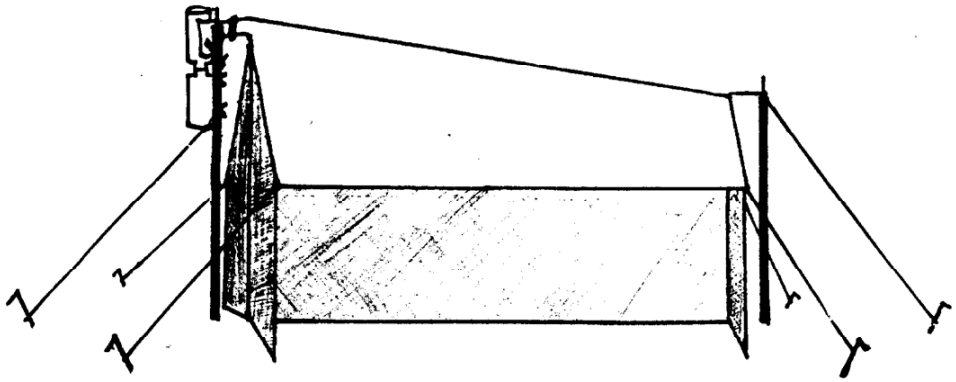


Figure 15: The fully erected 'ground' Malaise trap.

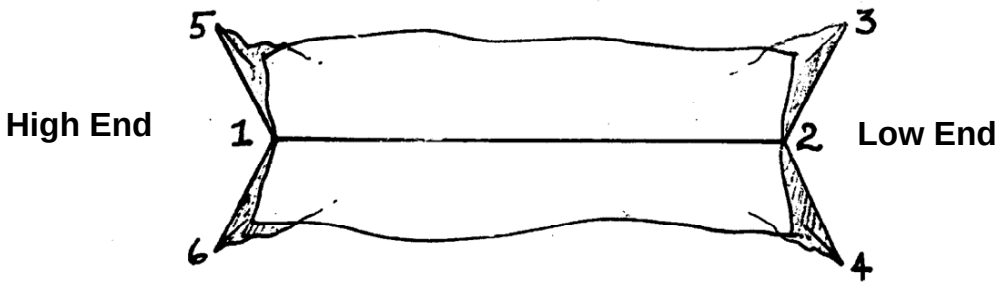


Figure 16: The Malaise trap 'tent' spread out to peg.

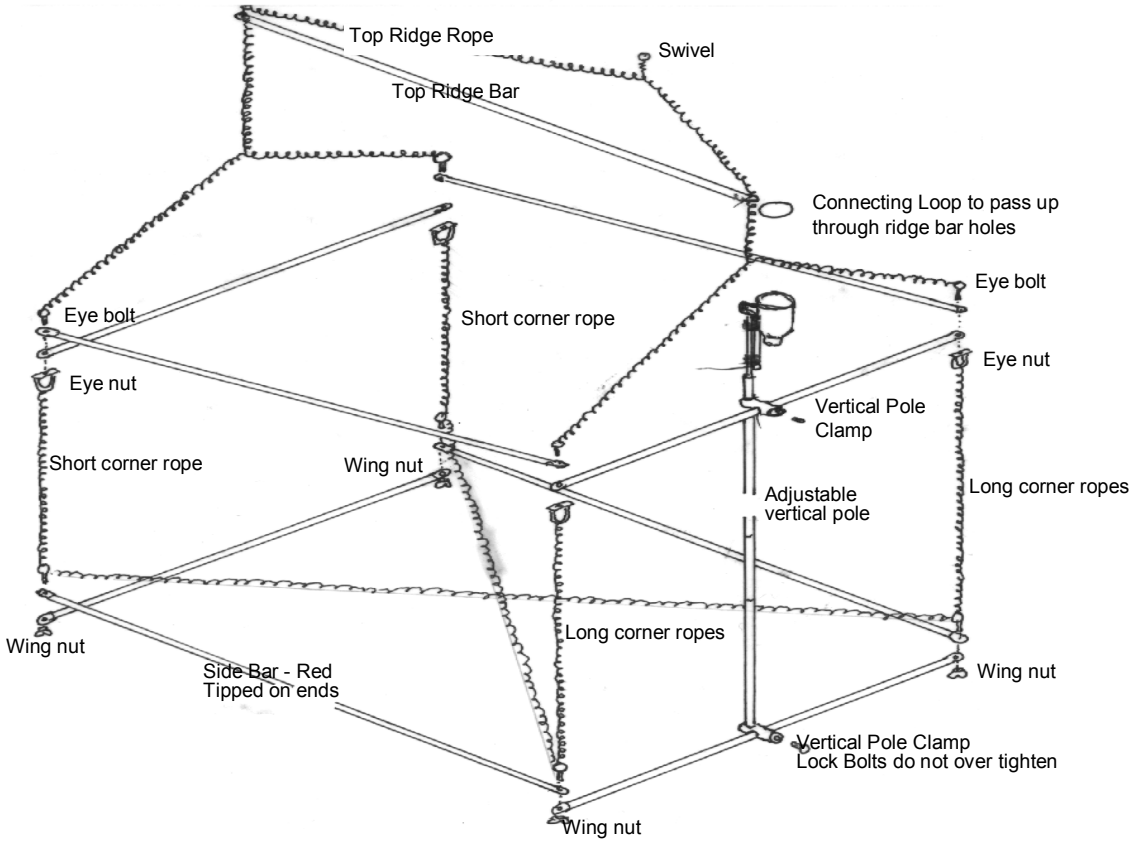


Figure 17: Malaise trap forest canopy frame.

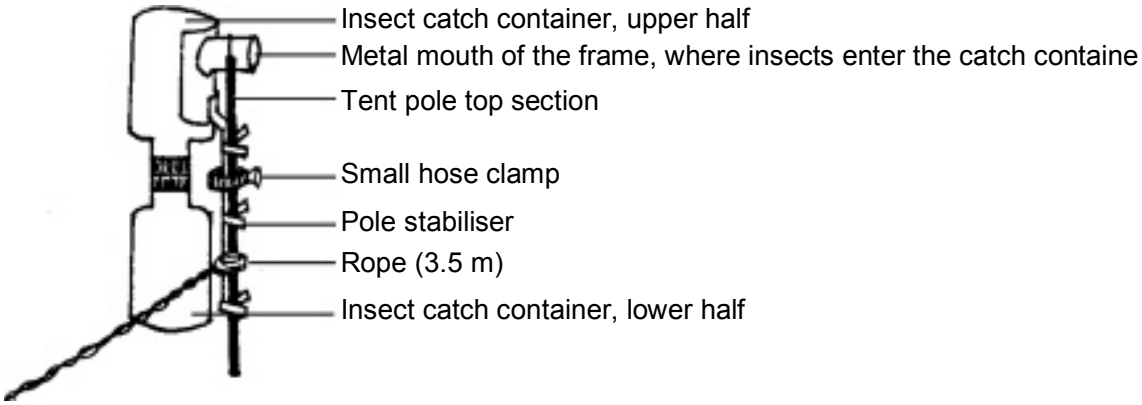


Figure 18: Malaise trap catch container.

In the Field

The erection of six traps, three of which have to be fitted within canopy frames, is a time-consuming process and will take a team of about four people a whole working day. Position the trap with great care. Look for natural flyways through the ground-zone vegetation, or natural openings within the canopy.

To suspend traps in the canopy, a single line with a pulley is hauled over a high, solid branch after a line has been shot into the canopy (see general description of canopy sampling, page 17). Canopy traps are manoeuvred through the lower foliage of the forest using two guide

ropes attached to opposite ends of the frame within which the trap is erected. We empty each Malaise trap daily for four days and transfer the daily catch of each trap into 80% ethanol as soon as is practicable after its collection. Figure 19 gives a sample outcome from ground zone trapping. Emptying the ground zone traps each day takes a matter of minutes but the lowering and raising of the canopy mounted traps is more time consuming and takes a minimum of three people given that the trap has to be 'steered' into place in the tree tops. Traps deployed in the canopy catch fewer insects than those erected on the ground, which also contain a component of animals that simply crawl up the fabric.

Malaise traps, as we use them, retain their sample dry. After each day of exposure the lower part of the collecting unit is unscrewed, the rectangle of killing agent removed, and the catch washed out into a storage vial using 80% ethanol. Of course label information must be transferred carefully at this point. If required selected groups such as larger Lepidoptera and Coleoptera can be removed for separate curation from the collecting jar before ethanol-washing. Once empty, the collecting container must be wiped out, the killing agent restored and the container screwed back on to the upper part of the collection assembly. It always pays to ensure that the connection between the collecting assembly and the trap itself remains undisturbed with free ingress for flying and crawling insects each time the trap is manipulated.

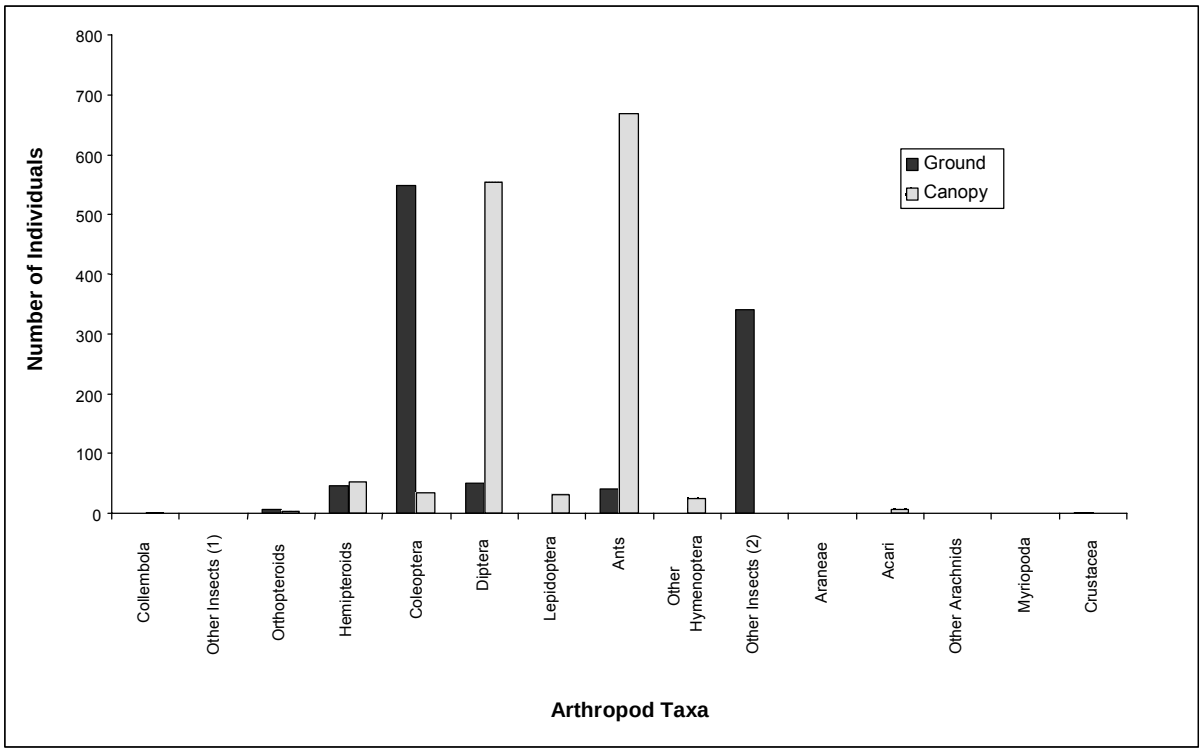


Figure 19: Example of the total catch from ground and canopy zones Malaise traps in tropical rainforest at Baitabag Village, Madang, Papua New Guinea.

Special Notes

Particular tips for operating Malaise traps are listed below.

- Keep all the equipment including tent pegs and guide ropes necessary for a single trap plus a separately packaged amount of killing strip (purchased as Pestox™ or Shelltox™ household insecticide) in a single carry bag. Tools required – a hammer, screwdriver, pair of scissors and sewing kit – should be in a separate bag where they cannot tear the fabric of the traps.
- Always handle the insecticide strips using plastic gloves.
- Look for particularly high branches from which to suspend Malaise traps – the sheer size of the traps means that to sample the canopy fauna proper this must be the case.
- Spend time and care ensuring that the traps are put up securely. Time spent during trap erection is time well spent given that the whole four day trapping program depends upon it.
- Malaise traps are fragile objects and repair kits, supplied when the Malaise trap is purchased, and spare parts should always be carried. Check each trap each day for damage.
- Labelling procedures are particularly important in Malaise traps as an extra-step is involved in catch-handling – from the catch bottle of the trap itself to an ethanol-filled vial. Ensure that the same label or an exact duplicate is transferred with the catch.
- Number each trap in an unequivocal fashion and ensure that any numbers written on the trap bottles, for example, during previous uses do not confuse collectors.
- Wash and dry each trap immediately after use and repair any tears that may have arisen.

Equipment List

- 6 x tent type Malaise traps
- 3 x canopy frames as per Figure 17
- 3 x sets of ropes and pulleys for hauling into the canopy (each set includes single large rope with pulley, single hauling rope and two lighter ropes as guide ropes)
- 18 x pegs
- mallet
- 8 x Dichlorvos impregnated pest strips
- long-handled forceps
- gloves
- tissue paper (helps prevent insect damage).
- sample labels
- 24 x large vials (250 ml)
- pencils
- permanent marker pens
- sewing kit

LIGHT TRAPS

Light traps have been widely used for insect surveys for many years growing out of the simple observation that candles, carbide or storm lanterns used in earlier times attracted insects freely. The modern use of light traps began with Robinson and Robinson (1950) and Frost (1952). Many designs are available for different uses and many have been developed along the way. Light traps have been widely used for insect surveys both for particular target species (Bogush 1958, Geier 1960) and for whole faunas (Taylor and Taylor 1977). They target a wide range of insects but are particularly successful for Lepidoptera and Coleoptera. Light trap catches are affected by a wide variety of environmental factors and there is a large literature dealing with these variables (Hollingsworth *et al.* 1961). Perhaps the most important of these is the phase of the moon (Bowden 1973, Bowden and Church 1973). Light traps operated close to the period of full moon, in general, attract fewer moths than at other times, although these may be of different species.

Equipment Design and Preparation

We use a commercially available design based on the so-called Pennsylvania (or Texas) trap (Frost 1957). Essentially this comprises a vertically mounted 'black' light fluorescent tube with three transparent plastic vanes mounted equidistantly around it. These vanes are shaped to fit within a funnel and the funnel sits within a replaceable bucket in which the catch accumulates. The light operates using a 12 Volt gel battery of the type used for powering motorcycles. To this commercially available model we add a rain protector of a size larger than the bucket (we use an alloy dustbin lid around 600 mm diameter) and a sandwich of wooden boards beneath in which the battery can be mounted.

These modifications are specifically so that:

(a) the trap can be used in wet to very wet conditions; and
(b) the trap and its battery can be hauled into the canopy by rope.

Figure 20 and Plate 8 illustrate the modified design we use. We use a block of Dichlorvos™ impregnated plastic as a killing agent placing it in the bucket together with torn baking paper to provide resting places for captured insects.

In the Field

We operate six traps simultaneously within our hectare, all at randomly determined points, three at ground level and three in the canopy. We select sites such that no trap is visible from any other trap. We run the traps for five nights within each field survey, avoiding the week around the full moon. Erecting the ropes and traps is a full days work for a team of two people. As with the Malaise traps, a rope and pulley system is set up over a sturdy branch for the three canopy samples. The assembled traps are hauled into the canopy once the light has been connected to the battery. The batteries will run for approximately 12 hours, so the traps must be set to run at about 5.00 pm and left to run through the night. They are emptied daily for the five-day period and the batteries must be recharged every day at the field laboratory. To allow ample recharge time, we carry two sets of batteries, so that one set will always be charging. The evening work we usually shorten by sending out two teams of two. These teams will reset the already empty traps by inserting a newly charged battery and the empty catch bucket with a new strip of Dichlorvos™. Canopy traps are hauled into the canopy. Check that each light is working and glowing steadily before leaving it in the field. When a light does not work we usually leave that trap overnight and service it the next day – trying to replace fluorescent tubes late in the evening, in the field and, often, in the rain, is not a good idea. Traps that have been left off for a night for whatever reason are kept open for

an additional night or nights after the original trapping period is completed. Although this unbalances the sampling design there is little alternative and the minor statistical disadvantages are outweighed by the fuller biodiversity survey so obtained.

