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SACEP/NORAD Publication Series on Biodiversity in South Asia No. 1

An Assessment Manual
For Faunal Biodiversity in South Asia

J. R. B. Alfred
R. K. Varshney
A. K. Ghosh

Zoological Survey of India

Published by:
South Asia Co-operative Environment Programme (SACEP)
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PREFACE

The human population density in South Asian countries is higher than any other continent in the world. Consequently pressures on areas of natural habitats of fauna and flora are especially severe. On the initiative of the South Asia Co-operative Environment Program (SACEP), The Zoological Survey of India (ZSI) took up a short term project on extending the training facilities to the member countries, in the study of (a) Biodiversity, (b) Ecological Impact Assessment and (c) Systematics of fauna. The Ministry of Environment & Forests, Govt. of India approved the proposal in February 1995. A "Letter of Agreement" was signed by both SACEP and ZSI in the same month, wherein it was clarified that the implementation of the project titled '*Assessment of Faunal Biodiversity in the Countries of South Asia*' will be funded by the Norwegian Agency for Development Co-operation (NORAD) through SACEP.

Accordingly, an eight-day Workshop was held at Calcutta, in the Ramakrishna Mission Institute of Culture, from the 12th to 19th June 1995. The following Govt. Representatives (National Co-ordinators) attended:

BANGLADESH	1.	Dr. Niaz Ahmad Siddiqi Divisional Officer Bangladesh Forest Research Institute **Chittagong.**
BHUTAN	2.	Mr. Tshewang R. Wangchuk Nature Conservation Section Forest Services Division Ministry of Agriculture **Thimphu.**
INDIA	3.	Dr. A.K.Ghosh Director Zoological Survey of India Prani Vigyan Bhawan New Alipore **Calcutta.**
MALDIVES	4.	Mr.Maizan Hassan Maniku Director Fisheries Research & Development Ministry of Fisheries & Agriculture **Male.**
PAKISTAN	5.	Mr. Z. B. Mirza Naturalist 229-B, Street No. 4, F-10/3 **Islamabad.**

SRI LANKA

6. Mr. W. A. Jayasinghe
Additional Secretary
Highways Division
Ministry of Health,
Highways & Social Services
Sethsiripaya
Batttaramulla.

Besides the above participants, a representative of the International Union for Conservation of Nature & Natural Resources (IUCN) also attended as a Resource person, namely:

7. Dr. Nirmalee Pallewatte
Department of Zoology
University of Colombo
Thurstan Road
Colombo

The Govt. of India nominated two National Experts-cum-Consultants, who in effect organised the Workshop and acted as Resource persons, namely:

8. Dr. J. R. B. Alfred
Additional Director
Zoological Survey of India
Prani Vigyan Bhawan
New Alipore
Calcutta.

9. Dr. R. K. Varshney
Additional Director
Zoological Survey of India
Prani Vigyan Bhawan
New Alipore
Calcutta.

The SACEP was represented by the following two officials :

10. Mr. Hussain Shihab
Director
South Asia Co-operative Environment Programme
No. 10 Anderson Road
Colombo 5.

11. Mr. Prasantha Dias Abeygunawardene
Deputy Director Programmes
South Asia Co-operative Environment Program
No. 10 Anderson Road
Colombo 5.

The Workshop was inaugurated by Shri Kalyan Biswas, IAS, Principal Secretary, Department of Environment, Govt. of West Bengal and the function was covered by the Calcutta Press and Doordarshan. The scientific sessions were addressed by 23 senior scientists of the ZSI, besides four experts from the Department of Science & Technology and Department of Forests, Govt. of West Bengal and from Central Inland Capture Fisheries Research Station (ICAR).

As the National Co-ordinator from Nepal could not attend the Workshop of June 1995, on the initiative of SACEP a Satellite Session for Nepal was again organised in ZSI during 16-20 October 1995. It was attended by:

NEPAL	12.	Mr. Bodh Raj Subedi Asstt. Conservation Officer Department of National Parks & Wildlife Conservation, Chitwan National Park **Nepal.**

Subsequently, Prof. Sarath Kotagama has replaced Mr. W. A. Jayasinghe as the National Co-ordinator for Sri Lanka.

It is now hoped that the country Reports on their Faunal Biodiversity will soon be ready for all countries of South Asia and the next meeting due at Male (Maldives) will review the progress made in this regard.

South Asia being one of the richest areas in faunal Biodiversity in the world, it is in fitness of things that proper faunal inventories are made for all the ecosystems in this region. It is hoped that this Manual for assessment will serve the purpose and may prove a baseline document for the future.

We thank the SACEP, the NORAD, the faculty of the Workshop, the Contributes of this Manual and all others connected with this project for the co-operation.

J.R.B.Alfred
R.K.Varshney
A.K.Ghosh

Calcutta
1st July 1996

PART I

INTRODUCTION

PREPARATION OF COUNTRY PAPER

A. K. GHOSH*

Survey of Primary and Secondary Sources

The assessment of Biodiversity is an important component of strategic planning process for conservation and management. The process of assessment can be a two dimensional exercise, i.e. identifying areas of maximum diversity within distinctive ecosystems and exploration, collection, preservation and identification of species to provide data for species diversity and related information on population, distribution, status and endemicity. But the starting point for preparation of a country report for assessment should be -

(i) Literature search from available information: group-wise, down to the species level, wherever possible (Research papers; books; journals; catalogues; records); and

(ii) Literature search from available information on ecosystem diversity and genetic diversity.

After the literature search is over, all possible collections present in private, institutional -government or academic levels may be examined. For developing countries much of the collection may not be physically present in the country of origin but deposited in musea of a developed country. Hence, identified international depositories of collections for any possible holding from the country under study has to be enquired.

Analysis

The preliminary assessment when completed would reveal:

(i) Areas so far explored - geographically and ecosystem wise and thereby identifying the gaps;

(ii) Species diversity under each major group, their status and endemic features;

(iii) Expertise capability available for bio-systematic work within country - level;

(iv) Sources of major collections; and

(v) Major literature sources - which can yield a bibliographic index.

The exercise would also help to assess in -

(i) Identifying the components important for conservation and sustainable use;

(ii) The thrust area for future monitoring; and

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* Former Director, Zoological Survey of India, Calcutta.

(iii) The major elements of threat for biodiversity conservation.

In a rapid assessment procedure the Country Paper can only be confined to the basic features listed above. A future in-depth study, while following the identified gaps and future action plan can lead to -

(i) Determination of economic values of biological and genetic resources - based on current economic return, potential use pattern that may emerge on current R & D activities; and

(ii) Determining economic implication of conservation and sustainable use.

Country Paper and Action Plan

The aim of any Country Paper is to assess the present status of available knowledge, material and expertise and to identify the gaps for future action. As such the rapid assessment procedure should lead to a National Action Plan on a realistic assessment.

The plan of action must invariably include -

(i) Intensive studies on maximum diversity areas;

(ii) Building up of a central referral collection at the national level;

(iii) Setting up of a data centre for storage, retrieval and information networking;

(iv) Capacity building up of expertise on a realistic time frame; and

(v) Building up of infrastructure with appropriate security measures for conserving material in *ex-situ* condition.

The signatories to the Convention on Biological Diversity are obliged to report to the Conference of Parties on national measures taken (Article 26). As such much of the recommended procedure listed earlier would assist the country in the process of reporting specially with regard to Articles 6,7,8,9,10,11,12,13,14 of the Convention (IUCN, 1994).

Regional Sources

The faunal biodiversity, as such has to be assessed at (i) Species level, (ii) Ecosystem level, and (iii) Genetic level.

While secondary data source can yield information on (i) and (ii), most of the developing countries still lack sufficient research and data on genetic diversity. Most of the genetic data, wherever available, are related to domesticated species; these data also need serious documentation and assessment in view of potential economic implications. Likewise, data on fish genetic resources, if available, should form a part of the exercise under rapid assessment procedure.

While United Nations Environment Program's has set out 20 guiding principles to assist countries in planning their country study, the stress remains on to "concentrate on readily available data, rather than attempting to achieve comprehensive coverage through new research".

In many countries, much of the data may be restricted to some selected group of taxa, or area or ecosystem but the report in such cases should take into account of such information; in case data from neighbouring country (politically sovereign, but sharing same ecosystem) is available on other groups, cognisance of the same may help to arrive at a more broad based perspective.

Reference

IUCN, 1994. A Guide to the Convention on Biological Diversity.