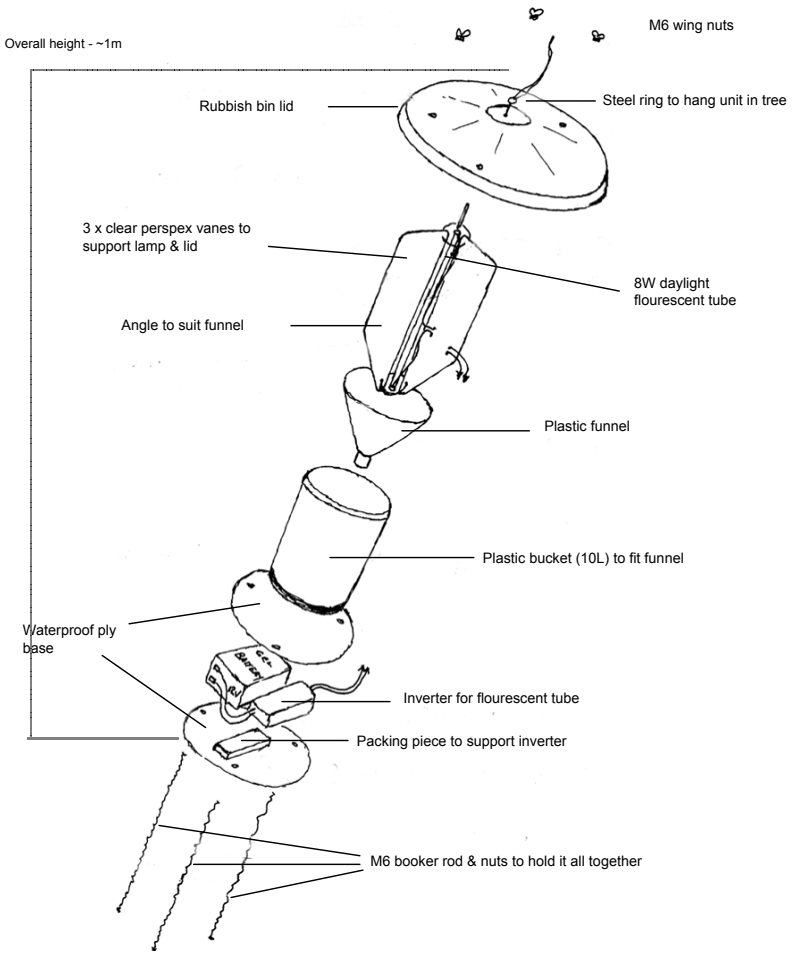


Figure 20: Pennsylvania style light trap modified for rainforest use.

Emptying the catch each morning represents about an hours work for a team of two. These team members will remove both the used battery and the bucket in which the catch lies. The bucket funnel can be simply 'plugged' with tissue to prevent the escape of any reviving samples. Returning the full buckets to the field laboratory needs to be a high priority **first thing** each morning so that material can be handled when fresh.

Special Notes

We use light traps solely for sampling the Lepidoptera and Coleoptera, discarding other portions of the catch. Indeed we restrict our Lepidoptera surveys to the macro-Lepidoptera (in the phylogenetically defined sense of Minet (1991), that is to include any or all of the Mimalloidea, Lasiocampoidea, Bombycoidea, Axioidea, Calliduloidea, Hesperioidea, Hedyloidea, Papilionoidea, Drepanoidea, Geometroidea, Noctuoidea) plus the Pyraloidea and all Coleoptera.

The Lepidoptera from these light traps are the only insects that we curate in the field and we sort many of them to putative morpho-species at the same time. This enables us to discard many very common moths once they have been counted. We use standard setting boards and spread the moths each day. These are stored in a portable drying cabinet for the three

subsequent days and are then removed from the boards, labelled and sorted. They are then stored in specially constructed wooden trays within which standard museum 'unit trays' fit tightly. This preserves the material in good condition and minimises direct handling subsequently.

We generally pick out all moths with greater than 1 cm forewing length in this fashion, discarding later those which do not fit within our defined taxonomic ranges. Where possible we pick out the very few smaller moths that fall within our target groups (e.g. some Noctuidae and Geometridae). These processes of curation are time-consuming and require appropriate expertise. Catches could simply be layered among tissue paper within an airtight box together with some anti-fungal agent such as chlorocresol crystals. However, if we have a full team of people in the field not only is this unnecessary, but the quality of the material we return with from the field is much enhanced. In general, the more work one is able to do in the field or field lab, the better. The Coleoptera are simply picked from the catches and preserved in 80% ethanol for later attention. In general, light traps catch larger (as well as smaller) beetles and are undoubtedly targeting a fraction of the fauna which none of our other methods sample.

The sorting, mounting and subsequent curation of the specimens occupies three to four people full time during the two-week period. At least two of these helpers need to have been trained in the art of setting moths.

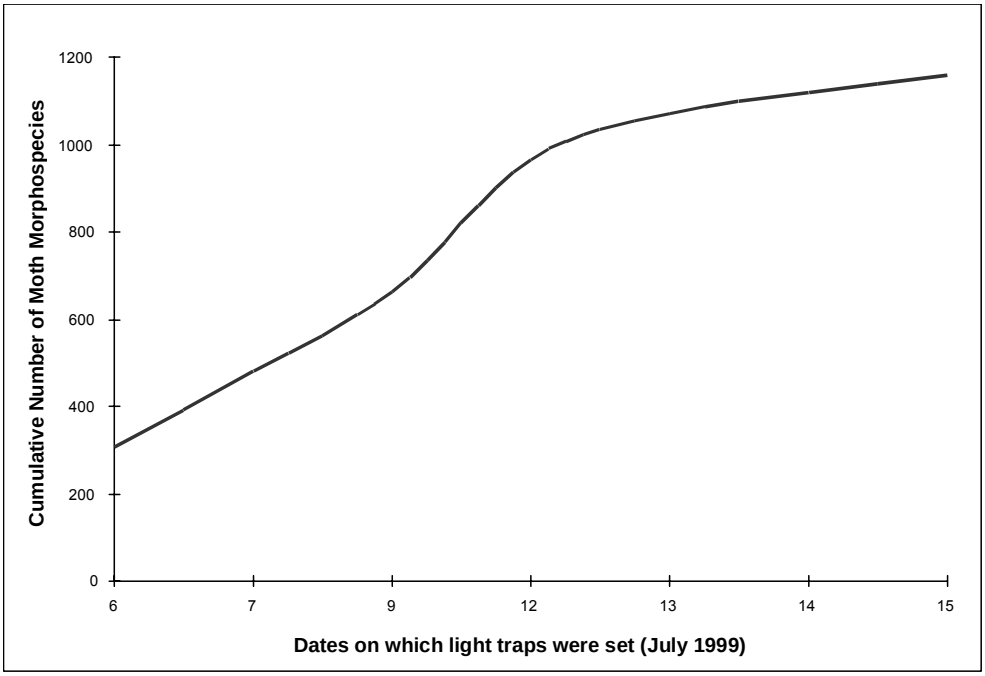


Figure 21: Example of the cumulative catch of moths in light traps at Baitabag Village, Madang, Papua New Guinea.

Points of particular practical note when light trapping include the following:

- Protect mounted material from moisture, cockroaches, ants and clumsy people carefully and continuously.
- We maintain two full sets of batteries so that one set is always fully charged. Check that batteries have 'held' their charge using a voltmeter before deploying them in the field.

- Remember that the traps need to be opened late in the day every day for five days – decide early in the day who is going to do this – it is not a popular job at the end of a hard days work!
- Maintain effective killing bottles (four or five, best based on cyanide or ethyl acetate) in the laboratory for dealing with moribund material in the trap catches once they have been returned to the laboratory. Remember such killing bottles represent highly hazardous material and should be marked appropriately.
- Take tissues to the field when emptying the light traps in order to stop the entry at the base of the funnel before removing it and, occasionally, to dry the inside of the bucket before carrying it from the site.
- Moths and beetles are frequently perched on but not in the traps when they are visited each morning – we simply sweep these into the funnel and add them to the catch.
- Number each trap clearly and check, preferably twice, that this number is retained with the catch throughout.
- In the field laboratory traps must be tipped out into sorting trays. Sometimes catches are subdivided for ease of handling particularly during the curation process. It is essential to ensure that at every subdivision of the catch a label is created so that at any time in the prolonged process of curation the trap number and day of capture of every specimen can be clearly identified.

Equipment List

- 6 x Pennsylvania Light Traps (modified as per Figure 20)
- 12 x 12 volt batteries
- 3 x canopy ropes and pulleys
- 3 x haul ropes
- 6 x guide ropes
- Dichlorvos impregnated pest strips
- long handled forceps
- baking paper/tissue paper
- sample labels
- spare wing nuts
- nylon rope to suspend ground traps at head height

In the field lab:

- entomological pins
- moth setting boards
- polyporous pith
- mylar setting tape
- drying cupboard
- taxon labels

CANOPY KNOCKDOWN

Sampling the free-living arthropod fauna of the forest canopy using a cloud of short-lived, quick-acting pyrethrum (or pyrethroid) insecticide has become the method of choice for general canopy collecting. In a little known paper, Erwin (1990) described the history of the technique, collecting together many key references. The technique was first applied in temperate forests by Martin (1966) but opened up a new era of study of arthropods in tropical forest canopies after Roberts (1973) used a canopy fogging technique to sample canopy Orthoptera in Costa Rica (although he used the more powerful insecticide Dichlorvos™ for this purpose). Subsequently, Gagné (1979) and Erwin (1982) modified the technique so it could be used for the quantitative sampling of canopy faunas. Other key references include Southwood *et al.* (1982a, 1982b) and Stork (1987a, 1987b, 1988).

We were the first to apply this technique on a large scale in Australia and our basic methods are in Kitching *et al.* (1993). We used a modified backpack sprayer producing an insecticidal cloud of slightly higher droplet size than the Dynafog™ machine of earlier authors (although we have used such machines in other studies).

Equipment Design and Preparation

We have used a Solo™ backpack mister for our surveys. We simply modify the backpack to create an attachment point for the canopy rope ensuring the machine will be held upright and the nozzle at a 45º angle. We have previously used pyrethroid-based insecticides, but have switched to a pyrethrum insecticide, Pyfog™ (Rudchem Pty Ltd, Melbourne). This insecticide comprises a mixture of natural pyrethrums. The mixture is made up at the concentration recommended by the manufacturer for general use. Collection of the arthropods falling from the canopy is made using collection hoops. These collecting hoops are a funnel of white plasticised fabric at the bottom of which is sewn an elasticised sleeve into which an ethanol filled collecting vial is fitted (see Figure 22). Into the top rim of the hoops is sewn a circle of 5 mm wire to which three lengths of fishing wire are attached. These are joined where they meet above the centre of the hoop to allow a point from which to hang the hoops. Equip each funnel with a simple clip for attaching it to the suspending ropes. We use No. 2 size ‘snap swivels’ available from fishing shops, but any small clip that can be closed and then undone easily will suffice.

In the Field

We carry out three spraying events within each plot, each one of which targets a 10 m x 10 m segment of high canopy. Again we locate the centres of these three 10 m x 10 m subplots randomly but frequently must modify these to prevent overlap, and because a stout horizontal branch is required in the canopy to bear the considerable weight of the fogging machine. At each site a rope with a pulley attached at one end is pulled over a high branch (see general description of canopy sampling, page 17). The hauling rope is thread through the pulley. Two further light weight ropes are attached to the base of the sprayer to guide the machine past any obstacles when hauling into the canopy.

A cat’s cradle of lighter ropes is constructed at head height around this central suspended rope. We hang twenty 0.5m^2 collecting hoops within the 10 m x 10 m plot from this cat’s cradle (Plate 1). Setting up of the canopy ropes, cat’s cradle and hoops should be done the day before spraying is planned to enable an early spray. Vials should not be placed in the hoops until the morning of fogging as these hoops act as very effective leaf and rain catchers. A vial half filled with 80% ethanol is placed in the base of each collecting hoop on the morning of spraying.

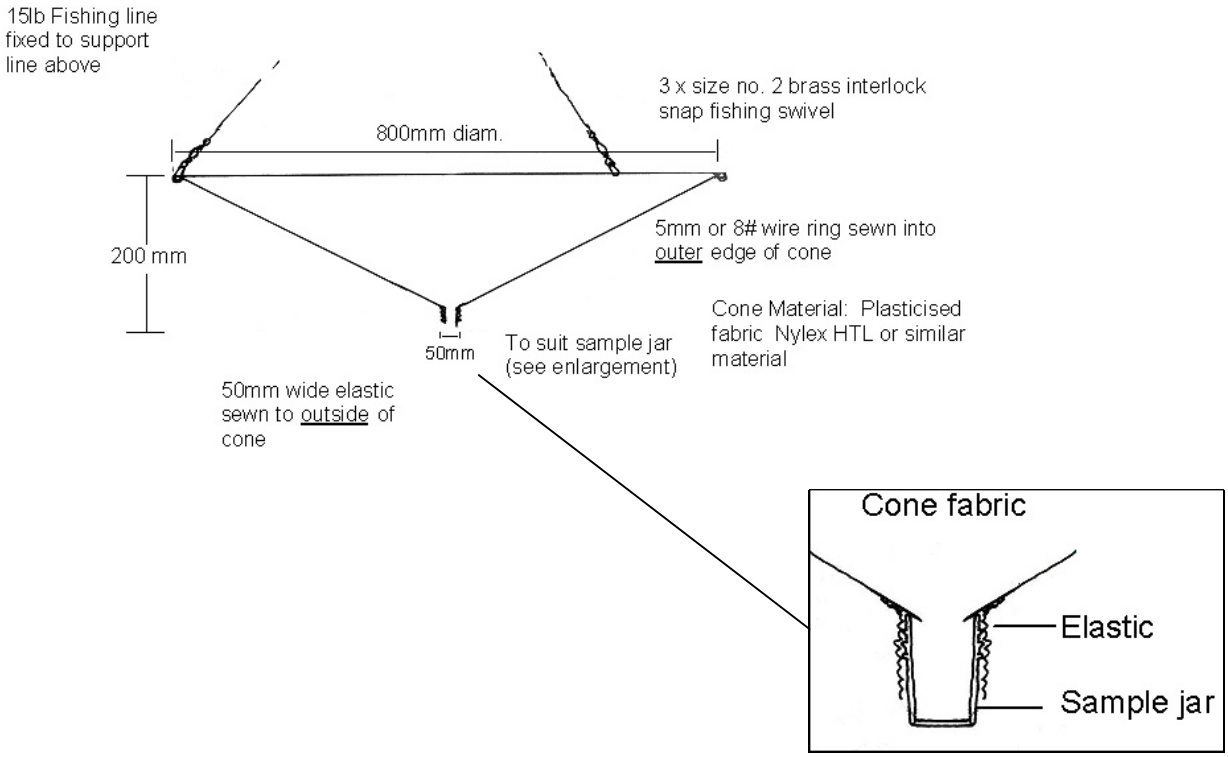


Figure 22: A fogging 'hoop' or collecting funnel.

Establishing a spray site once the rope is in place is an hour's work for two or three people. The actual spraying requires three people, two on the haul rope and one on the guide rope. These three workers must wear appropriate all over protection including coveralls, earmuffs, gloves, respirators and goggles. Ensure before beginning a spray that all twenty hoops have vials placed in them and all workers have appropriate all over protection.

We start the machine on the ground and open the spray nozzle so that a fine mist of insecticide is ejected to about six metres. We then haul the sprayer into the canopy guided by ropes attached to its base. We deliver insecticide for five minutes at each site, during which time the machine, inevitably, spins on the rope and creates a cloud throughout the canopy above the collecting funnels. After five minutes we lower and shut off the sprayer. Once the spray is complete we leave the area for thirty minutes returning subsequently to go around the funnels brushing any arthropods that fall into them down into the collecting vial. We collect for three to four hours after spraying. Tending the funnels and, finally, emptying them after three to four hours is patient and painstaking work for one person. Ensure that all twenty vials have a sample label that is numbered with both the number of the site and of the vial itself. The samples are then returned to the field laboratory for sorting.

All spraying is done in the mornings (pre-noon), as early as is possible utilising windless periods. Two sites can be sampled in one morning although we normally aim to sample one site a day in this fashion. In general, we carry out our spray samples late in the survey to minimise any interference between the insecticide and any other trapping method.

Each spray event produces twenty catches for sorting but these are not themselves replicates. Although the catches are kept separate, the counts from all twenty are combined as the sampling outcome of the spraying event. The replicates we use for analysis are the three separate spraying events within the hectare.

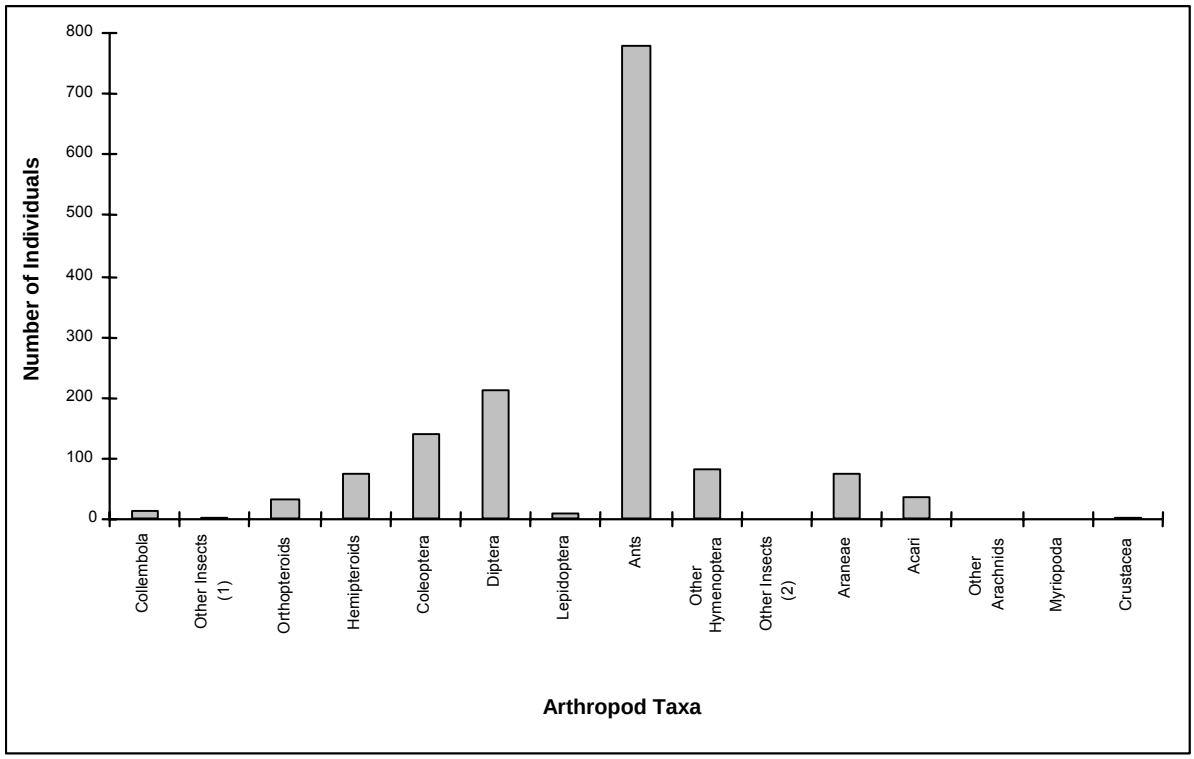


Figure 23: Total number of individuals captured from one day's canopy knockdown sampling in tropical rainforest at Baitabag, Madang, Papua New Guinea.

Special Notes

Particular hints for efficient canopy sampling are as follows:

- Begin the canopy sampling process sufficiently early during each field trip so that several unusable mornings (with the winds that make canopy misting impossible) can be dealt with if need be. We allocate the fogging to a day when no other collection is being made in the morning.
- Carry tools and spare parts for the mister – these machines are based on a two-stroke engine and need constant cleaning, maintenance and 'tweaking' if they are to work efficiently. Having the machine serviced prior to the trip can help minimise such problems.
- Prepare dilutions of insecticide from concentrate before taking the machine into the field – one tank-full of chemical in the mister is sufficient for all three sprays, provided the nozzle aperture is maintained at 'fine'.
- Store both insecticide and fuel carefully at the field laboratory.
- No other workers are to be on site during spraying.
- Ensure that the branch to which the rope pulley is attached is stout and healthy – sufficient to bear the weight of the heaviest of the research team. Many kilos of operating backpack mister filled with fuel and insecticide makes a very awkward missile should the branch give way.
- Ensure that any insecticidal spillage on clothes or skin is rinsed off using copious quantities of water immediately.
- Ensure that aspirators and other safety equipment are serviced regularly and that disposable filters are replaced.

- Wash down equipment with soapy water following use. This includes the chemical tank and outside of the fogger, goggles, earmuffs and coveralls. Never store the sprayer with residual chemicals and ensure leftovers are correctly labelled and waste disposed of appropriately.

Equipment List

For a single fogging event:

- 1 x backpack sprayer (Solo™ Mistblower Model Port 423 or similar)
- bow and arrow
- 1 x haul rope and pulley (heavy duty)
- 2 x guide ropes
- pyrethrum insecticide
- fuel container
- fuel (usually two stroke)
- water container
- thin rope for 'cat's cradle' support for hoops
- 20 x wooden stakes
- 20 x hoops
- 20 x sample jars
- 20 x sample labels
- randomly generated coordinates
- wash bottle
- 80% ethanol
- 3 x paint brushes for dry-brushing hoops
- 3 x coveralls
- 3 x goggles
- 3 x respirators
- 6 x respirator filters (spare)
- 3 x gloves
- 3 x ear muffs

BARK SPRAYING

The arthropod fauna of the bark surface of trees is rich and interesting. It is particularly rich in arachnids and beetles and its sampling adds a novel segment of the forest fauna to any survey. Bark surfaces are easy to sample.

Equipment Design and Preparation

We have developed a simple technique that involves suspending a modified collecting hoop (Figure 24 below) against a segment of tree bark at about head height. The hoop has had its wire frame removed from about a third of its circumference (in fact we use damaged canopy spray hoops for this purpose). The hoop is pinned tightly to the bark using large thumbtacks. We then mark the corners of a vertically oriented segment of bark, 1 m x 0.5 m above the edge of the collecting hoop. A vial containing 80% ethanol is placed into the elasticized cuff of the hoop.

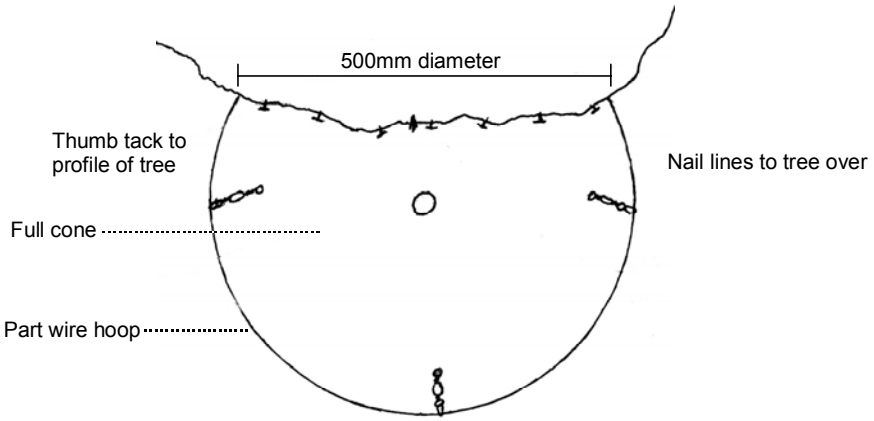


Figure 24: Bark spray hoop.

In the Field

The outlined segment of bark is sprayed using an aerosol can of proprietary household insecticide, again based on simple pyrethrums with piperonyl butoxide. We spray each half square metre for about twenty seconds from a distance of about one metre (Plate 4). Holding the can any closer results in the condensation of liquid insecticide on the bark and the 'blowing' of insects off the bark. Over the next thirty minutes we brush down the bark gently using a camel-hair paintbrush, finally removing the catch in the vial placed in the collecting hoop. Ensure a sample label identifying the tree species and replicate of that species has been placed in the vial. We then mark each tree with surveying tape to ensure it is not inadvertently re-sampled. This surveying tape is removed at the completion of all sampling. Routinely we sample thirty or forty trees on the one-hectare plot in this fashion. Generally we sample ten each of the commonest larger trees on the plot, as identified by our vegetation survey.

Two people can comfortably carry out bark sampling procedures and should be able to sample five to six trees sequentially (depending on the availability of suitably modified collecting hoops. A team of two can sample thirty or more trees over a two day period. Figure 25 below is a sample result from bark spraying one species of tree.

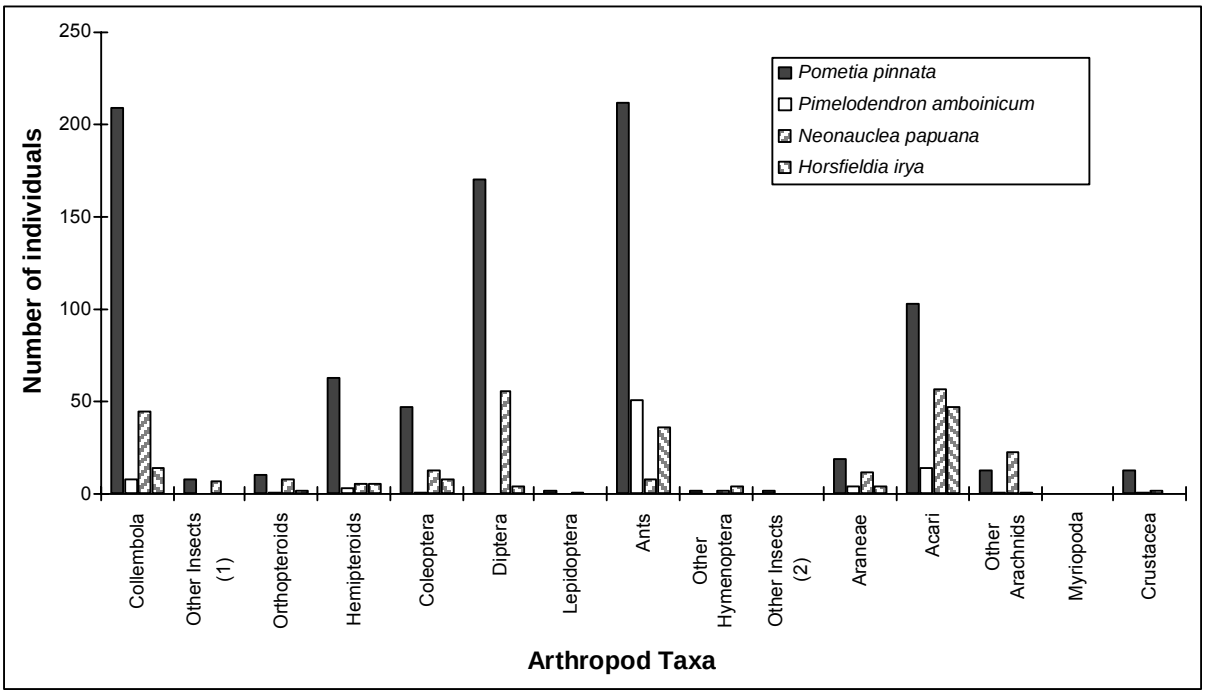


Figure 25: An ordinal profile from bark spray sampling of four tree species from tropical rainforest at Baitabag Village, Madang, Papua New Guinea.

Special Notes

Important tips for bark collecting include the following:

- The labels of bark collections must include the species of tree involved. Ensure this is clearly entered on the label so that those who sort the samples can read it easily. Any abbreviations used need to be self evident and standard.
- Do not routinely sample the same aspect of every tree. Spread the samples on different trees of the same species around all points of the compass.
- Avoid segments of bark that have foliose epiphytes on them. Encrusting epiphytes cannot be avoided but dense mats of moss or liverwort, or climbers in leaf, should be avoided.

Equipment List

The equipment is given for one team of sprayers:

- half hoop
- 10 x large map or thumb tacks
- string with knots tied at the corners of a half metre square (i.e. at 500 mm, 1500 mm, 2000 mm, 3000 mm from the end)
- collecting vials
- 80% ethanol
- sample labels
- paintbrush
- can of household insecticide
- stopwatch

SPECIMEN HANDLING AND DATA ENTRY

Having spent so much time, effort and money on sampling thus far, data in the form of samples becomes both highly valuable, and essentially irreplaceable. Careful and well planned data trails are essential to avoid both loss and errors in data. There are many points at which data can be lost or altered and it is essential that volunteers be fully briefed on the need for following protocols. It is also wise to ensure at least one staff member is made responsible for careful checking and recording of data. This person will also be responsible for recording general information about the survey in a master file. This should include:

- trapping days, dates and locations (coordinates) of all samples;
- any missed samples (e.g. light trap failure) and subsequently replacement samples;
- daily weather conditions; and
- the position and altitude of the plot (obtained using a GPS if available).

SPECIMEN HANDLING

This section deals with the treatment of samples after they are brought from the field into the field laboratory. The handling of the light trap catches has been discussed above and so the following account refers to the 'wet' material only. Arthropod samples return from the field in a variety of conditions. These vials contain arthropods in ethanol and may contain plant and soil remains, water and other contaminants. Sorting the arthropods from the rest can be a time-consuming process but the first task is to ensure that material remains well preserved until the sorting process can begin. If there is any possibility that the samples have been contaminated with water, then they should be drained and part filled with fresh 80% ethanol. Similarly, any bottles that have dried up need to be replenished.

On arrival in the field laboratory every sample should be checked to ensure it contains a sample label. Labels can then be added while the origin of the sample is still known. Sorting samples without labels is simply a waste of time and any samples that do not have labels when they come to be sorted should be discarded. On arrival in the laboratory sample bottles are entered onto a master datasheet designed specifically for the purpose (Appendix 7). As they are sorted, this too can be registered on the same sheet. This logging of samples in and out enables any omissions and errors to be traced

SORTING AND IDENTIFICATION

Exact sorting procedures differ from worker to worker and each person sorting will develop their own detailed approach. The protocol we follow is described in detail below. In general the sample is emptied (or part-emptied if it is large) into a Petri dish which has had a grid drawn or scratched onto its underside. This is then searched repeatedly under a binocular microscope. All arthropods are identified to Order (in most cases), removed and placed in small vials. Each category of arthropod has a separate vial. For the more abundant categories (such as beetles, flies, ants etc) we have found it most efficient to search through a sample picking out and counting all the individuals of a particular category (or two categories in some cases). Once the common categories are removed, then more general sorting for the rarer material can be carried out until no arthropods remain in the sample. In general, fine (#8) watchmaker's forceps are ideal for handling material but occasionally a fine paintbrush or pipette is more efficient. Mites (Acari), in particular, generally need a pipette for efficient handling.

We sort immediately to Order with two exceptions. We keep 'ants' separate from 'Other Hymenoptera' because (a) it is easy to do, and (b) different specialists generally deal with the two groups. We also separate Homoptera and Heteroptera, again because it is easy to do. Immature and adult examples of the same group are placed and counted together. With a new sorting team we precede any sorting with a lecture on how to recognise the more common categories. For a beginner, learning to confidently recognise the Coleoptera and the Diptera for example, means that they can begin work on a sample almost immediately. We provide each new worker with an illustrated manual to the more common groups and we have on hand a reference library of key manuals (see Appendix 5). Most important of all, though, we have one or two well-trained experienced general entomologists in the sorting room to assist the sorters at any time. They also provide the on-the-spot more advanced training required. We do NOT recommend the use of Keys to Orders for beginners. These are often technical, very time-consuming and seldom comprehensive. A professional entomologist will see a greater variety of arthropods in one survey expedition in a rainforest than she or he will have seen in a lifetime of work elsewhere. All the obscure groups which are the 'exceptions' in the keys will turn up from time to time – wingless Diptera, coleopteroid Hymenoptera, male scale insects and so on. Tackling this immense diversity needs experience, not keys.