ECOLOGY AND BIODIVERSITY

J. R. B. ALFRED*

What is Biodiversity?: It is contraction of **biological diversity** - where diversity is a concept which refers to the range of variation on differences among some set of entities, **biological diversity** thus refers to variety within the living world. To-day biodiversity is defined in terms of genes, species and ecosystems - corresponding to three fundamentals and hierarchically - related levels of biological organisation.

Genetic diversity: represents the heritable variation within and between populations of organisms, that resides in the variations in the sequence of the four base pairs, as components of nuclei acids constitute the genetic code.

Species diversity: Since the living world is most widely considered in terms of species, biodiversity is very commonly used as a synonym of **species diversity**, in particular of **species richness** which is the number of species in a site or habitat.

Ecosystem diversity: The quantitative assessment of diversity at the ecosystem, habitat or community level remains problematic. There is very little knowledge on a unique definition and classification of ecosystems at the global level - and they become further complicated in that they include a biotic component. To know more about ecosystems we should go back to that subject called Ecology.

The word ecology itself (from the Greek word **oekas**, meaning home, habitat) was coined in 1869 by the German scientist Ernst Hackle - who used it to define the science which studies the relationship between an organism and its environment.

The analytical or reductionist sciences on the one hand set out to dissect and dissociate the elements of a structure in order to define and study them. On the other hand the synthetic or holistic sciences (from the Greek word **holos**, meaning whole), of which ecology is the best example - grasps a system in its entirety by studying the interactions between all its elements. These two scientific approaches, both equally important are by nature complementary and this might well be reflected in practice.

Ecology

At the beginning of the present century, was a descriptive study of nature, a sort of natural history, which drew inspiration from the works of explorers and naturalists. Before long, detailed studies were made of the environment, in which a given species live, and other of their symbiotic, and antagonistic relationships with other species. This **auto - ecology** or ecology centred on single species had and still has important applications in the biological control of plant pests, research on disease carriers and prevention of parasitic borne infections.

But each species, even when studied in conjunction with those that influence it directly, is only a tiny instance among the thousands of plant, animal and microbial species which inhabit a given area - a forest, a pond or a beach. This study developed into **synecology** - viz. ecology of communities of species. Basic concepts like **food - chain**, and the **pyramid of numbers** in which the number of individuals decreases progressively from plants at the base to herbivores and predatory animals at the

* Zoological Survey of India, Calcutta.

summit. Then came mathematical laws governing the population dynamics of interacting groups of species (these proved useful in aquatic ecology) helped solve problems to sea fishing and understand the phenomenon of insect invasions.

But very early in the 1950s it was recognised that ecology in spite of its wider ranging disciplines had adopted a strictly scientific approach - but it also revealed that this science was dissipating its effort in too many directions - and most of all it lacked a basic study unit as was available to all other scientific disciplines.

This basic unit or entity would have to be precisely defined in space and time including all the organisms inhabiting it as well as the physical conditions of elements and soil and most of all should include all interactions between different organisms and between the organisms and the physical conditions. Such a unit was to be the **ecosystem**, first proposed in 1935 by Arthur George Tansley and it was Raymond Lindemann who pioneered the conceptual and methodological basis for studying these highly complex systems - the energy flow and nutritional cycles which pass through all the living and non-living components of the ecosystem.

Ecosystem mapping may either take into account the attempt to produce a realistic, contemporary map of land-cover types, or may create potential vegetation maps from an analysis of both climate and other environmental variables.

For example, we have 4 major global classification systems:

1. The classification of Bio-geographical Realms of the world.
2. The Eco-regions of the Continents.
3. The Major World Ecosystems.
4. The Holdridge Life Zone Classification.

Bio-geographical Realms of the World: Depicts the terrestrial bio-geographical realms of the world and was produced for IUCN (Udvardy, 1975). It provides a generalised framework to represent the distribution of bio-geographical region, biotic provinces and biomes. Vegetation and forest maps were utilised to produce the map categories.

Eco-regions of the continents: Showing the distribution of ecosystems at the regional scale across the globe based upon existing climatic and vegetation data (Bailey, 1989 a, b). Here the three levels representing the ecosystems are domains, divisions and provinces. These are obtained by defining aggregates of ecosystems into large biomas categories.

The Major World Ecosystems: This is after Olson et al., 1983 and represents large areas within which local ecosystems are present.

The Holdridge Life Zone Classification: This is a scheme for identifying undisturbed vegetation based generally on the effects of temperature, rainfall, and evapo-transpiration (Holdridge, 1967). It is based on the annual climatic variables, bio-temperature (mean positive temperatures), total annual precipitation and evaporation (defined as a function of bio-temperatures). The Life Zones are delineated by hexagons derived from a triangular graph of these three variables.

Indian Region

According to world biographic classification, India represents two of the major realms (the Palaearctic and Indo-Malayan) and three biomes (Tropical Humid Forest, Tropical Dry/Deciduous Forests, and Warm Deserts/Semi deserts). These can also be seen as 12 bio-geographical regions. The Wildlife Institute of India, recently has prepared a slightly modified classification which divides the country into 10 bio-geographic regions (Table - 1): Trans-Himalayan, Himalayan, Indian Desert, Semi-Arid, Western Ghats, Deacan Peninsula, Gangetic Plains, north-east India, Islands and Coasts. India is one of the 12 identified mega-diversity centres. It also has two of the 18 identified hotspots - the Eastern Himalaya and the Western Ghats. India's marine habitat covers 7,500 km of coastline extending over 200 nautical miles in the offshore forming the Exclusive Economic Zone (EEZ).

IUCN, has developed a system of classification for different types of protected areas based upon the management objectives. The system has 10 different classes of protected area: I. Scientific Reserve, II. National Park, III. Natural Monument, IV. Managed Nature Reserve, V. Protected Landscape, VI. Resource Reserve, VII. Natural Biotic Reserve, VIII. Multiple Use Area and two international designations: IX. Biosphere Reserves and X. World Heritage Sites.

The two international designations of protected areas - *World Heritage Sites* - The Convention concerning the Protection of the World Cultural, and *Natural Heritage* was adopted in Paris in 1972 and came into force in December 1975. Under the World Heritage Sites, five natural sites of India have been declared as World Heritage Sites.

Biosphere Reserve - Not necessarily exclusively designated to protect unique areas or important wetlands but for a range of objectives which include research, monitoring training and demonstration, as well as conservation. In most cases the human components is vital to the functioning of the biosphere reserve, which does not necessarily hold for to others - though in India it is now well recognised that this is pertinent for any protected area.

In order to ensure the unhindered evolution of micro-organisms, plants and animals in their totality and as part of the natural ecosystems, the Government of India has designed eight Biosphere Reserves, based on the comprehensive concept of conservation, evolved by the UNESCO's Man and Biosphere (MAB) Programme initiated in 1971. In addition, there are 80 National Parks and 441 Wildlife sanctuaries covering 148,700-sq. km. area. Project Tiger, which was launched in 1973 is spread over 23 Tiger Reserves covering a total area of 33,046 sq. km. Conservation of Biodiversity outside the network of protected area, including sensitive/rich ecosystem like coral reefs, mangroves, wetlands - have enabled identify 21 wetlands, 15 mangrove and 4 coral referees. Six significant wetlands of India have been declared as "Ramsar site" under the Ramsar Convention, 1971.

Hence, Ecosystem diversity enables the understanding of:

1. The local and geographic distribution of organisms.
2. Regional variations in the abundance of organisms.
3. Temporal changes in the occurrence, abundance and activities of organisms.
4. The interrelations between organisms as populations and communities.
5. The structural adaptations and functional adjustments of organisms to their physical environment.

6. The behaviour of organisms under natural conditions/

7. The evolutionary development of all these interrelations.

8. The biological productivity of nature and how this may best serve mankind.

Selected References

Bailey, R .G. 1989a. *Eco-regions of the Continents.* U.S. Deputy of Agriculture, Forest Service, Washington D.C.

Bailey, R. G. 1989b. Explanatory supplement to Eco-regions map of the Continents. *Environmental Cons.,* 16(4): 307-309.

Holdridge, L. R. 1967. *Life Zone Ecology.* Tropical Science Centre, San Jose: 206pp.

Olson, J. S. Watt, J. A. & Allison, L. J. 1983. *Carbon in Live Vegetation of Major World Ecosystems.* Oak Ridge National Laboratory, Oak Ridge.

Udvardy, M. D. F. 1975. *A Classification of the Bio-geographical Provinces of the World.* IUCN Occ. Paper, No. 18.