We have found that we can train virtually anybody to sort efficiently to the level we require in two or three days although constant supervision and checking is provided. The more they persevere, the better they become. Some categories give more difficulty than others. The variety of body forms in the Collembola and the Homoptera both require considerable exposure before a sorter can confidently pick all the individuals involved. Sorting Heteroptera from some beetles, tiny beetles from oribatid mites, Psocoptera from some Homoptera, and larvae in general all need special attention.

The counts of each category of arthropod are entered on a pre-printed tally sheet, which has the locality, date, type of trap and trap code entered onto it (Appendix 3). Sorters tally individual organisms as they count them, place animals of particular Orders into separate vials, and then total each category. These tally sheets are VERY IMPORTANT. Once completed they are collected together and the information on them transferred to an Excel data file on a computer (see Appendix 6). The tally sheets are retained and securely stored so that actual counts can be checked at a later date if any questions arise over accuracy. Finally each vial is topped up with 80% ethanol to within 50mm of the top and a plastic stopper is securely inserted ready for long-term storage (see Storage below).

SORTING PROTOCOL

The following provides a step-by-step procedure for sorters to follow. In initially introducing volunteers to sorting we brief them on health and safety in the lab, the need for accurate data transfer and checking and a brief description of the major orders and their distinguishing features.

1. Sample issued and logged on master sample sheet (Appendix 7).
2. Contents of the vial are emptied into a Petri dish. The vial and lid are washed out into Petri dish using a wash bottle of 80% ethanol to ensure all organisms are removed.
3. The sample label is removed using forceps and washed into the Petri dish again to ensure no organisms are stuck on the label. The trapping type, sample number and date of trapping are then transcribed onto a sorting sheet (Appendix 3).
4. The contents of the Petri dish are scanned for organisms. Once identified the organism is lifted out of the Petri dish and placed in an individual (10 ml or 12 ml) vial. All organisms from an individual order are placed into a single vial. We identify the contents of each small vial during the sorting process by writing the Order onto the wooden sorting trays in

which the vials stand. The individual is scored using a tally mark on the sorting sheet against the appropriate Order (Appendix 3).

5. The Petri dish must be scanned methodically (e.g. left to right) and should be rescanned at successively lower magnifications of the microscope until no organism is found on a complete scan.
6. At this point one of our trained staff will check both the residue and the sorted vials. For new sorters this invariably reveals a few missed and mistakenly identified individuals.
7. Once checked, individual taxon labels are filled in for each vial identifying the sample number, taxon and date of survey (see example page 3) and placed into the vials. The vials are topped to about three-quarters full with ethanol and a cap fitted.
8. The data sheet is tallied both for total individuals in each taxa and the total number of individuals. The total number of Orders represented by individuals on the sorting sheet is also recorded.
9. The sorted vials are bundled together with a rubber band and the original sample label. The number of vials are counted and checked against the number of Orders recorded on the sorting sheet. The sorting sheet is wrapped around the bundle of vials and secured with another rubber band.
10. The staff member responsible for the data recording marks this back in against the master data log (Appendix 7). Before removing the data sheet for data entry, the information on the sheet can be checked against the original sample label and the individually labelled taxon vials. Any mistakes can be corrected before the sheet is separated from the sample.

LABELLING

A sample that has been sorted into ordinal categories occupies ten to twenty small vials rather than a single sample container. Each of these vials needs a label, which carries forward the label information that was with the original sample. We pre-print such labels with locality and trap-type information on it, with spaces for insertion of the date of collection, the category of animal contained in the vial, and the sample number. An example of this type of taxon label is given on page 3.

STORAGE

Material is usually returned from the field laboratory in bundles of vials for each sample. Once in its permanent home the bundles of vials are stored so that all the vials containing insects of the same Order – or other category – from the same trapping method are put together. Surveys of this kind produce large quantities of material, which soon present storage problems. The way in which this is handled is usually a compromise between the ideal and the possible.

Ideally all vials should be placed in jars containing a base of cotton wool, kept part-filled with ethanol, and should be stored at about 4°C until further sorting is required. In addition soft-bodied groups should be retained in 100% ethanol, rather than 80% ethanol, so that internal characters are retained for subsequent taxonomic analysis. In practise we store our vials in jars of 80% ethanol as suggested. Most groups are retained at room temperature in an air-conditioned laboratory. Collembola and Hymenoptera, as far as space permits, are stored in a freezer.

We routinely mount Coleoptera using ‘points’ once the survey is over. Other groups either receive further attention while in ethanol, such as immediate sorting to the family or species level by others who specialise in a particular taxon or taxa; or are stored for future long term study. All our material ultimately will be passed to the Australian National Insect Collection.

We urge all those carrying out large-scale surveys to make appropriate arrangements for the permanent retention of their material in a national collection of an appropriate sort.

DATA STORAGE AND HANDLING

The process of sorting several replicates of each trapping method to order obviously creates a great deal of data. This data is analysed within our own labs (see Data Analysis: Arthropod Survey, page 53), but may also be needed at any time in the future, possibly by experts in particular fields from outside our group. For these reasons, we have adopted a simple yet comprehensive and flexible approach to data storage and handling.

Microsoft Excel is used to tabulate all the raw data. Excel has been chosen as: it is easy to use, for both our team and for volunteers in the field; it is readily available and widely used, allowing easy transfer of information to others; it facilitates easy data manipulation and simple statistical analysis; and it allows for easy data transfer and reformatting for use in more sophisticated statistics packages.

A workbook is created for each trapping method with a separate sheet for each catch (e.g. a workbook for Yellow Pan traps with a worksheet for each day's catch). Each worksheet has the site, time of year and name of trapping method at the top. A list of each order (and other groups) is listed down the rows. It is important that there be a zero entry within every cell. These are then replaced with the total number of each order that is found for that replicate of that trap method. If the zeros are missing, Excel will incorrectly calculate values such as means and standard errors. Further worksheets are then created with summary data such as totals, counts, means, standard errors, and proportions. These can then be graphed or used for further analysis elsewhere.

Standard Excel workbooks are created before going into the field, with basic headings and zeros in place. In the field laboratory, provided conditions are suitable (i.e. there is electricity), a portable computer is used by a volunteer to enter data as sorting is completed. It is preferable that one person be responsible for this, to ensure consistency in all data entry.

BACKUPS

Another important aspect to data entry, one that cannot be over emphasised, is the backing up of work done. We achieve this two ways – firstly a print of completed data is made and secondly a backup to external media, such as a CD ROM or external memory device, is made periodically as work progresses, and again once counting is finished. Upon completion of data entry, it is prudent to make a separate backup to be kept separately off site. Should a major event, such as fire, happen in the normal work place, this raw data can then be quickly accessed. Another way to maintain a record of all work is of course a printed copy. If the information printed is considered crucial, (e.g. raw data as opposed to a summary which could be easily reproduced) then a copy of this too should be kept off site. All original data sheets (sorting sheets) are stored in the lab or office. These should never be thrown away, even after data is entered.

DATA ANALYSIS

VEGETATION DATA

As in any ecological analysis, the procedures used will reflect the questions being asked. Here we focus on biodiversity assessment, but suggest other ways in which this data can be used, primarily to discuss the floristic composition and structure of study sites and the distribution of species within them.

Diversity

The measurement of diversity allows the richness and relative abundance of life forms within a community to be assessed. Although there are many methods of diversity assessment, only those most commonly used in vegetation community analysis are discussed here. Calculation of a diversity measure, such as the Shannon-Wiener Index of diversity, combines in a single number the number of species (richness) and the relative distribution of numbers of individuals of those species (evenness). These two basic components are also useful (and somewhat easier to understand) when considered separately. Richness is simply the count of species present at a site, in a sample, or in a region. Evenness may be measured using a variety of indices related to the more complex indices of diversity (see, e.g. Southwood 1978, Magurran 1988).

Floristic Composition

One of the first things that you may wish to assess about the sites you have studied is their floristic composition. Initially, a list should be compiled of all species and their relative abundances for each site examined. The relative abundances of each family, genera and species can then be ascertained. It may be necessary to use higher classifications such as order, depending upon the types of questions you wish to answer with your analysis.

Once the floristic composition of each study site is ascertained, it is then possible to make within and between site comparisons. The relative abundances of each taxonomic grouping can be compared using a variety of statistical software packages such as SAS or Microsoft Excel, for example, an analysis of variance procedure (ANOVA) may be used to determine if a significant difference exists at each of these levels at each study site.

It is likely in areas of high vegetation diversity that many of the species encountered will be at low abundances. In order to simplify analysis, it may be necessary to place restrictions on which species are included. One way to decide which species are most influential in an area being studied is to isolate the information from those species which make up a large percentage of the total number of stems, for example, those species which make up 85% of the total abundance, or basal area, of plant species. Concentrating on these ‘dominant’ species may allow the major vegetation associations at each site to be identified.

Comparing the floristic composition of several sites is most simply done by determination of the degree of similarity among them. This may also help to identify locally endemic species. Many similarity indices are available (see, e.g. Wolda 1981). The simplest of these, based only on presence or absence of species, is the Sørenson measure. Such measures have the advantage of simplicity but do over-emphasise the presence of rare species. More sophisticated measures such as the Bray-Curtis metric and the Morisita-Horn Index circumvent these problems (see Wolda 1981, Southwood 1978). Magurran (1988) provides ready access to the necessary formulae and presents very useful sample calculations of each.

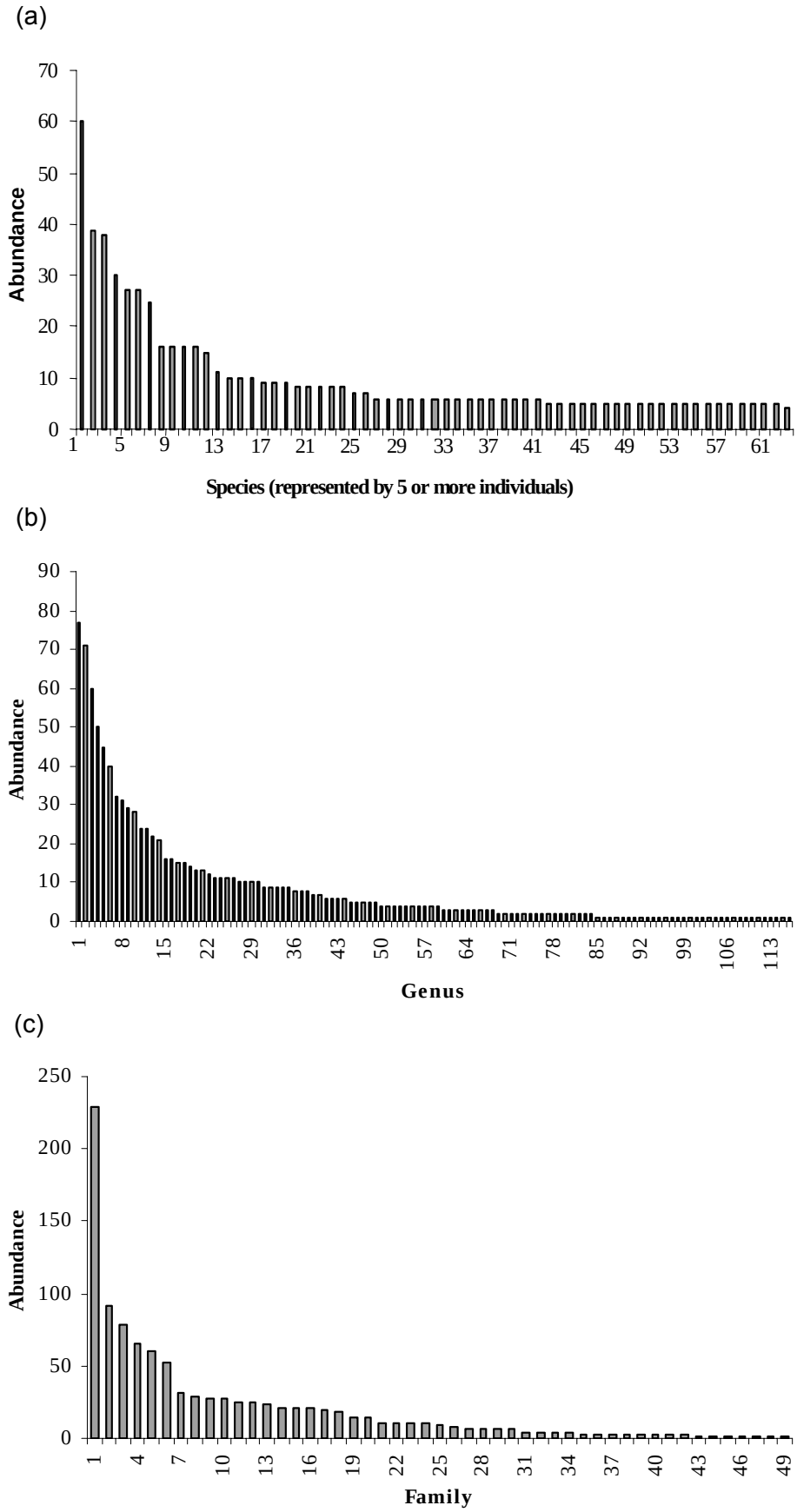


Figure 26: Examples of taxon/abundance plots from the one-hectare plot at Kuala Belalong, Brunei, (a) species level (shown are species represented by ≥ 5 individuals), (b) generic level and (c) family level.

Structure

After identifying the dominant species at each study site it is possible to determine the mean basal area, diameter and height for each of these species. Diameter measurements can be used to calculate the basal area of each tree at a height of 1.3 m by using the formula for the area of a circle, πr^2 (where r = dbh/2). This can be summarised for each species by finding the mean basal area for each species. Comparisons among species can be simply made using standard statistical methods. The total basal area at each site can then be determined by adding the totals for all individuals. The total basal areas of several sites can then be compared by using an ANOVA procedure to determine if any sites are significantly different.

Once the average heights and diameters are found for the most dominant species, a simple way to present the structural complexity of a study site is to construct a forest profile diagram (see Unwin 1989). Average attributes for the species present on the plot can be drawn to scale allowing the forest structure to be easily visualised (Figure 27).

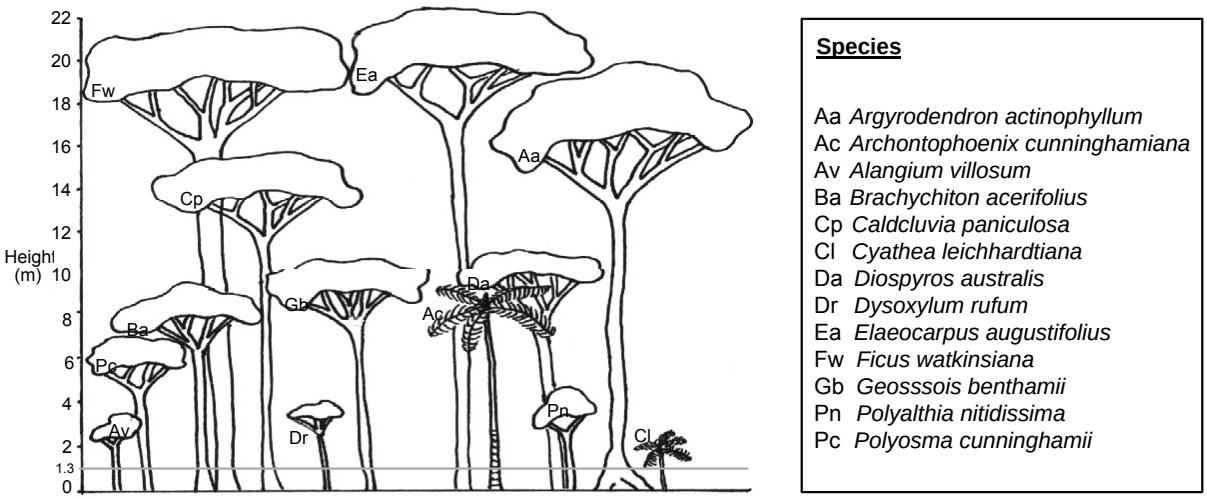


Figure 27: Vegetation profile for part of the one-hectare reference site at Lamington National Park, southeast Queensland.

Distributions

A further obvious and useful way to analyse data from the tree surveys is to examine the two-dimensional distribution of the trees on the plot. In its simplest form, such analysis allows discrimination between aggregated and more uniform dispersion patterns for individual species, groups of species or particular size classes of trees. The distribution of trees on a study plot is most easily studied through graphical presentation as x and y coordinates. This can be done using graphing packages such as Microsoft Excel or Cricket Graph (Figure 28). Alternatively, a mapping or Geographical Information System (GIS) package such as ArcGIS™ can be used. By identifying patterns of distribution within or between species, we can develop hypotheses regarding species, environmental or landscape attributes which may have produced the observed patterns. These include questions of dispersion, size distribution and/or the detection of disturbance. Size classes, height classes and particular groups of species can be mapped through the use of GIS packages. Dispersion patterns of trees within a plot may be investigated either using an approach that sub-samples the hectare and then compares the distribution of numbers within quadrats against those predicted from a Poisson distribution or using a nearest neighbour analysis. The latter method is useful where trees are widely dispersed (Southwood 1978).

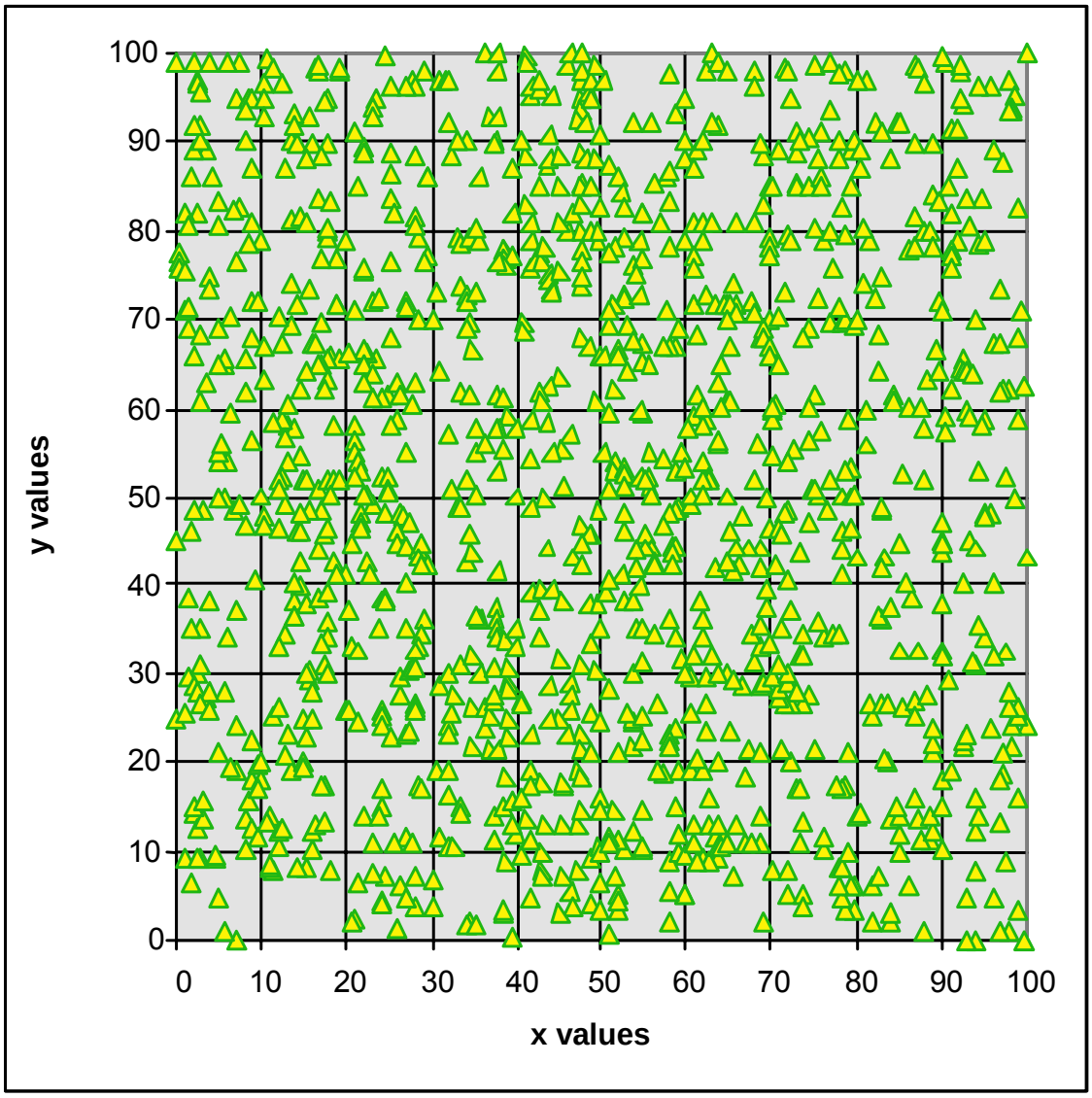


Figure 28: Graphical presentation of spatial distribution of all individuals on the Lamington National Park one-hectare plot.

The spatial association among species can also be established in order to determine species alliances within a habitat type. Again, these patterns are likely to be induced by environmental conditions. These associations may be positive or negative and can be determined by comparing replicate samples of species abundances from within or between the one-hectare plots. The significance of these associations can be determined by calculating the correlation coefficient between species counts or by using a contingency table of frequency classes.

Environmental Variables

In order to identify the possible causes of patterns observed in a vegetation survey, it may be useful to examine abiotic variables such as annual rainfall, temperature, latitude, altitude, soil type or topography. Each of these variables can be compared with information on the number of stems, diversity, evenness, number of families and so forth using standard methods of linear regression. This analysis will identify relationships between calculated variables and their correlates. The same process may be used to identify relationships among measured variables (which are unlikely to be statistically independent) such as the number of stems and diversity, the number of families and the number of species.

ARTHROPOD DATA

There are many ways in which the arthropod data that accrue from surveys that use this protocol may be analysed and we do not intend to give a full account here. Some general points can be made however about data reduction, statistical summaries, and a productive approach to data analysis through a combination of multivariate analysis and analyses of variance. A similar approach is taken to the analysis of the vegetation.

Data Reduction

The data obtained during these biodiversity surveys are multivariate with each datum representing the numbers of individuals of each category of arthropod obtained by a particular trapping method on a particular day. There may be as many as 38 categories in one line of data, each representing the numbers of individuals of each and every taxonomic category present in the full set of samples. In general this will mean that many of the entries in the overall data matrix will be zero. For example we seldom catch Mecoptera, Strepsiptera, Embioptera or Megaloptera, although these do appear in the samples on some occasions. For analytical purposes a data matrix that is filled with zeroes is harder to deal with than one with only a few zeroes.

So we have analysed our data initially by combining the counts for related groups of Orders as outlined in Table 3 below.

Table 3: Taxonomic groupings used in data analysis.

Taxa Category	Orders included
'Collembola'	Collembola
Other Apterygotes and Exopterygotes ('Other Insects 1')	Protura, Diplura, Archaeognatha, Thysanura, Ephemeroptera, Odonata
'Orthopteroids'	Orthoptera, Dermaptera, Phasmatodea, Mantodea, Blattodea, Isoptera, Embioptera
'Hemipteroids'	Homoptera, Heteroptera, Psocoptera, Thysanoptera
'Coleoptera'	Coleoptera
'Diptera'	Diptera
'Lepidoptera'	Lepidoptera
'Ants'	Ants
'Other Hymenoptera'	Other Hymenoptera
Other Endophtygotes ('Other Insects 2)'	Plecoptera, Megaloptera, Neuroptera, Strepsiptera, Mecoptera, Trichoptera
'Araneae'	Araneae
'Acari'	Acari
'Other arachnids'	Opiliones, Scorpiones, Pseudoscorpiones
'Myriapoda'	Diplopoda, Chilopoda, Symphyla
'Crustacea'	Isopoda, Amphipoda, Copepoda

This reduces a cumbersome 38 categories to a much more amenable fourteen. The way in which this reduction is done is entirely for convenience and different workers can do it in different ways. Of course, any reduction in data in this fashion does not prevent the individual categories being analysed separately in more detail.

In addition to reducing the number of categories in this fashion the data may also be reduced by combining catches from different samples. We have already discussed the way in which catches from individual pitfalls are combined in the field, and the counts from the twenty collecting hoops of each canopy misting are combined. Where a four (or any other) day period of collection is used then the catches from individual days may be combined or left separately. For simple analyses we have combined the daily catches and then computed a number *per sampling effort* as a means of standardising the data. Individual daily catches can be retained, however, and 'day' entered as a treatment variable or co-variate in the subsequent statistical analyses.

We standardise our catches as follows:

Litter Extraction:	per gram (or kilogram) dry weight of litter
Pitfall Traps:	per trap array per day
Yellow Pan Traps:	per trap per day
Malaise Traps:	per trap per day
Light Traps:	per trap per day
Flight Intercept Traps	per trap per day
Canopy Knockdown:	per 0.5 m^2 collecting hoop (or per 20 hoops)
Bark Spraying:	per 0.5 m^2 of bark

STATISTICAL SUMMARIES

Raw data is seldom directly useable. The process of pattern description is best begun by a process of summarising the data. The most obvious way of doing this is to calculate a set of summary statistics. There are many desktop packages that make this process very simple.

We use any or all of the following measures:

Mean:	the most fundamental and instructive statistic of all
Maximum, Minimum:	statistics capturing the *range* of values of the data
Standard Error:	the most widely used measure of *central tendency* in the data
Coefficient of Variation:	a measure of central tendency standardised by the mean value

These summarising statistics are also usefully expressed as a set of histograms of numbers (y axis) against categories (x axis). Such frequency plots also allow for the identification of higher statistical moments such as skewness and kurtosis. Remember that a histogram of means without any indication of the standard errors may be very misleading. We have used the graphing package CricketGraph™ (Computer Associates Software) and Microsoft Excel for the production of graphics in the past, but many alternate packages are now available.

We summarise data separately based on raw *abundances* and on *proportions*. The proportions represented by each taxon as a fraction of the entire sample will capture

assemblage differences between locations or sampling methods better than comparing mere abundances because proportions are independent of the overall size of the sample. The overall size of the sample may be influenced by many uncontrolled factors that do not necessarily represent genuine differences in assemblage structure between locations.

Transformations

Data need only be transformed if it is drawn from a larger 'population' of numbers, which deviate substantially from a normal distribution. Simple tests of normality will confirm whether or not such transformations are required. In general a logarithmic transformation of the raw counts will compensate for occasional very high numbers. Proportions may be properly transformed using an arc-sine square root function to compensate for the zero-to-one boundedness of proportional data. When making comparisons across sites, times or sampling methods either all or none of the data should be appropriately transformed.

MULTIVARIATE ANALYSES

Biodiversity inventory is a measure of *pattern*. This pattern may be measured to test pre-stated hypotheses or the analysis of pattern may be a pre-hypothesis exploratory stage. Biodiversity data is multivariate where each data item is a set of numbers representing the abundances (or proportions) of each taxon in a particular sample. This lends itself to multivariate pattern analysis in which each of these n-dimensional points is plotted in n-space by an analytical package (we use, principally, the PATN package developed in Australia by CSIRO – see Belbin 1995). The way in which these points are grouped in multi-dimensional space and the degree to which particular taxon-counts affect these groupings are then used to erect hypotheses about the factors which generate this pattern. These can then be further investigated using a variety of approaches either by further multivariate analyses or using simple or multiple analyses of variance (see below).

There is a great diversity of books written on multivariate analysis (see, for example, Digby and Kempton 1987, Draper and Smith 1966, Jongman *et al.* 1987. Krzanowski 1988, Zar 1996) and this is not an appropriate place to give an account of the many alternative methods available. We have used both classificatory and ordination analyses in analysing data. In a classificatory analysis, trees of similarity are constructed using one of a number of alternative multiple correlation techniques. These indicate the degree to which particular data sets are similar or different from each other. This enables us to decide which sampling methods give the most novel information, which sets of sites across a latitudinal gradient (for example) are most alike, or the degree to which the assemblage structure changes with season. We have used a hierarchical agglomerative clustering algorithm based on Bray-Curtis measures of dissimilarity. In addition, the simpler measures of taxon overlap such as Sørensen's Index (based on the presence or absence of species only) and the Morisita-Horn Index (which also incorporates relative abundance measures) (Wolda 1981, Magurran 1988) has provide useful.

Ordination methods plot the data in n-space and then analyse the associations of points so formed. In general we expect that data collected from the same site, or using similar methods, or collected at the same time of year should cluster together in some interpretable fashion. We have used two contrasting methods (of the many available) in our analyses.

Multi-dimensional scaling (MDS) approaches this problem by representing the multi-dimensional distances between samples (in the n-space reflecting the n taxa in the sample under analysis) and then reducing this n-dimensionality to a much smaller number of dimensions (usually two or three) in order to simplify ecological interpretation of patterns in the data. Such ordinations can then be overlain with regression-based vectors reflecting the occurrence of particular taxa across sites to suggest which groups of animals are producing

the observed patterns. Once such 'key' taxa are identified they can be further analysed either by using more sophisticated multi-variate methods or by separate analyses of variance in which the selected taxa are taken as the response variables. This second phase of the analysis represents the testing of specific hypotheses generated by the earlier general pattern analysis.

In some more specialised analyses we have also used principal components analysis (PCA) in which the eigenvectors of the variable-to-variable correlation matrix among taxa are used to define new meta-variables (the principal components) which capture the underlying pattern of assemblage variation. Within each of these principal components, the relative loadings of each taxonomic group are available and the relative importance of each can be assessed.

ANALYSIS OF VARIANCE

The analysis of variance (ANOVA) and its multi-variate equivalent (MANOVA) are the most used and best known techniques in ecology. In our analyses this involves attempting to assess the site-to-site ('treatment') effects upon selected response variables. These response variables may usefully be composites based on the whole taxonomic assemblage (such as species richness values, number of taxa, evenness, dominance or other diversity measures, total numbers of individuals, etc.) or the numbers or proportions of individuals or species belonging to particular taxa. The approach can either be a broad one in which each taxonomic group, or other statistic, is analysed in turn and the formally significant ones so identified. Alternatively, it may be based on the analysis of previously selected taxa. Selections of taxa may be on *a priori* grounds following inspection of graphs and histograms, or derived from the result of multivariate analyses. The first approach runs the risk of Type II errors as more and more response variables are analysed, the second offers Type I errors if the impacts of particular taxa on the overall assemblage pattern have been masked in the multivariate analyses.

Again this is not the place to discuss the massive subject of analyses of variance in ecology. The recent work by Underwood (1997) provides a comprehensive treatment of many of the issues involved.

OTHER ANALYTICAL METHODS

There are a growing variety of other methods available for the analyses of communities, and our choices have been and will remain personal ones. We note in particular the growing importance of Analyses of Similarity (ANOSIM) (see Clarke and Green 1988, Rodgers and Kitching 1998), and the suite of species estimation techniques produced by Colwell, named EstimateS (http://viceroy.eeb.uconn.edu/EstimateS).

REFERENCES

Belbin, L. (1995). *Users Guide. PATN Pattern Analysis Package*, CSIRO, Australia.

Bogush, P. P. (1958). Some results of collecting click beetles (Coleoptera, Elateridae) with light traps in Central Asia. *Entomological Review* **39:** 291-299.

Bowden, J. (1973). The influence of moonlight on catches of insects in light traps. Part I. The moon and moonlight. *Bulletin of Entomological Research* **63:** 113-128.

Bowden, J. and Church, B. M. (1973). The influence of moonlight on catches of insects in light traps. Part. II The effect of moon phase on light trap catches. *Bulletin of Entomological Research* **63:** 129-142.

Briggs, J. B. (1961). *A Comparison of Pitfall Trapping and Soil Sampling in Assessing Populations of Two Species of Ground Beetles (Col.: Carabidae)*. Report of the East Malling Research Station 1960**,** 108-112.

Clarke, K. R. and Green, R. H. (1988). Statistical design and analysis for a 'biological effects' study. *Marine Ecology Progress Series* **46:** 213-226.