Table - 1. **Biogeographic Classification of India**

	Biogeographic Zone		*Biotic Province*
1.	Trans-Himalayan	1A.	Tibetan
2.	Himalayan	2A.	North Wet Himalaya
		2B.	West Himalaya
		2C.	Central Himalaya
		2D.	East Himalaya
3.	Desert	3A.	Kutch
		3B.	Thar
4.	Semi-Arid	4A.	Punjab
		4B.	Gujarat-Rajwara
5.	Western Ghats	5A.	Malabar Coast
		5B.	Western Ghat Mountains
6.	Deccan Peninsula	6A.	Deccan Plateau South
		6B.	Central Plateau
		6C.	Eastern Plateau
		6D.	Chota Nagpur
		6E.	Central Highlands
7.	Gangetic Plain	7A.	Upper Gangetic Plain
		7B.	Lower Gangetic Plain
8.	North-East India	8A.	Brahmputra Valley
		8B.	Assam Hills

9.	Islands	9A.	Andaman Islands
		9B.	Nicobar Islands
10.	Coats	10A.	West Coast
		10B.	East Coast

SPECIES BIODIVERSITY

R. K. VARSHNEY*

Biodiversity is defined as the variety and variability among living organisms and the ecological complexes in which they occur. These are simply approached from three angles: i) Ecological biodiversity, ii) Species biodiversity, and iii) Genetical biodiversity.

The Species biodiversity among organisms is most important out of the three, as it shows the richness of various kinds of plants and animals in a given ecosystem, thus exposing the health of that ecological niche to sustain and maintain the quality of life among the inhabitants and the inter-relationships among them.

Species as a Unit

To define an animal species, the prevalent concepts are as follows:

Morphological Concept of Species (Phonetic): Species is a taxon composed of groups of natural populations having a high degree of *similarity* in their *phenotype*.

Biological Concept of Species (Genetic): Species is a taxon composed of groups of *Interbreeding* natural populations, which have similar *genotype* but are *reproductively isolated* from other such groups of natural populations.

Nomenclatural Concept of Species: A category below the Genus-group; the fundamental *Unit of classification*; and which is objectively defined by its *Type specimen.*

The species is a qualitative unit. It will be a worthwhile exercise to assess the biodiversity by collecting data on various animal groups occurring in the countries of South Asia and then to analyse and inventories the species for the respective country and the region.

Taxonomy

To determine and document animal species, taxonomical studies are necessary. What is Taxonomy? Taxonomy (Taxis = arrangement, Nomos - law) is the theory and practice of classifying organisms or the study of classification including its bases, principles, procedures and rules.

Some well-known definitions are as follows:

Taxon: A taxonomic group that is sufficiently distinct to be distinguished by name and to be ranked in a definite category.

Alpha taxonomy: The level of taxonomy concerned with the characterisation and naming of species. (Analytical stage, to characterise differences between taxa).

Beta Taxonomy: The level of taxonomy concerned with the arranging of species into a natural system of lower and higher taxa. (Synthesis of characters for grouping under supraspecific categories).

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* Zoological Survey of India, Calcutta.

Gamma Taxonomy: The level of taxonomy dealing with various biological aspects of taxa; the study of infraspecific populations, speciation and evolutionary trends. (Details of infraspecific categorisation).

Genus: A category for a taxon including one or a group of species, presumably of common phylogenetic origin, which is separated from related similar genera by a decided gap, the size of the gap being in inverse ratio to the size of the genus.

Phylogeny: The study of the history of the lines of evolution in a group of organisms; the origin and evolution of higher taxa.

Sibling species: Pairs of groups of closely related species, which are reproductively isolated but morphologically identical or nearly so.

Variety: An ambiguous term of classical taxonomy for a heterogeneous group or phenomenon, including nongenetic variations of the phenotype, morphs, domestic breeds and geographic races.

Categories of Classification

There are standard categories of the animal classification, well recognised in taxonomy.

A descending chart is presented below showing the major categories. Of these, names of the categories from Super family downwards upto Subspecies are governed by an International Code of Zoological Nomenclature.

Categories of Animal Classification

Kingdom
Phylum
Class
Order

Superfamily
Family
Subfamily
Tribe
Subtribe
Genus
Subgenus
Species
Subspecies
(Geographic Race)

Varieties
(Forms, Morphs,
Breeds, etc.)

Identification

The most important thing is to accurately distinguish between closely related species. The correct identification is necessary for all purposes, including the applied ones. This is facilitated by standard dichotomous *keys*, which are available on many animal groups, to sort out taxa. The diagnostic characters mentioned therein, thought often inconspicuous, are literally the key characters.

Zoological Nomenclature

All animal groups at every category of classification have been provided with distinct scientific names. In these categories, from Superfamily to subspecies level, these names are governed by the International Code of Zoological Nomenclature. This branch of Taxonomy has helped us to deal with the synonyms, homonyms, Law of Priority and rules for formation of new names. Wide practice of these rules has stabilised scientific names to a great extent. Binomial and trinomial nomenclature of species have become familiar in diverse animal groups and the style of citing these with author and year has been uniformly standardised. This may be shown as an example for some hypothetical species in the Box.

Documentation of a Species name

Species name [Binomial]
Rasa indica
Rasa punctatus

With Author
Rasa indica Linnaeus

With Year
Rasa indica Linnaeus, 1762

With Changed Genus
Navarasa indica (Linnaeus)

With Year
Navarasas indica (Linnaeus, 1762)

When Subgenus is known [Binomial]
Rasa (Rasa) indica
Rasa (Terias) punctatus

When Subspecies is known [Trinomial]
Rasa indica indica
Rasa indica bengalensis

With Author
Rasa indica bengalensis Crowe

With Changed Genus
Navarasa indica bengalensis (Crowe)

When Subgenus is known [Trinomial]
Rasa (Rasa) indica bengalensis

With Author
Rasa (Rasa) indica bengalensis Crowe

With Changed Genus
Navarasas (Terias) punctatus punctatus (Stoll)

With Year
Navarasa (Terias) punctatus punctatus (Stoll, 1802)

Current Estimates of Species

To prepare a Country Report, it is vital to estimate as to how many species are already recorded from there (Table - 1-3). This is an exhaustive task but a very important one. Literature has to be surveyed, monographs and review to be consulted. *Zoological Records* volumes to be scanned and data culled from all primary (i.e. unpublished Museum catalogues, Registers, cards) and secondary (i.e. published) sources.

From time to time various workers have attempted to estimate the number of species of an animal group occurring in a country or part of a country. These figures keep on changing due to many factors, like addition of new species and new records of known species, as well as dropping of synonyms etc. Thus, no final count can be made for all time to come. It will be better to follow the figures provided by an established worker after a thorough search in his or her most recent work.

Table - 1. **Estimated numbers of species in all animals groups**

	Kingdom/Phyla	*No. of species*		*Indian Share in world %*
		World	*India*	
	PROTISTA			
1.	Sarcomastigophora	18,450	1,080	5.85
2.	Ciliophora	7,200	600	8.33
3.	Apicoriplexa	4,550	750	16.48
4.	Microspora	550	20	3.64
5.	Myxozoa	500	125	25.00
6.	Ascetosphora	30	2	6.67
7.	Labyrinthomorpha	10	-	
	Total	**31,290**	**2,577**	**8.24**
	ANIMALIA			
1.	Porifera	5,100	516	10.12
2.	Cnidaria	10,000	850	8.5
3.	Ctenophora	100	10	10.00
4.	Platyhelminthes	17,500	1,650	9.42
5.	Nemertinea	600	-	-
6.	Mesozoa	50	-	-

7.	Gastrotricha	500	88	17.60
8.	Rotifera	2,550	310	12.15
9.	Kinorhyncha	99	10	10.10
10.	Nematoda	25,000	2,850	11.40
11.	Nematomorpha	250	-	-
12.	Acanthocephala	800	110	13.75
13.	Gnathostomulida	100	-	-
14.	Annelida	12,700	840	6.61
15.	Mollusca	66,535	5,050	7.59
16.	Bryozoa (Ectoprocta)	4,000	170	4.25
17.	Entoprocta	60	10	16.67
18.	Phoronida	11	3	27.27
19.	Brachiopoda	300	3	1.00
20.	Arthropoda	9,83,744*	60,383	6.13
21.	Pogonophora	80	-	-
22.	Onychophora	100	1	1.00
23.	Sipuncula	202	38	18.81
24.	Echiura	127	33	25.98
25.	Priapulida	8	-	-
26.	Pentastomida	70	-	-
27.	Tardigrada	370	30	8.10
28.	Chaetognatha	100	30	30.00
29.	Echinodermata	6,226	765	12.28
30.	Hemichordata	118	12	10.16
31.	Chordata			
	i) Protochordata	2,173	116	5.34
	ii) Pisces	21,723	2,546	11.72
	iii) Amphibia	5,145	204	3.96
	iv) Reptilia	5,680	446	7.85
	v) Aves	9,672	1,228	12.69
	vi) Mammalia	4,629**	372	8.03
	Total	**11,86,422**	**78,674**	**6.63**
	(Protista+Animalia)	**12,17,712**	**81,251**	**6.67**

Note : The Phylum Loricifera is not included under the Kingdom Animalia since no data is available for the same. A dash (-) means the information is not available and not that the group is absent in India.

* Further break-up is shown in Table 2.

** According to the latest publication: Mammal Species of the World (2nd Edition) (Eds. Wilson & Reeder, 1993).

Table -2. **Estimated numbers of species of Phylum Arthropod**

		World	*India*	*% In India*
Class	1. Crustacea	38,840	3,220	8.3
	2. Insecta	8,53,080*	53,430**	6.26
	3. Arachnida	80,240	3,449	4.3
	Acari	36,800	2,025	
	Araneae	35,000	1,015	
	Scorpions	1,500	102	
	Pseudoscorpiones	2,300	100	
	Solifuge	900	15	
	Palpigradi	85	25	
	Uropygi	130	?	
	Ricinulei	25	?	
	Opiliones	3,500	167	
		80,240	**3,449**	
	4. Pycnogonida	600	16	2.67
	5. Pauropoda	360	?	?
	6. Chilopoda	3,000	100	3.33
	7. Diplopoda	7,500	162	2.16
	8. Symphyla	120	4	3.33
	9. Merostomata	4	2	50.00
	TOTAL	**9,83,744**	**60,383**	**6.13**

* A total of 9,50,000 (in "Global Biodiversity" Ed. Brian Groombridge, 1992).

** There are references, which give a figure of 50,000 to one lack species in India.

Table - 3. **Estimated numbers of Species of Class Insecta**

Insect Order (arranged systematically)	*No. of Families in India*	*No. of Spp. in India*	*No. of Spp. in World*	*% of Spp. in India.*
APTERA				
Tysanura	3	27	1254	2.15
Diplura	3	18	357	5.04
Protura	3	20	260	7.69
Collembola	8	205	5005	4.09
HEMIMETABOLA				
Ephemeroptera	12	106	2200	4.81
Odonata	19	494	5500	8.98
Plecoptera	7	113	2100	5.38
Orthoptera	30	900	19500	4.61
Phasmida	6	60	2500	2.40
Dermaptera	7	320	1800	17.77
Embioptera	2	33	200	16.50
Blattaria	14	156	4200	3.71
Mantodea	6	162	2210	7.33
Isoptera	7	300	2000	15.00
Psocoptera	12	85	2500	3.40
Phthiraptera	8	400	3000	13.33
Hemiptera	49	6500	80000	8.12
Thysanoptera	5	691	6200	11.14
HOLOMETABOLA				
Neuroptera	13	315	5000	6.30
Coleoptera	103+	15000+	350000+	4.28
Strepsiptera	3	8	300	2.66
Macoptera	2	15	350	4.28
Siphonaptera	8	52	2000	2.60
Diptera	87	6093	100000+	6.09
Lepidoptera	89	13000+	142500	9.12
Trichoptera	18	812	7000	11.60
Hymenoptera	65	5565	111266	5.00
TOTAL	**589**	**51450**	**859202**	**5.98**

PART II

METHODS AND APPROACHES TO DATA COLLECTION

MAMMALS

S. CHAKRABORTY*

Introduction

By virtue of great adaptability, about 4230 mammalian species are represented in almost all the ecological zones and habitat of this plane. They occur in deep sea to alpine zone, hot sand dunes to snow and cold desert, dense tropical rain forest to grass land, crop fields to our houses. Some species are restricted to a particular habitat, while many are found in different habitats. For example, in the coniferous forest of Pakistan and northern India, flying squirrels, *Petaurista* live and find their food in the tree top and they never descend to the ground, while a vole, *Hyperacrius wynnei* is a burrower and root feeder of the same forest and always lives under the ground. Thus within the same ecosystem, different species may be restricted to different zones.

Collection

For the collection of the mammalian specimens, arrangement of collection, preservation, and labelling equipment's, such as traps, mist nets, firearms, forceps, axe, knife, hammer, plaster of Paris, hand lens, scissors, trays, aluminium poles, ropes, scales, chemicals (ethyl alcohol, formaldehyde, chloroform, alum, arsenic soap etc.) pencil, scale, labelling paper, silk thread, collection vials, plastic bags, head lights, torches, battery, haversacks, hunting shoes, camera, compass, altimeter, field note book etc. should be made in the laboratory. Both random or systematic selection of grids has its own merits and demerits. However, attempts should be made to include all the habitats of the survey area within the selected grids.

Four methods are usually adopted for the collection of mammalian species:

A. *Trapping*: This is the most traditional technique for capturing small (head and body length less than 30 cm.) and medium-sized (head and body length 30-100 cm.) species. Some species are bold, others are shy, neophobic and avoid traps. Again, there are many fossorial and arboreal species, which not appear on the ground. There are also differences among species in respect of food habit, activity hours and pattern. Thus, traps, baits and trapping period are to be selected according to the target species. However, for random collection a multiple combination should be tried. The traps, used for the collection of mammals, may be broadly classified into two categories, viz. Snap or Break back and Live. These traps are variously designed under different trade names and used for the specific purposes. However, it is economical to manufacture the required types of trap by providing good drawing to the local blacksmith.

Placement of traps is also very important for trap success. For woodland species, it is better to place the traps along natural drift fences, such as, logs or stumps; for grassland species in runways or along natural drift fences; for desert species in bare areas between clumps of vegetation or around dens. Semi aquatic species can be best collected by setting traps at water edge or in dry land inside marshes. For totally fossorial species traps can be pushed into the tunnel mouth after removing the loose topsoil.

* Zoological Survey of India, Calcutta.

Trap stations are, in general, spaced about 10 m. it is often advantageous to set more than one type of trap at each station. Trap line should be marked at the beginning and end to prevent loss.

B. *Netting* : Use of mist net has brought about at revolution in the collection of flying mammals (bats). Only a few species of bat roost in an open area, which may be collected either by shooting or by netting using a large butterfly net attached to a bamboo or aluminium pole. Crevice roosting species can be collected by stretching Japanese mist nets across cave entrances or over the eaves of old deserted building where bat roosts are detected by the presence of faeces, smell or cries. For collection of bats from their foraging places mist net should be placed with the help of bamboo or aluminium poles at the edge of water tanks, natural streams, edge of groves of trees, orchards, and wherever it is noted that bats frequently hunt. It is advisable to visit the nets at intervals of one or two hours rather than to leave the net throughout the night. One should be very careful while removing the bats from the net as they inflict painful bites.

C. *Shooting*: Shooting with firearms is an important method for collection, particularly of large (more than 100 cm.) to medium sized mammals and arboreal species. However, certain amount of precaution are to be taken while hunting with firearms for the safety of the hunter, unwanted intruder and non-target species. While shooting, range of the shot as well as the arms in use should carefully be judged in order to be sure to kill or fatally injure the target animal. With shotguns the number of cartridge should also be taken for the possible changes of revival of the shot specimen. From our practical experiences, it was found that following firearms and ammunition may serve the purpose of collection:

(i) 410 shot gun with cartridges Nos. 4,6,8,9 and dust.
(ii) 12 bore shotgun with L G, S G, dust, 1-9 Nos. cartridges.
(iii) 22 rifle with dust and bullet.

D. *Excavation of burrows*: Capture of live fossorial specimens may be done by excavation of burrows. In this process help of a second person with a net is required.

Under present day Wildlife Acts of different countries, collection or killing of many of the mammalian species are banned. It is also very difficult to observe many of the species in the field. Inventory of these species along with their population and feeding behaviour can be made from the study of their tracks and scats in the field. Several studies have been conducted for the specific identification of tracks and scats. Analysis of scats of carnivorous species often reveals animals which could not be collected by trapping, netting etc. Tracks and scats may be collected in the following ways:

Tracks:

(1) By careful drawing of the natural size drawing of the track. Length of few sample stride or leaps as well as dimension of each individual print should also be noted.
(2) It is sometimes possible to cut around the track with a knife and remove it out intact.
(3) By taking cast of tracks with plaster of Paris.

Scats:

Scats of most of the species have specific features. Collected scats should be kept in original shape by suitable packing in the field. Coating with some glue varnish will help to prevent attack of moths or dermestid beetles.