Constanza, R., d'Arge, R., de Groot, R., Farber, S., Grasso, M., Hannon, B., Limburg, K., Naeem, S., O'Neill, R. V. O., Paruelo, J., Raskin, R. G., Sutton, P. and van den Belt, M. (1998). The value of the world's ecosystem services and natural capital. *Nature (London)* **387:** 253-260.

Digby, P. G. N. and Kempton, R. A. (1987). *Multivariate Analysis of Ecological Communities*. Chapman and Hall, London.

Draper, N. R. and Smith, H. (1966). *Applied Regression Analysis*. John Wiley, New York.

Erwin, T. L. (1982). Tropical forests: their richness in Coleoptera and other arthropod species. *Coleopterists Bulletin* **36:** 74-75.

Erwin, T. L. (1990). Canopy arthropod biodiversity: a chronology of sampling techniques and results. *Revista Peruana de Entomologia* **32:** 71-77.

Farrell, B. (1998). "Inordinate fondness" explained: why there are so many beetles. *Science (New York)* **281:** 555-559.

Ford, J. (1937). Fluctuations in natural populations of Collembola and Acarina. *Journal of Animal Ecology* **6:** 98-111.

Frost, S. W. (1952). Light traps for insect collection, survey and control. *Bulletin of the Pennsylvania Agricultural Experimental Station* **550:** 32pp.

Frost, S. W. (1957). The Pennsylvania light trap. *Journal of Economic Entomology* **50:** 287-292.

Gagné, W. C. (1979). Canopy-associated arthropods in *Acacia kea* and *Metrosideros* tree communities along an altitudinal transect on Hawaii Island. *Pacific Insects* **21:** 56-82.

Geier, P. W. (1960). Physiological age of codling moth females (*Cydia pomonella*) caught in bait and light traps. *Nature (London)* **265:** 415-421.

Greenslade, J. (1973). Sampling ants with pitfall traps: digging–in effects. *Insectes Sociaux* **20:** 343-353.

Greenslade, P. and Greenslade, P. J. M. (1971). The use of baits and preservatives in pitfall traps. *Journal of the Australian Entomological Society* **10:** 253-260.

Gressitt, J. L. and Gressitt, M. K. (1962). An improved Malaise trap. *Pacific insects* **3:** 549-555.

Grove, S. J. (2000). Trunk window trapping: An effective technique for sampling saproxylic beetles. *Memoirs of the Queensland Museum* **46:** 149-60.

Harper, A. M. and Story, T. P. (1962). Reliability of trapping in determining emergence period and sex ratio of the sugar beet maggot *Tetanops myopaeformis* (Röder) (Diptera: Tephritidae). *Canadian Entomologist* **94:** 268-271.

Hollingsworth, J. P., Briggs, C. P., Glick, P. A. and Graham, H. M. (1961). Some factors influencing light trap collections. *Journal of Economic Entomology* **54:** 305-308.

Jongman, R. H. G., ter Braak, C. J. F. and van Tongeren, O. F. R. (1987). *Data Analysis in Community and Landscape Ecology.* Pudoc, Wageningen.

Juillet, J. A. (1963). A comparison of four types of trap used for capturing flying insects. *Canadian Journal of Zoology* **41:** 219-223.

Kitching, R. L., Bergelson, J. M., Lowman, M. D., McIntyre, S. and Carruthers, G. (1993). The biodiversity of arthropods from Australian rainforest canopies: general introduction, methods, sites and ordinal results. *Australian Journal of Ecology* **18:** 181-191.

Kitching, R. L., Orr, A. G., Small, A., Martin T., Wong K. M. and Mitchell, H. (1995). *A multiple-method insect and plant inventory on a Kuala Belalong hectare*. Brunei Museum Journal.

Krzanowski, W. J. (1988). *Principles of Multivariate Analysis. A User's Perspective.* Clarendon Press, Oxford.

Luff, M. L. (1975). Some factors affecting the efficiency of pitfall traps. *Oecologia (Berlin)* **19:** 345-357.

Macfadyen, A. (1953). Notes on methods for the extraction of small micro-arthropods. *Journal of Animal Ecology* **22:** 65-78.

Macfadyen, A. (1955). A comparison of methods for extracting soil arthropods. In: McE. Kevan, D. K. (ed.). *Soil Zoology*. University of Nottingham, Nottingham.

Macfayden, A. (1962). Soil arthropod sampling. In: Cragg, J. B. (ed.), *Advances in Ecological Research 1*, Academic Press, London.

Magurran, A. (1988). *Ecological Diversity and its Measurement.* Princeton University Press, Princeton.

Malaise, R. (1937). A new insect trap. *Entomologische Tidjschrift* **58:** 148-160.

Martin, J. L. (1966). The insect ecology of red pine plantations in central Ontario. IV. The crown fauna. *Canadian Entomologist* **98:** 10-27.

Minet, J. (1991). Tentative reconstruction of the ditrysian phylogeny. *Entomologica Scandinavica* **22:** 69-95.

Mitchell, B. (1963). Ecology of two carabid beetles, *Bembidion lambros* (Herbst.) and *Trechus quadristriatus* (Schrank). II. *Journal of Animal Ecology* **32:** 377-392.

Moericke, V. (1951). Über den Farbensinn der Pfirsichblattlaus *Myzodes persicae* Sulz. *Zeitschrift für Tierpsychologie* **7:** 265-274.

Paris, O. A. and Pitelka, F. A. (1962). Population characteristics of the terrestrial isopod *Armadillidium vulgare* in California grassland. *Ecology* **43:** 229-248.

Parker, S. P. (ed.) (1982). *Synopsis and Classification of Living Organisms.* McGraw-Hill, New York.

Roberts, R. H. (1970). Color of Malaise traps and collection of Tabanidae. *Mosquito News* **39:** 567-571.

Roberts, H. R. (1973). Arboreal Orthoptera in the rain forests of Costa Rica collected with insecticide: a report on grasshoppers (Acrididae) including new species. *Proceedings of the Academy of Natural Sciences, Philadelphia* **125:** 46-66.

Robinson, H. S. and Robinson, P. J. M. (1950). Some notes on the observed behaviour of Lepidoptera in flight in the vicinity of light-sources together with a description of a light-trap designed to take entomological samples. *Entomologists' Gazette* **1:** 3-15.

Rodgers, D. J. and Kitching, R. L. (1998). Vertical stratification of rainforest collembolan (Collembola: Insecta) assemblages: description of ecological patterns and hypotheses concerning their generation. *Ecography* **21:** 392-400.

Scoble, M. J. (1992). *The Lepidoptera, Form, Function and Diversity.* Oxford University Press, Oxford.

Southwood, T. R. E. (1978). *Ecological Methods with particular reference to Insect Populations.* Second Edition, Chapman and Hall, London.

Southwood, T. R. E., Moran, V. C. and Kennedy, C. E. J. (1982a). The richness, abundance and biomass of the arthropod communities of trees. *Journal of Animal Ecology* **51:** 635-650.

Southwood, T. R. E., Moran, V. C. and Kennedy, C. E. J. (1982b). The assessment of arboreal insect fauna: comparisons of knockdown sampling and faunal lists. *Ecological Entomology* **7:** 331-340.

Stork, N. E. (1987a). Guild structure of arthropods from Bornean rain forest trees. *Ecological Entomology* **12:** 69-80.

Stork, N. E. (1987b). Arthropod faunal similarity of Bornean rain forest trees. *Ecological Entomology* **12:** 219-226.

Stork, N. E. (1988). Insect diversity: facts, fiction and speculation. *Biological Journal of the Linnean Society* **35:** 321-337.

Taylor, L. R. and Taylor, R. (1977). Aggregation, migration and population mechanisms. *Nature (London)* **265:** 415-421.

Townes, H. (1962). Design for a Malaise trap. *Proceedings of the Entomological Society of Washington* **64:** 253-262.

Underwood, A. J. (1997). *Experiments in Ecology: their logical design and interpretation using analysis of variance.* Cambridge University Press, Cambridge.

Unwin, G. L. (1989). Structure and composition of the abrupt rainforest boundary in the Herberton Highland, North Queensland. *Australian Journal of Botany* **37:** 413-428.

Wolda, H. (1981). Similarity indices, sample sizes and diversity. *Oecologia (Berlin)* **50:** 296-302.

Zar, J. H. (1996). *Biostatistical Analysis.* Prentice Hall, Upper Saddle River, New Jersey.

APPENDIX 1: BIBLIOGRAPHY

Publications of the Arthropod Biodiversity Laboratory, Griffith University (and its precursor at the University Of New England, Armidale) resulting from biodiversity work based on the techniques described in this book, presented here in chronological order.

Jones, R. E. and Kitching, R. L. (1991). Biological inventory. In: Goudberg, N. and Bonell, M. (eds.), *Tropical Rainforest Research in Australia*. Institute for Tropical Rainforest Studies, Townsville.

Basset, Y. and Kitching, R. L. (1991). Species number, species abundance and body length of arboreal arthropods associated with an Australian rainforest tree. *Ecological Entomology* **16:** 391-402.

Kitching, R. L. (1992). Biodiversity, research and conservation in Australian rainforests. *Malayan Nature Journal* **45:** 31-54.

Kitching, R. L., Bergelson, J., Lowman, M. D., McIntyre, S. and Carruthers, G. (1993). The biodiversity of arthropods in Australian rain forest canopies: introduction, methods, study sites and ordinal results. *Australian Journal of Ecology* **18:** 181-191.

Kitching, R. L. and Arthur, M. (1993). The biodiversity of arthropods in Australian rain forest canopies: summary and the impact of drought. *Selbyana* **14:** 29-35.

Kitching, R. L. (1993). *Ecology, Biodiversity and the Future of Australia.* Inaugural Lecture, Griffith University, 40 pp.

Kitching, R. L. (1993). Rainforest canopy arthropods: problems for rapid biodiversity assessment. In: *Rapid Biodiversity Assessment, Proceedings of a Workshop.* Unit for Biodiversity and Bioresources, Macquarie University, pp. 26-30.

Kitching, R. L., Floater, G. and Mitchell, H. (1994). The biodiversity of arthropods in Australian rain forest canopies: ecological questions and management challenges. In: Yasuno, M. and Watanabe, M. M. (eds.), *Biodiversity: its Complexity and Role.* Global Environmental Forum, Tokyo, pp. 119-137.

McIntyre, S. M., Kitching, R. L. and Jessup, L. W. (1994). Vegetation structure in rainforest plots at Cape Tribulation, North Queensland*. Proceedings of the Royal Society of Queensland* **104:** 25-41.

Kitching, R. L. (1994). Biodiversity and taxonomy: impediment or opportunity. In: Moritz, C. and Kikkawa, J. (eds.), *Conservation Biology in Australasia and Oceania*. Surrey Beatty, Sydney, pp.253-268.

Kitching, R. L. (1994). Exploring the upper limits: insects of the rainforest canopy. *Wildlife Australia* Summer **1994:** 18-21.

Kitching, R. L. (1995). Biodiversity – political responsibilities and agendas for research and management. *Pacific Conservation Biology* **1:** 279-283.

Kitching, R. L. (1995). The sum of its parts: should we save individuals or ecosystems? *Earthwatch* Jan/Feb **1995:** 12-13.

Lowman, M. D., Kitching, R. L. and Carruthers, G. (1996). Arthropod sampling in Australian subtropical rain forests – how accurate are some of the more common techniques? *Selbyana* **17:** 36-42.

Kitching, R. L. and Zalucki, J. M. (1996). The biodiversity of arthropods in Australian rain forest canopies: some results on the role of the tree species. In: Booth, W. and Choy, S. (eds.), *Rain Forest Research: Current Issues.* Kluwer, Amsterdam, pp. 21-28.

Kitching, R. L. and Theischinger, G. (1996). The biodiversity of arthropods in Australian rain forest canopies: Tipulidae, with a description of the new species *Leptomastix alfie* Theischinger. *Entomologist* **115:** 140-153.

Kitching, R. L., Mitchell, H., Morse, G. and Thebaud, C. (1997). Determinants of species richness in assemblages of canopy arthropods in rainforests. In: Stork, N., Didham, R. and Adis, J. (eds.), *Canopy Arthropods.* Chapman and Hall, London, pp. 131-150.

Hammond, P. M., Kitching, R. L. and Stork, N. E. (1997). The biodiversity of arthropods from Australian rainforest canopies: Coleoptera from subtropical tree-crowns. *Ecotropica* **2:** 99-108.

Walter, D. E., Seeman, O., Rodgers, D. and Kitching, R. L. (1998). Mites in a mist: microhabitat distribution complementarity and body size distribution of Acari from a subtropical rainforest. *Australian Journal of Ecology* **23:** 501-508.

Rodgers, D. and Kitching, R. L. (1998). Vertical stratification of rainforest collembolan (Collembola: Insecta) assemblages: ecological patterns and hypotheses concerning their generation. *Ecography* **21:** 392-400.

Orr, A. G. and Kitching, R. L. (1999). A checklist of macrolepidoptera collected from rain forest and former forest areas on basalt soils on the Atherton Tableland. *Australian Entomologist* **26:** 15-27.

Thalib, L. and Kitching, R. L. (1999). Principal component analysis for grouped data – a case study. *Envirometrics* **10:** 565-574.

Kitching, R. L., Olsen, M. and Small, A. (1999). The use of higher taxonomic categories in the measurement of forest tree diversity: an example from Old-world rainforests. *Tropical Biodiversity* **5:** 185-195.

Kitching, R. L., Orr, A. G., Thalib, L., Mitchell, H., Hopkins, M. S. and Graham, A. W. (2000). Moth assemblages as indicators of environmental quality in remnants of upland Australian rain forest. *Journal of applied Ecology* **37:** 284-297.

Laidlaw, M., Olsen, M., Kitching, R. L. and Greenway, M. (2000). Tree floristic and structural characteristics of one-hectare of subtropical rainforest in Lamington National Park, Queensland. *Proceedings of the Royal Society of Queensland* **109:** 91-105.

Kitching, R. L. (2000). Biodiversity, hotspots and defiance. *Trends in Ecology and Evolution* **15:** 484-485.

Kitching, R. L., Vickerman, G., Laidlaw, M. and Hurley, K. (2000). *The Comparative Assessment of Arthropod and Tree Biodiversity in Old-World Forests: the Rainforest CRC / Earthwatch Protocol Manual.* Technical Report, Rainforest CRC, Cairns.

Kitching, R. L., Li, D. Q. and Stork, N. E. (2001). Assessing biodiversity 'sampling packages': how similar are arthropod assemblages in different tropical rainforests? *Biodiversity and Conservation* **10:** 793-813.

Kitching, R. L. (2001). Biodiversity survey of arthropods from Australia to Borneo: patterns and processes. In: Ganashalah, K. N., Shankar, R. U. and Bawa, K. S. (eds.), *Tropical Ecosystems: Structure, Diversity and Human Welfare.* Oxford-IBH, New Delhi, pp. 372-375.

Majer, J. D., Kitching, R. L., Heterick, B. E., Hurley, K and Brennan, K. E. C. (2001). North-south patterns within arboreal ant assemblages from rainforests in Eastern Australia. *Biotropica* **33:** 643-661.

Kitching, R., Basset, Y., Ozanne, C. and Winchester, N. (2002). Canopy knockdown techniques for sampling canopy arthropod fauna. In: Mitchell, A. W., Secoy, K. and Jackson, T. (eds.), *The Global Canopy Handbook.* Global Canopy Programme, Oxford, pp. 134-139.

Kitching, R. L. and Shaw, D. C. (2002). Volunteers: their use and management. In: Mitchell, A. W., Secoy, K. and Jackson, T. (eds.), *The Global Canopy Handbook,* Global Canopy Programme, Oxford, pp. 219-224.

Basset, Y., Novotny, V., Miller, S. and Kitching, R. L. (2003). *Arthropods of Tropical Forests: Spatio-temporal Dynamics and Resource Use in the Canopy,* Cambridge University Press, Cambridge, xvi +474pp.

Basset, Y., Novotny, V., Miller, S. and Kitching, R. L. (2003). Canopy entomology, an expanding field of natural science. In: Y. Basset *et al*. (eds.), *ibid.* pp. 4-6.