How many individuals of a species should be collected or what is the ideal sample size? There is no precise answer to this question as so many variables are involved. For the study of geographic variations, ideal sample size will be 20 adult animals consisting of both the sexes and the sample should be collected at about every 160 km. throughout the geographic range of species. For individual, age and sexual variation a sample size of 50-70 specimens will be ideal. However, collection should be made in all the seasons to document the seasonal changes and reproductive data.

Preservation and Fixation

Wet preservation: After killing the specimen and nothing its field data, a solution of 10% formaldehyde or 90% ethyl alcohol is injected in the abdominal cavity of the specimen so as to somewhat distend it. Afterwards abdomen is cut open in the mid ventral line to expose the viscera and the specimen is then kept in 90% ethyl alcohol or 10% formaldehyde for 24 hours. Next the specimen is taken out from preservative, washed in water, semidried and put in 4% formaldehyde or 70% ethyl alcohol.

Dry preservation: The skin and skeleton can be dry preserved. In this process an incision is given in the mid ventral line of the specimen. The entire flesh and skeleton is then taken out through that slit.

On return to the laboratory, preservatives of the wet specimens should be changed. Dry skins should be tanned, rolled or mounted. However, skinning, tanning, rolling and mounting are extremely specialist job. It requires taxidermy.

After final preservation all the specimens should be identified by the specialist and properly documented in the institutional register and catalogues. The specimens and field note book should be placed systematically in insect-dust-damp proof cabinets with proper indexing.

Data Recording and Documentation

While recording data from collected specimens, information on each individual must be maximised, so that each individual may be referred to in future. For the specific identification in future, it is necessary to note some external measurements, like head and body length, ear length, etc., immediately after killing the specimens in the field.

While making collection, a collector should also try to obtain data on the population status of the different species occurring in the area of collection. From direct and indirect observations, the collector can record in his field note book about the population as plenty, common, rare, etc.

Other techniques for the collection of population data and statistical analysis are as follows:

Census data: It involves direct complete count or samples count of an animal population in a specified area in a given time. This method is most effective for diurnal herd of animals, which feed or take shelter in some relatively open area.

Sample plots: Sample plots are useful for estimating the burrowing animals. Here all the burrows of the sample plot are sealed with sand or loose soil. Next day population estimate is made by counting the emergence holes.

Count indices: In this method estimate of animal population is made from the count of animal signs such as pug-marks, scats etc. This is most effective method for the estimate of nocturnal and secretive species like tiger, panther, etc.

Roadside count: This method is most suitable for those species, which forage in a larger area and occasionally cross the forest roads. It involves counting of animals sighted along a particular distance of a road. By this method, census index or relative population of different species in a particular area could be obtained.

C. M. R.method: This method is suitable for species, which can be live-trapped or netted. This involves capturing of a portion of population from a given area and releasing them in the same area after marking each individual. After a certain time, trapping or netting is done with same number of traps or nets. The total number of animals captured is counted along with the number of recapture. Population estimate is made from the following formula:

$$N = nT/t$$

Where

N	=	Population size estimate.
T	=	Number of specimens originally marked.
t	=	Number of marked animals recaptured.
n	=	Total number of animals captured the second time.

A good collection of mammalian specimens from different parts of the South Asian countries are maintained in the Zoological Survey of India, Zoological Survey of Pakistan, Colombo Museum, Bombay Natural History Society, British Museum, Carnage Museum, different Universities and research laboratories. These not only serve the public in education, but also act as an enormous reservoir of data base for studies in biodiversity and its changes through ages. From the study of old museum specimens one may collect the data on the following aspects:

A. Mammal species composition of an area.
B. Infraspecific morphological variation.
C. Taxonomy and Systematics.
D. Anatomy of soft and hard parts.
E. Feeding.
F. Breeding behaviour.
G. Zoo geography.
H. Relative population density.
I. Changes in the level of environmental pollutants, such as heavy metal, organopesticides, etc.

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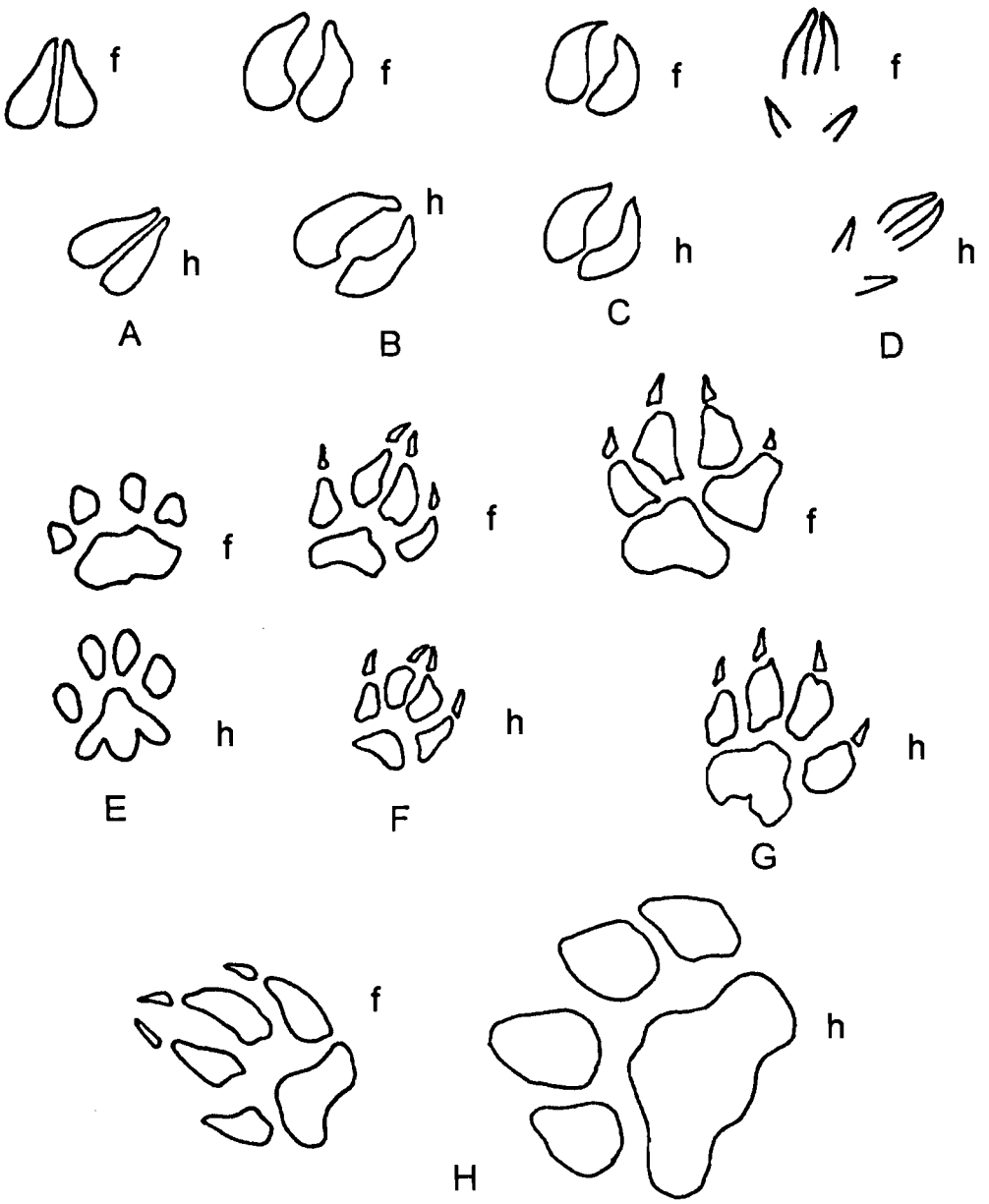

Fig. 1 - **Pugmarks of Mammals**

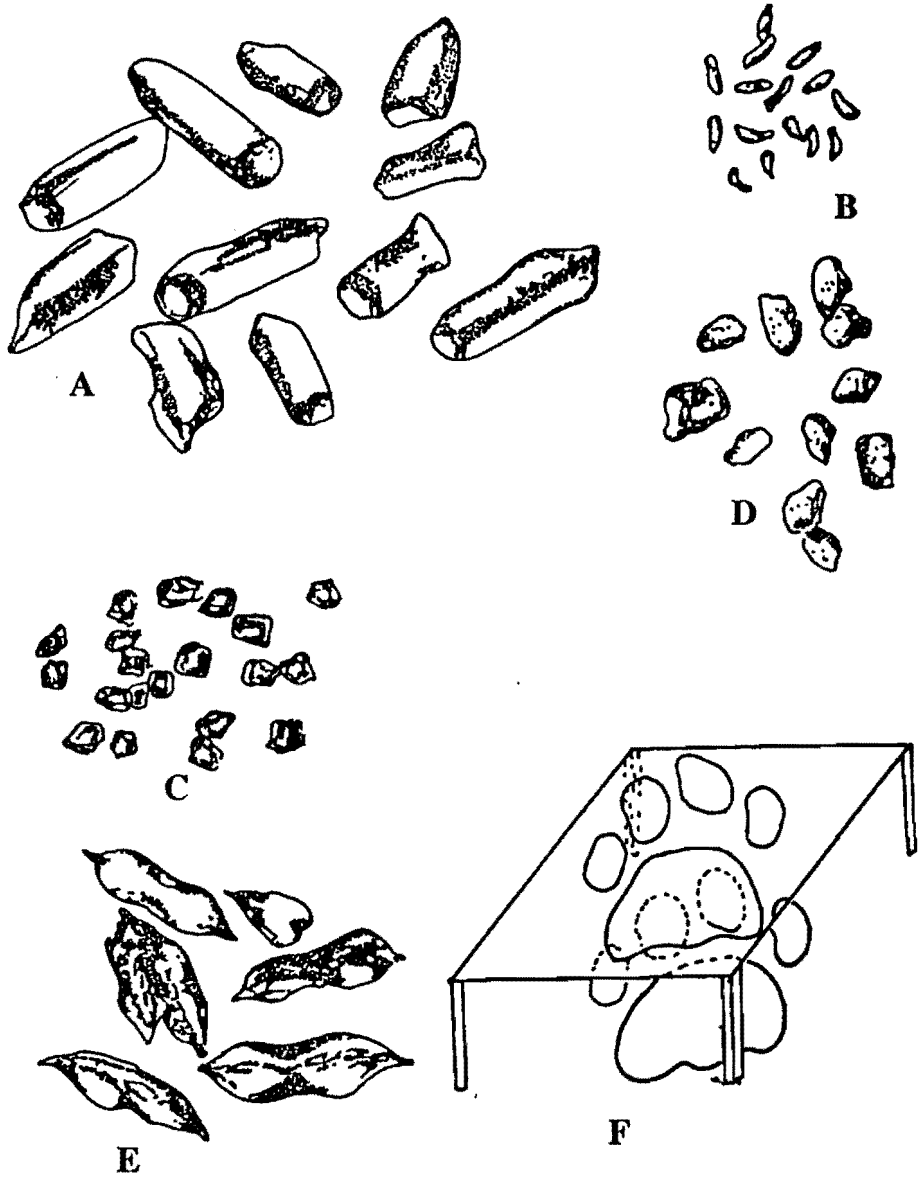

Fig.2 - **Faecal pellets (scats) of some Mammals**

BIRDS

S. S. SAHA*

Introduction

Although birds as a group have spread all over the planet but species are accustomed to live in restricted areas according to its origin and adaptations. Every species is adapted structurally, physiologically and psychologically to certain sets of environmental conditions. Based on the intensity of solar radiation the globe is divided into several bio-climatic or life supporting belts. This succession of belt is also reflected in the latitudinal gains. One degree of latitudinal change (roughly 69 miles) is equivalent to change in 250 ft of altitude. Besides the solar radiation, temperature, geographical location like topography, rocks and soil of the area, climate and other variable factors govern the condition on which the life supporting system depends.

Estimate of Taxa

There are 27 Orders, 171 Families, 8528 species, and over 14,534 subspecies of living birds of the world. Of them 437 species and subspecies have been designated as threatened for their survival, and many more are likely to follow them; some are at the brink of extinction also.

There are 1232 species in 405 Genera, 73 Families, 20 Orders, and over 2094 Forms (Species and Subspecies) recorded from the Indian subcontinent. Of them, about 350 species are migratory. There are 176 Species and 11 genera Endemic to the Indian Subcontinent. Of them, 48 species are in India, 19 in Sri Lanka, 18 in both India and Sri Lanka, 14 in India and Pakistan. Many of the other endemics share their range with neighbouring countries. Three species are supposed to be extinct in recent times, namely, Pink-Headed Duck, Mountain Quail, and the Forest Owlet. At least 24 species are exceedingly rare, and an additional 29 other species are endangered.

Collection

A majority of bird species may be identified by its plumage alone, in fact, every bird species has its own plumage characteristic, but there are many siblings or identical bird species also, only to be ascertained by proper taxonomic studies, i.e., by the help of measurements, comparison with reference collection, analysing field notes etc. Any information on the birds of the area may be a source of valuable data. Sum total of the data will form the basis for assessment of Avian biodiversity.

In the field first hand information may be procured either by recording visuals, i.e. by Bird Watching and documenting with photographic records supplemented by sound recordings; or actually taking specimens sample for reference and studied in the laboratory.

Equipment's

i) For Bird Watching:

A pair of Field Binoculars, and/or Telescopes. 8 x 40 is the most suitable binocular for bird watching.

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A field Note Book.
A field Guide Book for ready recognition of birds in the field.
A Camera with Telephoto lenses of x 300 or more magnification.
Tape Recorder.

ii) For Specimen collecting:

Mist Nets of 1/2", 3/4" or 1" mesh may be used to capture live birds with least damage.
Firearms and ammunitions have been used for ages for procurement of bird specimen (killed). Shotguns in 12 bore, 16 bore, 20 bore or '410 bore with shots numbering BB, 1 to 12 may be used for collecting birds of different sizes. Even '22 bore sporting rifle also may be used to collect some large birds.

Traps and professional Bird Trappers may be engaged for procurement of bird specimen live or dead. But for trapping, netting or hunting birds an appropriate Permit should be obtained.

Specimens procured through any of the above means need to be prepared in the field to make it a sample specimen for future reference and preservation. For preparation a highly skilled Taxidermist is required. The data (external) are noted from the specimen and gonads are checked when the bird is skinned for sex and age determination. Sex of birds can be determined positively by examining the gonads. The bird is then 'skinned; i.e. the entire skin with features, limbs, tail, beak with skull are retained discarding the flesh and bone, and soft tissues (including brain and the eyes). After skinning, the innerside of the skin is applied with a preparation of chemicals for preservation and poisoning so that moths will not readily damage the skin which is otherwise very delicate and readily damaged. The skin is then stuffed by cotton wool aiming to replace the discarded part of the body, and given a shape as the dead bird is lying on its back. Thus the specimen is curated.

To the curated specimen a 'label' is tied containing vital data like the *Locality* (from where the specimen has been collected; exact location preferably with co-ordinates and altitudes, district and state name); *Date of Collection* giving day, month (abbreviated first three letters) and the year; *Name of the Collector, Sex of the Bird,* and the *Collection Number* (each specimen should have its own number and serially recorded simultaneously in the label and the Field Register). The other side of the label should have the vital measurements of Wing, Tail, Tarsus and Bill; colour of the soft parts like Iris, orbital skin or any other soft tissue exposed; and that of beak and tarsus, legs and feet. Weight of the specimen before skinning should be recorded. In addition condition of the gonad should be briefly described or a simple sketch may be inscribed. Ecological notes are essential but should be recorded in the Field Note Book.

Preservation & Curatorial Work

The Curatorial work begins by confirming the identify of the specimen, often necessitated by scanning standard literature and also comparing with standard collection. After confirmation of identification the data is incorporated on the label in the Permanent Register with the Permanent Number of the Institution. Same data is also incorporated in the Catalogue Card and Species Index Card. Finally the specimen is stored in the appropriate place in the storing cabinet.

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HERPETO - FAUNA

D. P. SANYAL*

Introduction

The Herpeto-fauna includes:

Testudines (Chelonians: Turtles, Tortoises, Terrapins) Rhynchocephalia (confined to a single species, the Tuatera, *sphenodon punctatus* of New Zealand, a living fossil). Crocodylia (Gharials and Crocodiles). Squamata (Lizards and Snakes). Gymnophiana (Caecilians): Limbless snake - like burrowing Amphibians). Caudata (Salamanders, tailed Amphibians). Anurans (Toads and Frogs).

Out of about 4120 species under 288 genera and 17 families of the Amphibian fauna of world, about 200 species under 24 genera and 6 families are represented in India. Out of them 16 species belonging to the order Gymnophiana, one species under Caudate, and rest are under Anurans. About 49% species are endemic.

The reptilian fauna of India is represented by about 446 species, comprising 3 species of crocodiles, 32 species of turtles, 174 species of lizards and 237 species of snakes, as against 5680 species found in the world.