Basset, Y., Novotny, V., Miller, S. and Kitching, R. L. (2003). Methodological advances and limitations in canopy entomology. In: Y. Basset *et al*. (eds.), *ibid.* pp. 5-15.

Novotny, V., Basset, Y. and Kitching, R. L. (2003). Herbivore assemblages and their food resources. In: Y. Basset *et al*. (eds.), *ibid.* pp. 40-53.

Kitching, R. L., Hurley, K. L. and Thalib, L. (2003). Tree relatedness and the similarity of insect assemblages: pushing the limits? In: Y. Basset *et al*. (eds.), *ibid.* pp. 329-340.

Basset, Y., Novotny, V., Miller, S. and Kitching, R. L. (2003). Conclusion: arthropods, canopies and interpretable pattern. In: Y. Basset *et al*. (eds.), *ibid.* pp. 394-405.

Orr, A. G. and Kitching, R. L. (2003). A faunistic analysis of Macrolepidoptera from complex notophyll vine forest, North Queensland, Australia. *Journal of Natural History* **37:** 1537-1554.

Ozanne, C. M. P., Anhuf, D., Boulter, S. L., Keller, M., Kitching, R. L., Körner, C., Meinzer, F. C., Mitchell, A. W., Nakashizuka, T., Silva Dias, P. L., Stork, N. E., Wright, S. J. and Yoshimura, M. (2003). Forest canopies: understanding global ecosystems. *Science (New York)* **301:** 183-186.

Kitching, R. L., Orr, A. G., Small, A, Martin, T., Wong, K. M. and Mitchell, H. (2003) [dated 1995]. A multiple-method insect and plant inventory on a Kuala Belalong hectare. *Brunei Museum Journal* **1995:** 65-80.

Basset, Y., Mavoungou, J. F., Mikissa, J. B., Missa, O., Miller, S. E., Kitching, R. L. and Alonso, A. (2004) Discriminatory power of different arthropod data sets for the monitoring of anthropogenic disturbance in tropical forests. *Biodiversity and Conservation* **13:** 709-732.

Small, A., Martin, T., Kitching, R. L. and Wong, K. M. (2004). Contribution of tree species to the biodiversity of a one-hectare plot of Old-World rainforest, Brunei, Borneo. *Biodiversity and Conservation* **13:** 2067-2088.

Kitching, R. L., Bickel, D., Creagh, A. C., Hurley, K. and Symonds, C. (2004). The biodiversity of Diptera in Old-world rainforest surveys: a comparative analysis. *Journal of Biogeography* **31:** 1185-1200.

Menzel, F., Kitching, R. L. and Boulter, S. L. (2004). Host specificity or habitat structure? - The epicortical beetle assemblages in an Australian subtropical rain forest. *European Journal of Entomology* **101:** 251-259*.*

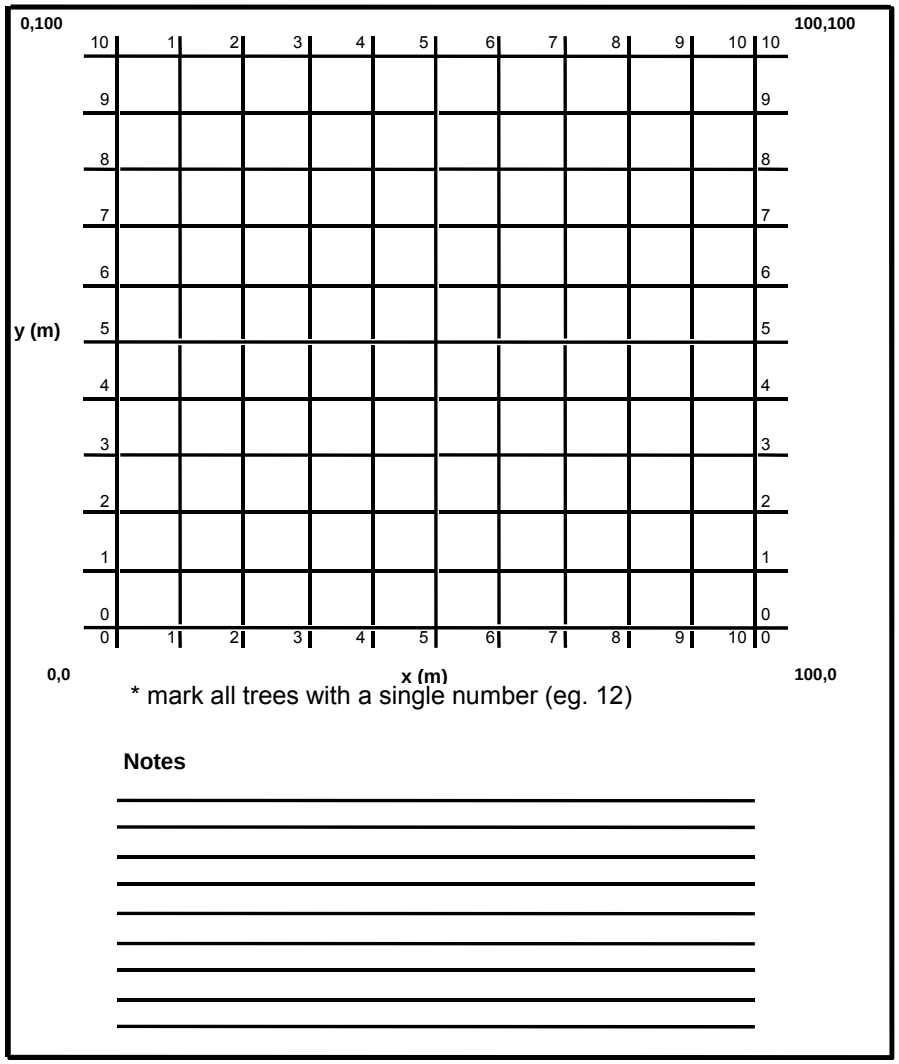
Laidlaw, M. J., Kitching, R. L., Damas, K. and Kiapranis, R. (In press). Tree floristic and structural attributes of two lowland rainforest plots in Papua New Guinea. *Biotropica.*

Kitching, R. L., Bickel, D., Boulter, S. L. (In press). Guild analyses of dipteran assemblages: a rationale and investigation of seasonality and stratification in selected rainforest faunas. In: Yeates, D. and Wiegmann, B. (eds.), *Evolutionary Biology of Flies,* Colombia University Press.

APPENDIX 2: SAMPLE VEGETATION SURVEY TALLY SHEET

Vegetation Biodiversity Survey
Site :
Date :
Quadrat : ___(_____,_____)_ __

Plant No.	Dbh (cm) at 1.3m	Height (m)	X coord	Y coord	Species
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
16					
17					
18					
19					
20					
21					
22					
23					
24					
25					
26					
27					
28					
29					
30					
31					
32					
33					
34					
35					



* mark all trees with a single number (eg. 12)

Notes

APPENDIX 3: SAMPLE ARTHROPOD TALLY SHEET

NB: This tally sheet includes only those Orders we have encountered to date in our surveys. Additional taxa may be encountered from time to time.

Site location e.g. Lamington National Park 1/4 ha	**LOCATION:**	*Lamington National Park*
Sample type e.g. Yellow Pan 1	**SAMPLE:**	*Yellow Pan 1*
This is the date of collection NOT the date sorted	**DATE ON LABEL:**	*5/01/04*
Person who does the sorting	**SORTED BY:**	*Roger Kitching*

Order	Tally	Total
Collembola	*IIII*	*4*
Diplura		
Archaeognatha		
Thysanura		
Ephemeroptera		
Odonata		
Plecoptera		
Blattodea		
Isoptera		
Mantodea		
Orthoptera		
Dermaptera		
Phasmatodea		
Embioptera		
Psocoptera		
Homoptera		
Heteroptera		
Thysanoptera		
Neuroptera		
Coleoptera		
Diptera	𝍸 𝍸 𝍸 II	*17*
Lepidoptera		
Trichoptera		
Ants		
Other Hymenoptera		
Isopoda		
Amphipoda		
Araneida		
Acari		
Opiliones		
Pseudoscorpiones		
Chilopoda		
Diplopoda		
Symphyla		
	Orders:	***21***
	Total:	***2***

APPENDIX 4: EQUIPMENT

We provide here a comprehensive 'packing list' for a complete one-hectare guide. It includes equipment for each trapping method and general lab set-up. Quantities given here include spares. This list should be tailored to the plot being surveyed, accommodation and number of volunteers on any survey.

Item	Quantity	Trap Type
FIELD:		
Aluminium tags	200	Marking out plot
Ameter and spare batteries	1	Light Trap
Back pack sprayer	1	Canopy Knockdown
Batteries – 12 Volt gel cell	12	Light Trap
Battery chargers	8	Light Trap
Blu-tack	1 pkt	Canopy Knockdown
Bow and arrow (including fishing reel)	1	Canopy Knockdown
Branch clipper / pruner	1	Vegetation Survey
Brushes (1 inch)	15	Bark Spraying / Canopy Knockdown
Catch containers (5 L)	12	Flight Intercept Trap (FIT)
Clinometer	1	Vegetation Survey
Clipboards	3	Vegetation Survey
Compass	4	Marking out plot
Cotton wool	1 pkt	Light Trap / General
Detergent	1 L	FIT / Yellow Pan / General
Dichlorvos™ impregnated pest strips	20	Light Trap / Malaise Trap
Entomological pins, size 0 (38 x 0.40 mm)	2000	Light Trap
Ethanol	40 L	Bark Spraying / Canopy Knockdown / FIT / Leaf Litter Extraction / Pitfall Trap / Yellow Pan
Ethanol carry container (5 L)	2	Canopy Knockdown
Ethyl acetate	100 ml	Light Trap / General
Extension cord	2	Leaf Litter Extraction / Light Trap
Fine gauze (1028 microns)	3	FIT / Pitfall Trap / Yellow Pan
Fishing line	1 roll	Canopy Knockdown / Bark Spraying
FIT rainhoods	14	FIT
Flagging tape – colour 1	50 rolls	Vegetation Survey
Flagging tape – colour 2	4 rolls	Bark Spraying / FIT / Pitfall Trap / Yellow Pan
Flagging tape – colour 3	10 rolls	Marking out plot
Fuel (check backpack sprayer for type)	5 L	Canopy Knockdown
Fuel can (labelled for contents)	1	Canopy Knockdown
Funnel (fuel)	1	Canopy Knockdown
Funnel / sieve	5	FIT / Pitfall trap / Yellow Pan
Gloves – for use in insect spraying	3 pairs	Canopy Knockdown
Gloves – gardening	2 pairs	Leaf litter Extraction
Gloves – latex	1 box	Malaise Trap
Goggles	3	Canopy Knockdown
Half hoops	5	Bark Spraying
Hammer	2	FIT / Malaise Trap / Marking out plot
Hoops	25	Canopy Knockdown
Intercept screens	14	FIT
Killing jars	2	Light Trap
Light bulbs – 40 W (spare)	6	Leaf litter Extraction
Light traps	6	Light Trap
Long handled foceps	2	Light Trap / Malaise
Malaise canopy frames	3	Malaise Trap
Malaise traps	6	Malaise Trap
Mallet	2	Pitfall Trap
Mallet / post rammer	1	Marking out plot
Measuring tape – 50 m	3	Canopy Knockdown / Vegetation survey / Marking out plot
Measuring tape – seamstress or similar	3	Vegetation survey
Metal spike – small	1	Pitfall Trap
Nails (100 mm)	288	Pitfall Trap
Newspaper		Vegetation survey
Nylon cord (spare)	1 roll	FIT / Malaise Trap
Pegs	90	FIT / Malaise Trap / Pitfall Trap
Pencils	3 boxes	All trapping

Item	Quantity	Trap Type
FIELD – continued:		
Permanent markers	5 boxes	Vegetation survey / all trapping
Pins – large map / drawing	40	Bark spraying
Pitfall test tubes, medium wall, 25 x 150 mm	40	Pitfall Trap
Pitfall extraction tool	1	Pitfall Trap
Pitfall trap rainhoods	72	Pitfall Trap
Pitfall trap rainhoods	80	Pitfall Trap
Pitfall tubes (large), 70 ml vials	40	Pitfall Trap
Plant press	1	Vegetation Survey
Plastic container – 1 litre	1	Leaf Litter Extraction
Plastic vials (250 ml)	70	Malaise Trap / Pitfall Trap
Plastic vials (70 ml)	180	Yellow Pan / Canopy Knockdown / Leaf Litter Extraction / FIT / Bark Spraying
Power board (3 plug)	3	Light Trap / Leaf Litter Extraction
Protective suit / coverall	3	Canopy knockdown
Punches	1 set	Marking out plot
PVC pitfall trap sleeves – small	40	Pitfall Trap
PVC pitfall trap sleeves with ring – large	40	Pitfall Trap
Pyrethrin spray cans	10	Bark Spraying
Pyrethrum insecticide fog (Pyfog™)	5 L	Canopy Knockdown
Randomly generated coordinates	100	All trapping
Respirator	3	Canopy Knockdown
Respirator filters	6	Canopy Knockdown
Rope – haul (50 m)	8	Light Trap / Malaise Trap / Canopy Knockdown
Rope – heavy with pulley (50 m)	8	Canopy Knockdown / Malaise / Light Trap
Rope – light nylon (3 m)	3	Light Traps
Rope – guide (30 m)	13	Light Trap / Malaise Trap / Canopy Knockdown
Rope – light nylon (~ 8 m)	20	Canopy Knockdown
Rubber bands	2 boxes	Bark Spraying / Canopy Knockdown
Sash cord (50 m)	2	Shooting canopy lines
Sewing kit	1	Malaise Trap
Stopwatch	3	Bark Spraying
String	Ball	Bark Spraying
Swivels (spare)	20	Bark Spraying / Canopy Knockdown
Takeaway containers painted yellow	15	Yellow Pan
Tape – gaffa	1 roll	Canopy Knockdown
Tissue paper / baking paper	1pkt	Light Trap / Malaise Trap
Tüllgren extraction cabinet to hold 10 funnels	1	Leaf Litter Extraction
UV tubes (spare)	8	Light Trap
Wash bottle	10	Bark Spraying /. Canopy Knockdown / Pitfall Trap
Water carrier 10 L	1	FIT
Water carrier 5 L	2	Yellow Pan / Canopy Knockdown
Waterproof paper (printed with data sheets)	100 sheets	Vegetation Survey
Wing nuts (spare)	10	Light Trap
Wooden stakes	20	Canopy Knockdown
Wooden stakes / 1 m PVC conduit	121	Marking out plot
Yellow paint	400 ml	Yellow Pan
Ziplock / resealable plastic bags, A4	350	Vegetation Survey / Leaf Litter Extraction / Yellow Pan / Pitfall Trap / FIT
LAB / GENERAL:		
Butterfly net	2	General / Lab
Clipboards	5	General / Lab
Communications (radio, EPIRB, mobile phone)		General / Lab
Drying cabinet	1	Moth pinning
Entomological pins, size 3 (38 x 0.53 mm)	500	Moth pinning
Erasers	5	General / Lab
Ethanol (100%)	40 L	General / Lab
Ethanol containers with tap (5 L)	2	General / Lab
Extension leads	2	General / Lab
External memory / zip disk	1	Data Entry
Fine paint brushes	10	General / Lab
First aid kit	2	General / Lab
Fold-up table	2	General / Lab
Forceps – course	10	General / Lab
Forceps – fine	15	General / Lab
Garbage bags	1 pkt	General / Lab
GPS & batteries	1	General / Lab