Collection

The aquatic forms of amphibians are collected by an ordinary water net fixed on a long handle. Terrestrial forms are caught by hand or net. Arboreal forms are difficult to be located because of camouflage. Tree trunks, leaves and tree holes and bushes should be searched for these. Burrowing forms have to be dug out from under the soil. They could be located by the help of faint tell-tale marks left on the soil surface. Collection should be done both during day and night. Nocturnal forms are easier to be caught when blinded by a torchlight or a petromax light. Male of frogs and toads could also be located by their call which is often ventriloquial. Tadpoles are collected with the help of a water net or by hand. Though they are found almost always in stagnant or running water, tadpoles of some species are found only on the moist surface or rocks where they grow. Care should be taken to collect the various stages of tadpoles including two-legged, four legged and metamorphosed ones.

Since colour of the specimens is lost by preservation, it is important to note the colour in living condition. Shape of the pupil and the exact habitat should also be recorded in a field note book.

Turtles and tortoises live in fresh water, seas and oceans, muddy habitat and on land. Smaller forms can be easily collected by means of a hand net, whereas larger forms are collected by large nets, shooting or harpooning. Crocodiles come on land for basking or catching domestic animals. Like turtles and tortoises, smaller specimens can be easily netted and the large ones can be collected by shooting or harpooning. Trapping is hardly possible. Lizards with diurnal habits can be trapped as well as netted. In case of fast runners, shooting by means of dust shot may be resorted to. Cryptozoic and nocturnal lizards can be collected by raiding their haunts. Snakes are placed in cloth bags when captured. For poisonous or large non poisonous specimens, bags made of stout canvas are useful.

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Provide the sacks with a string and tie to secure the bag. Snakes are specially adapt at working through weak spots in the seam of a bag or through the neck of the bag, unless it is properly tied.

Equipment and Chemicals: water net fixed on a long handle, cast net, two forceps of 30 cm. and 12 cm., a pair of scissors, polythene bags, absorbent cotton wool, metal covered large syringe fitted with hypodermic needles, hammer and chisel; hack saw with blades, 22 calibre pistol with ammunition, rubber gloves; a measuring tape, about 10 m long; a pair of fine pointed dividers; a pair of dissecting scissors with about 6 cm. long blade; a pair of dissecting forceps, about 12 cm.; a pair of forceps, about 25 cm. long; a pair of bone cutters, empty big mouth polythene container, chloroform; formalin; ethyl alcohol; a scalpel with 4 cm. long blade, a scalpel with about 6-7 cm. long blade; altimeter, compass, thermometer for recording maximum and minimum temperature, arsenical soap; common salt etc.

A geology pick is also an useful field tool. This is used for turning rocks, removing bark, splitting open old logs, tearing up rodent burrows and doing many things too dangerous to be done by hand. High long leather boots and heavy leather gloves are also useful. For arboreal (tree-dwelling) species collection by, 22 calibre pistol with bird-shot shells is very useful. Approach within 10 feet of a specimen and then shoot at it. Specimens collected in this manner must be preserved almost immediately to prevent spoilage.

The following data should be included in field notes:

1. Locality should be noted carefully with all details. Distance should be estimated carefully from a good atlas to which future reference can be made. If the collecting locality is very remote then its distance should be referred with the nearest police station or post office. Elevation of the locality should also be recorded.

2. Write the date with month or indicate month by a Roman numeral, such as 8.6.1995.

3. Name(s) of collector(s).

4. Time of collection.

5. Air temperature and other appropriate weather notes.

Turtles may be chloroformed by confining them with a chloroform moistened rag or cotton wad in a closed container for 15-30 minutes. The use of chloroform on other reptiles is not recommended, as severe contortion usually results. Trichloroethylene or ether may be substituted for chloroform with good results, and can be used on most reptiles. Most specimens can be killed by confinement with either Trichloroethylene or ether for 5 minutes beyond the time the animal losses the ability to right itself when turned over. Caution should be observed with ether, as it is highly inflammable and can, under certain storage conditions, explode.

All amphibians and a number of smaller reptiles are easily killed by immersing them in a solution of chloretone (hydrous chlorobutanol). A stock supply is commonly prepared as a saturated solution of chloretone in 95% ethanol, 2 cc. added to a pint of water is effective.

Various other means are suitable for killing reptiles and amphibians. Securing the animal(s) in a cloth sack and immersing the sack in the warm (43-47°C) water is effective, but specimens should be removed immediately after death. Specimens may also be immersed in alcohol (15-25% for amphibians; 50-60% for reptiles).

Fixation

The purpose of fixation is to preserve the actual morphological state and colour of the specimen and to prepare the tissues for microscopic examination. Hence, the fixative should kill tissues quickly, penetrate it uniformly and rapidly, prevent post-mortem decomposition; not to distort the tissue; and should prepare the tissue for staining.

a) Formalin: It is 40% formaldehyde in water. For purpose of dilution, commercial formalin is usually considered as 100% and can be used in 10% strength (1 part formalin: 9 parts water) for fixation. Formalin may be buffered (which helps to reduce discoloration of specimens) by mixing 1 tablespoon of baking soda or borax with each pint of 10% formalin.

b) FAA (Formalin-alcohol-acetic acid) prepared by mixing 10 parts commercial formalin, 50 parts of 96% alcohol, 40 parts water and 32 parts of glacial acetic acid. FAA penetrates tissue far better than formalin alone, and has less tendency to cause cell distortion. The rapid tissue penetration can also be an aid to preserving valuable specimens found dead and perhaps totally decomposed. If FAA is to be used extensively in hot regions, it is recommended that the acetic acid be added just prior to actual use, as it quickly evaporates from the solution. Containers may be cooled by wrapping them in wet rags and shading them to retard evaporation of acetic acid.

c) Alcohol: if neither formalin nor FAA are available, alcohol may be used as a fixative (95% for reptiles; 70% for amphibians).

It is always preferable to introduce fixative into the body cavity by injection or slit in body, as *Faunal* specimens (particularly reptiles and larger amphibians) can decompose internally if simply placed in the fixative. Enough fixative should be injected to fill, but not distend, the animal. Care should also be taken not to damage the femoral pores of lizards by puncturing them with the needle. The neck of turtles should be completely extended and the mouth held open with wood, cork, or tightly wadded paper prior to fixation. Excellent neck extension can be obtained by hooking the dead turtle's upper jaw over a nail or broken branch and letting the animal's hanging weight pull the neck out straight prior to injecting it. The upper jaw can also be hooked over a clip placed over the edge of the fixing tray, and the neck then drawn out. One hemipenis of male lizards and snakes should be partially everted with thumb pressure on the base of the tail, followed by injection to completely evert it. The hemipenis should not be permitted to remain incompletely everted, thread may be tied around the base of the fully everted hemipenis by injection of the fixative alone. Tails of lizards and snakes should be slit lengthways, being very carefully not to break the tail off. If no injection apparatus is available the specimen should be deeply slit in several places ventrally and placed belly-up in fixative. Spread the sides of the slits to admit fixative more easily. Avoid cutting the anal plates of snakes and femoral pores of lizards.

The caecilians should be fixed in the same position as snakes; it is useful to fix these with mouth open, as this greatly facilitates examination of oral characters later on. Lizards with long tail should be fixed with the tail bent. Frogs and toads may be positioned with the sole of the foot down. Toes and

fingers should always be straight and spread apart. Small amphibians need not be injected or slit prior to positioning, as the fixative will penetrate the body cavity quite easily. Small amphibians and lizards may have the field tag tied around the body just anterior tot he pelvic region.

Amphibian eggs and tadpoles are best fixed and stored by dropping them directly into jars of 10% formalin; preserve entire eggs clutches whenever possible. Many amphibians attach their eggs to leaves, twigs, etc. Whenever it is practical, these items should be preserved with the eggs *in situ*, as the latter are often severally damaged by attempts to disengage them. Change the formalin on eggs and larvae after 12 hours. Reptile eggs should be measured (Length and width in millimetres) then injected.

All specimens should be allowed to remain in fixative for 24 hours.

For large specimens, following steps be taken:

1. Snakes: Record the snout to vent and tail length (in mm). Then skin it by making a long ventral incision to the side of the midline; leave the head end attached to the skin; severing these from the carcass (avoid cutting anal plate), and then inject head and tail (evert hemipenis if male) with fixative. With boils, sever the hind part just ahead of the bony, vestigial pelvic elements. The skin may now be preserved by covering the flesh side with cloth or absorbent paper, rolling loosely and immersing in fixative, or by rubbing with borax or arsenical soap, rolling and drying. In this latter instance, it is best to preserve the head and tail separately in liquid. If the specimen is male, a testis should also be preserved. Reproductive condition of females should be noted (i.e. number of ova present, size of the largest ovum etc.) Embryos, especially those of poorly known species should be preserved in liquid. It is preferable to do this by preserving the entire oviduct rather than by removing embryos.
2. Turtles: Avoid cutting the shell. It is preferable to cut the head, neck and forelegs as one unit, the hind legs and tail as a second unit and preserve these in liquid. The stomach and reproductive organs should also to preserved in liquid. Carefully clean out and dry the shell.
3. Crocodilians: Measure and skin the specimen as for snakes, except that the tails should be skinned as well. Feet may be left attached (inject with fixative), instead of skinned out. Rub the skin with borax or arsenical soap and dry.