Item	Quantity	Trap Type
LAB / GENERAL – continued:		
Insect guide books		General / Lab
Insect repellent	1	General / Lab
Laptop	1	General / Lab
Lens tissues	1	General / lab
Light sources	10	General / Lab
Micro moth boxes	5	Moth pinning
Micro pins	20 boxes	Moth pinning
Microscopes – stereo dissecting	6-10	General / Lab
Mylar tape	1 roll	Moth pinning
Naphthalene / pest strip	40	Moth pinning
Note pads	5	General / Lab
Paper – A4	1 ream	General / Lab
Paper towel	2 rolls	General / Lab
Pens (box)	1	General / Lab
Permanent marker pens	10	General / Lab
Permits		General / Lab
Petri dishes (glass)	15	General / Lab
Pipettes	10	General / Lab
Polyporous strip	1 box	Moth pinning
Power boards	3	General / Lab
Printer	1	General / Lab
Printer cartridge (spare)	1	General / Lab
Rubber bands	1 box	General / Lab
Scalpel and blade	10	Moth pinning
Scissors	10	General / Lab
Setting boards	20	Moth pinning
Sharpeners	5	General / Lab
Silica gel		Moth pinning
Small back packs for field work	2	General / Lab
Sorting manual	10	General / Lab
Stapler and staples	1	General / Lab
Store boxes		Moth pinning
Sunscreen	1	General / Lab
Takeaway containers	20	General / Lab
Tape – packing	1	General / Lab
Tarpaulin	1	General / Lab
Toolkit (incl. Wire cutters, screw drivers, spares)	1	General / Lab
Topographic map of site	1	General / Lab
Torches and batteries	1	General / Lab
Unit trays with foam	100	Moth pinning
Utility knife	1	General / Lab
Vegetation keys / botanical guides		Books
Vial stands	10	General / Lab
Vials – glass specimen (10 / 12 ml), box of 100	20	General / Lab
Wash bottles	10	General / Lab
Watch makers glass	10	General / Lab
Whiteboard	1	General / Lab
Whiteboard pens	5	General / Lab
Wire	Assorted	General / Lab
Zip lock bags	Assorted	General / Lab
LABELS / DATA SHEETS:		
Data logging sheets	5 sheets	All trapping
Sample labels	4 sheets	Bark Spraying
Sample labels	2 sheets	Leaf Litter Extraction
Sample labels	2 sheets	Pitfall Trap, small
Sample labels	2 sheets	Pitfall Trap, large
Sample labels	4 sheets	Canopy Knockdown
Sample labels	2 sheets	Malaise Trap
Sample labels	4 sheets	FIT
Sample labels	4 sheets	Light Trap
Sample labels	4 sheets	Yellow Pan
Taxon labels	5 sheets	Bark Spraying
Taxon labels	2 sheets	Leaf Litter Extraction
Taxon labels	3 sheets	Pitfall Trap, small

Item	Quantity	Trap Type
LABELS / DATA SHEETS – continued:		
Taxon labels	3 sheets	Pitfall Trap, large
Taxon labels	6 sheets	Canopy Knockdown
Taxon labels	3 sheets	Malaise Trap
Taxon labels	5 sheets	FIT
Taxon labels	5 sheets	Yellow Pan
Moth taxon labels	10 sheets	Light Trap
Tally sheets (3 per page)	90 sheets	All trapping
Vegetation log sheets (on waterproof paper)	120 sheets	Vegetation Survey

APPENDIX 5: REFERENCES FOR ARTHROPOD IDENTIFICATION

There are literally thousands of manuals available for the identification of arthropods. Some are very general while others refer to small groups only.

In putting together this select bibliography we are inevitably biased towards Australian literature. However we believe even where a book or monograph refers to a particular fauna it can generally be useful if identification is only required to a higher taxonomic level e.g. to family. Of course in some cases no satisfactory specialised works are available, particularly for tropical rainforest faunas.

This list combines books on identification with those on general biology. Space constraints force us to restrict the list to one or two key works per taxon. We have included many of the 'new generation' CD-ROM based, interactive keys for particular taxa.

General

CSIRO (ed.) (1991). *The Insects of Australia: A Textbook for Students and Research Workers.* Melbourne University Press, Carlton.

Undoubtedly the best general book on insects currently available. Enables identification of all Orders to family and/or subfamily.

Zborowski, P. and Storey, R. I. (1995). *A Field Guide to the Insects in Australia.* Reed, Sydney.

A well-illustrated and user-friendly guide to the insect Orders.

Larvae

Stehr, F. W. (ed.) (1987, 1991). *Immature Insects.* 2 Volumes, Kendall/Hunt, Duboque, Iowa.

Collembola

Christiansen, K. and Bellinger, P. (1998). Collembola. *Insects of Hawaii* **15**, 1-445.

Christiansen, K. and Bellinger, P. (1998). *The Collembola of North America, North of the Rio Grande.* Grinnell College, Iowa.

Odonata

Corbet, P. S. (1983). *A Biology of Dragonflies.* Classey, Farringdon.

Watson, J. A. L., Theischinger, G. and Abbey, H. M. (1991). *The Australian Dragonflies.* CSIRO, Canberra.

Isoptera

Hadlington, P. (1987). *Australian Termites and Other Common Timber Pests.* University of New South Wales Press, Sydney.

Krishna, K. and Weesner, F. M. (eds.) (1969, 1970). *Biology of Termites.* 2 Volumes, Academic Press, New York.

Orthopteroid Orders

Clark, J. T. (1974). *Stick and Leaf Insects.* Barry Shurlock, Winchester.

Preston-Mafham, K. (1990). *Grasshoppers and Mantids of the World.* Blandford, London.

Rentz, D. (1996). *Grasshopper Country. The Abundant Orthopteroid Insects of Australia.* University of New South Wales Press, Sydney.

Probably the best general introduction to the Orthoptera, Blattodea, Mantodea and Phasmatodea currently available.

Dermaptera

Steinmann, H. (1990). *World Catalogue of Dermaptera.* Kluwer, Dordrecht.

Plecoptera

Zwick, P. (1980). Plecoptera (Steinfliegen). *Handbuch der Zoologie.* De Gruyter, Berlin.

Psocoptera

Smithers, C. N. (1967). A catalogue of the Psocoptera of the World. *The Australian Zoologist* **14:** 1-145.

Smithers, C.N. (1990). Keys to the families and genera of Psocoptera (Arthropoda, Insecta). *Technical Reports of the Australian Museum* **2:** 1-81.

Hemiptera

Dolling, W. R. (1991). *The Hemiptera.* Oxford University Press, Oxford.

McGavin, G. C. (1993). *Bugs of the World.* Blandford, London.

Thysanoptera

Palmer, J. M., Mound, L. A. and Heaume, D. U. (1989). Thysanoptera. In: *IIE Guides to the Insects of Importance to Man*. CAB International, London.

Moritz, G., Morris, D. and Mound, l. (2001). *ThripsID: Pest Thrips of the World*. CD-ROM. ACIAR, Australia.

Neuroptera

New, T. R. (1984). Plannipennia (Lace-wings). In: *Handbuch der Zoologie.* De Gruyter, Berlin.

Coleoptera

Arnett, R. H., Downie, N. M. and Jaques, H. E. (1980). *How to Know the Beetles.* Second Edition, Wm Brown, Duboque, Iowa.

A guide to the American fauna but also generally useful at the family level.

Lawrence, J. F., Hastings, A. M., Dallwitz, M. J., Paine, T. A. and Zucher, E. J. (1999). *Beetle Larvae of the World*. CD-ROM. CSIRO Publishing, Australia.

Lawrence, J. F., Hastings, A. M., Dallwitz, M. J., Paine, T. A. and Zucher, E. J. (1999). *Beetles of the World*. CD-ROM. CSIRO Publishing, Australia.

These CD-ROM keys are the ultimate guides to identifying beetles to family and often sub-family. An invaluable resource.

Matthews, E. G. (1980 onwards). *A Guide to the Genera of Beetles of South Australia,* Parts 1-8 and on-going. South Australian Museum, Adelaide.

Although of restricted geographical applicability these guides are particularly useful to the beginner because they are entirely based on an innovative picture-key system.

Diptera

Brauns, A. (1954). *Terricole Dipterenlarven.* Munsterschmidt Wissenschaftlicher Verlag, Göttingen.

An old but very useful guide to terrestrial dipterous larvae.

McAlpine, J. F., Peterson, B. V., SHEWELL, G. E., Teskey, H. J., Vockeroth, J. R. and Wood, D. M. (1981-89). *Manual of Nearctic Diptera* (Volumes 1, 2 and 3). Canada Agriculture, Hull, Quebec.

By far the best currently available introduction to the taxonomy and biology of the Diptera treated family by family.

Oldroyd, H. (1964). *The Natural History of Flies.* Weidenfield and Nicholson, London.

Lepidoptera

Common, I. F. B. (1990). *Moths of Australia.* Melbourne University Press.

A superb taxonomic and biological account of relevance far beyond the Australian continent.

Holloway, J. D., Bradley, J. D. and Carter, D. J. (1987). Lepidoptera. In: *CIE Guides to the Insects of Importance to Man*. CAB International, London.

Scoble, M. J. (1992). *The Lepidoptera: Form, Function and Diversity.* Oxford University press, Oxford.

Trichoptera

Neboiss, A. (1986). *Atlas of the Trichoptera of the South West Pacific.* Junnk, Dordrecht.

Hymenoptera – General

Gauld, I. and Bolton, B. (1988). *The Hymenoptera.* Oxford University Press, Oxford.

Hymenoptera – Ants

Bolton, B. (1994). *Identification Guide to the Ant Genera of the World.* Harvard University Press, Cambridge.

Shattuck, S. O. (1999). *Australian Ants: Their Biology and Identification.* CSIRO, Melbourne.

Aranei

Preston-Mafham, R. and Preston-Mafham, K. (1998). *Spiders of the World.* Sterling.

Simon-Brunet, B. (1994). *The Silken Web. A Natural History of Australian Spiders.* Reed, Sydney.

Raven, R. J., Baehr, B. C., and Harvey, M. S. (2002). *Spiders of Australia*. CD-ROM. CSIRO Publishing/ABRS, Australia.

Acari

Evans, G. O. (1992). *Principles of Acarology.* CAB International, Wallingford, UK.

Hunt, G. S., Coloff, M. J., Dallwitz, M. J. and Walter, D. E. (1998). *Oribatid Mites. An Interactive Key to Oribatid Mites of Australia.* CSIRO, Melbourne.

Krantz, G. W. (1978). *A Manual of Acarology.* Oregon State University, Corvallis.

Walter, D. E. and Proctor, H. C. (2001). *Mites in Soil.* CD-ROM. CSIRO Publishing/ABRS, Australia.
Another interactive key for identification.

Pseudoscorpiones

Weygoldt, P. (1969). *The Biology of Pseudoscorpions.* Harvard University Press, Cambridge, Mass.

APPENDIX 6: SAMPLE EXCEL SPREADSHEET

Malaise Traps
Lamington January 1995

	Trap #									
	1	1	1	1						
TAXON	12-Jan	13-Jan	14-Jan	15-Jan	*Total*	*Count*	*mean*	*SE*	*logav*	*logse*
COLLEMBOLA	14	5	1	4	24	4	6	2.7988	0.8451	0.5796
PROTURA	0	0	0	0	0	0	0	0	0	0
DIPLURA	0	0	0	0	0	0	0	0	0	0
ARCHAEOGNATHA	0	0	0	0	0	0	0	0	0	0
THYSANURA	0	0	0	0	0	0	0	0	0	0
EPHEMEROPTERA	0	0	0	0	0	0	0	0	0	0
ODONATA	0	0	0	0	0	0	0	0	0	0
PLECOPTERA	0	0	0	0	0	0	0	0	0	0
BLATTODEA	0	0	0	0	0	0	0	0	0	0
ISOPTERA	0	0	0	0	0	0	0	0	0	0
MANTODEA	0	0	0	0	0	0	0	0	0	0
ORTHOPTERA	0	0	0	2	2	1	0.5	0.5000	0.1761	0.1761
DERMAPTERA	0	0	0	0	0	0	0	0	0	0
PHASMATODEA	0	0	0	0	0	0	0	0	0	0
EMBIOPTERA	0	0	0	0	0	0	0	0	0	0
PSOCOPTERA	0	0	0	0	0	0	0	0	0	0
HOMOPTERA	2	2	6	3	13	4	3.25	0.9465	0.6284	0.2893
HETEROPTERA	0	0	0	0	0	0	0	0	0	0
HEMIPTERA	2	2	6	3	13	4	3.25	0.9465	0.6284	0.2893
THYSANOPTERA	0	0	0	0	0	0	0	0	0	0
NEUROPTERA	0	0	0	0	0	0	0	0	0	0
NEUROPTERA	0	0	0	0	0	0	0	0	0	0
COLEOPTERA	1	4	4	5	14	4	3.5	0.8660	0.6532	0.2709
DIPTERA	179	143	125	227	674	4	168.5	22.5000	2.2292	1.3711
LEPIDOPTERA	3	1	8	4	16	4	4	1.4720	0.6990	0.3930
ANTS	1	1	2	0	4	3	1	0.4082	0.3010	0.1487
OTHER HYMENOPTERA	14	6	6	10	36	4	9	1.9149	1.0000	0.4646
ISOPODA	0	0	0	0	0	0	0	0	0	0
AMPHIPODA	0	0	0	0	0	0	0	0	0	0
SPIDERS	1	0	0	2	3	2	0.75	0.4787	0.2430	0.1699
MITES AND TICKS	0	0	0	0	0	0	0	0	0	0
OPILONIDS	0	0	0	0	0	0	0	0	0	0
PSEUDOSCORPIO	0	0	0	0	0	0	0	0	0	0
CHILOPODA	0	0	0	0	0	0	0	0	0	0
DIPLOPODA	0	0	0	0	0	0	0	0	0	0
SYMPHYLA	0	0	0	0	0	0	0	0	0	0
TOTALS	217	164	158	260						

APPENDIX 7: MASTER DATA SHEET / LOG OF SAMPLES

This record helps ensure all samples are completed and collated centrally. In the example below, the first two days of sampling are complete.

Lamington January 2004
Kitching 1 hectare survey

Sample	Sample	Day	Date Collected	Issued to	Sorted	Comments
Leaf Litter	1	1	1/01/2004	KH	✓	
Leaf Litter	2	1	1/01/2004	ML	✓	
Leaf Litter	3	1	1/01/2004	RLK	✓	
Leaf Litter	4	1	1/01/2004	SB	✓	
Leaf Litter	5	1	1/01/2004	PG	✓	
Leaf Litter	6	1	1/01/2004	RLK	✓	
Leaf Litter	7	1	1/01/2004	ML	✓	
Leaf Litter	8	1	1/01/2004	SB	✓	
Leaf Litter	9	1	1/01/2004	PG	✓	
Leaf Litter	10	1	1/01/2004	KH		
Pitfall small	1	1	1/01/2004	GV	✓	
Pitfall small	2	1	1/01/2004	GV		
Pitfall small	3	1	1/01/2004	PG	✓	
Pitfall small	4	1	1/01/2004	SB		
Pitfall small	1	1	2/01/2004	ML		
Pitfall small	2	2	2/01/2004	RLK		
Pitfall small	3	2	2/01/2004	PG		
Pitfall small	4	2	2/01/2004			
Pitfall small	1	3				
Pitfall small	2	3				
Pitfall small	3	3				
Pitfall small	4	3				
Pitfall small	1	4				
Pitfall small	2	4				
Pitfall small	3	4				
Pitfall small	4	4				
Pitfall large	1	1	1/01/2004			
Pitfall large	2	1	1/01/2004			
Pitfall large	3	1	1/01/2004			
Pitfall large	4	1	1/01/2004			
Pitfall large	1	2	2/01/2004			
Pitfall large	2	2	2/01/2004			
Pitfall large	3	2	2/01/2004			
Pitfall large	4	2	2/01/2004			
Pitfall large	1	3				
Pitfall large	2	3				
Pitfall large	3	3				
Pitfall large	4	3				

COLOUR PLATES

Plate 1 (*left*)**:** Cat's cradle and hoops. (Photo: Guy Vickerman)

Plate 2(a) (*below left*)**:** Pitfall trap spike, sleeve and tube for small traps. (Photo: Sarah Boulter)

Plate 2(b) (*below right*)**:** Pitfall trap spike, sleeve and tube for large traps. (Photo: Sarah Boulter)

Plate 3: Fogging the canopy.
(Photo: Brett Taylor)

Plate 4: Bark spraying.
(Photo: Brett Taylor)

Plate 5: Yellow pan equipment. (Photo: Guy Vickerman)

Plate 6: Ground malaise trap. (Photo: Sarah Boulter)

Plate 7: Malaise trap frame.
(Photo: Roger Kitching)

Plate 8: Modified light trap.
(Photo: Sarah Boulter)