Final Preservation and Storage

Small and medium-sized specimens may be kept in 6% formalin, large specimens should remain in 10% formalin. Because of formalin's disagreeable nature, most workers prefer to transfer specimens from formalin to alcohol. 70% ethyl alcohol or 95% rectified spirit is suitable. Specimens should be transferred from formalin to alcohol by placing the specimens in a jar of clear water for 24 hours. Seal specimens in airtight museum jars, along with their field data. Specimens should be kept in the dark to prevent colour loss. Check museum specimens atleast twice a year and replace any liquid that has been lost as a result of evaporation.

Each specimen retained in the collection should be assigned a catalogue number (in addition to the field number). Eggs may be catalogued with a single tag designating one clutch or lot. This number should be entered in a permanent catalogue (using waterproof ink), along with the species name, locality, collector(s) name(s), date and sex. As a cross-reference, it is useful to maintain a card file (by taxonomic category) in which a single card is used for each species. On this card may be entered numbers from the catalogue that apply to these species. It is convenient to place a label bearing

species name, catalogue number and locality data with each container. These be written in permanent ink on thick durable paper.

FISHES

A. K. GHOSH*

Introduction

Both fresh and brackish water fishes can be categorised under lotic or lentic types. The former, also designated as running water-series, includes inland waters in which the water body as a whole is continuously in a state of motion in a definite direction. Rivers have a unidirectional flow of water and estuaries oscillate up and down with the tides. Lentic waters, also designated as standing-water-series, include lakes, ponds, swamps etc.

Collection

The spawn collection net, generally known as shooting net, in essence, is a funnel-shaped net of finely woven netting and is operated in shallow margins of a flooded river with the mouth of the net facing the current. At the end of the net, there is usually a receptacle, termed as "gamcha" which is sometimes a rectangular open piece of cloth and has the shape of monk's hood. The non-metallic strainer is used to remove larger organisms, debris etc. and spawn are stored in gapes or in specially prepared mud pits. The spawn collected on the muslin cloth after sieving is to be measured in 200 ml., 100 ml., 50 ml., 30 ml., 20 ml., and 5 ml. measuring cups, depending upon their bulk.

The results of experimental fishing in the reservoirs suggested that 50 mm. and 63 mm. meshed nets were the most efficient at Mettur and Krishnarajsagar respectively (Jhingran, 1983). To improve the catching efficiency, some workers recommend reconstruction of the indigenous gill nets on the following lines: i) application of a hanging co-efficient of 0.5 while rigging the nets, ii) complete framing of the nets attaching breast-lines and a lead line, iii) better distribution of the buoyancy along the float-line and similarly equal distribution of sinkers along the lead-line. The methods of fishing are restricted to gill or drag-netting. The latter being commonly used in small reservoirs which are free from tree stumps and weeds, the surface gill nets vary in length from a hundred feet to over a mile, the width varying from 2.4 m to 12.0 m. The meshes range from 38 mm - 100 mm bar. These nets have floats but no sinkers. The bottom-set nets are long and about 0.6 - 0.9 m wide, with a mesh of 25 - 76 mm bar. These have floats and balancing sinkers. Long lines, used for catching predatory fishes, are not very commonly used in the reservoirs. For the purpose of procuring a sustained yield of fish from the reservoir, avoiding waste of fishery resources, and of conserving the fish stocks, various rules and regulations under the Indian Fisheries Act are in force in various states.

Different types of fishing gears are operated in the estuaries. Bulk of total fish landings are contributed by a kind of fixed gear called bag-net. About 70% of the total landings from the estuary were taken by bag-nets which were operated in pairs from larger boats locally known as `chota nauka'. Collecting fishes varies from place to place and species to species. A majority of the fresh water fishes are caught by sieve nets which are operated across a river and either dragged or closed like a purse. Hill-stream fishes are often collected by damming a stretch of the river or diverting the water for the stream to dry up. Fishes are then picked up by hand from below stones, rocks and boulders. Often poisonous or narcotising roots and drugs are used to collect such fishes. Use of bait or fly-fish for some revering fishes are also common. The collector may use any means to capture fish with the

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sanction of local authorities, but the aim should be to obtain them intact with all their body parts undamaged for taxonomic studies.

Equipments

The fishing crafts and gears in use throughout the country are indigenous, non-mechanised and locally built. They have been designed to suit the local conditions. They are Catamaran, Masula boat, Dinghy, Nauka, Canoes and built-up boats. Fishing implements are of various types *viz.* Cast net, Fixed net, Bag net, Gill net, Drift-gill net, Shore Sieves and Boat Sieves and are used in sea-fishing. The inland fishing gears are varied right from catching with the hand to the operation of large and indigenously designed nets. They are Scoop net, Dip net, Patta jal, Drag net, Gill net, Changadam (For mullet catching), Drift gill net, Encircling net etc.

Quantitative Method of Sampling

Inland fisheries are traditionally classified into capture and culture fisheries, the former being mainly explorative of natural populations and the latter affording enough room for intense human intervention by stock control and other management practices. For estimating yields from various inland aquatic ecosystem, it is very much essential to classify them in different production regimes. Inland fishery resources are classified into the following categories:

1. Fresh water ponds and tanks
2. Brackish water impoundments
3. Small and large reservoirs
4. Fresh water lakes
5. Ox-bow lakes
6. Streams, rivers and canals
7. Estuaries and lagoons

There exist wide variations in fish yield from different classes of aquatic ecosystem in the country. The annual yield from a unit area of an intensively managed fish pond can be as much as thousand times that of an average annual fish catch from a reservoir. Hence suitable sampling methodology of collection and estimation should be adopted for the collection of data.

Population and Sampling

Taking samples is a procedure used in nearly all fisheries investigations and from the sample taken we intend to generalise about the population under investigation. For example taking a sample of catch from a vessel operated in water body, we want to say some-thing about the total catch of the fish from it. Firstly we must define what we mean by a population. In this instance the population may comprise all the fish landed at a particular landing centre in a particular month by the vessels. The efficiency of any sampling scheme is its amenability to a satisfactorily generalisation about the population from the sample or samples.

As in any sampling problem, it is essential to define the population being sampled and to choose an appropriate sampling unit. For this it is always easy to consider where the fish are landed or first became available for weighing or recording, rather then the position where the actual fishing operations take place. Obviously the fishery may be based on a number of distinct landing places each of which can be considered as a unit. In a more complex situation such as coastline or a river, the natural unit is a certain length of river or coastline. The size of the unit should be best taken

sufficiently small for one man to cover preferably in a day. Some of the primitive fisheries take place in any area an area i.e. in swamps or irrigation channel where fish either get consumed locally or sent to market in small lots. Here unit may be an area of ground.

The first step would be to achieve some stratification, dividing the units (landing places, stretches of river) according to the order of magnitude of fisheries. This needs some preliminary survey of the fishery and where there are no definite landing places, some geographical survey to delimit precisely the boundaries of the units may be done.

Estimation of Resource and Catch

For estimating the water resource and fish catch from ponds and tanks, a state should be divided into a number of agroclimatic zones for estimation purposes.

The sampling design for estimating the extent of culturable water area under ponds and tanks would be stratified into a two-stage cluster sampling. A distinct is divided into number of strata approximately in equal size of water area/no. of villages. A sample of n clusters of five villages each will be selected from each stratum. The cluster of villages will constitute the first stage unit and ponds within cluster as the second stage unit. The selected villages would be surveyed completely and all the water units in the villages are to be enumerated for estimating average area.

For estimating total catch of fish 'm' ponds/tanks would be selected from each cluster at random from the total no. of ponds in the cluster. Further sampling in time may also be adopted so that each water unit is visited atleast once in a month depending on the availability of manpower for recording the catch from each selected pond for providing the estimates of monthly catches.

Estimation Procedure

Notations

N = Total no. of clusters
n = No. of sample clusters
Mi = No. of units in sampled clusters (i = __n)
Xi = Area under resource in sampled clusters (i = 1,2, __n)
mi = Sampled ponds in each selected clusters (i = 1,2, __n)
Yi = Yield of sampled ponds (i = 1,2,__n)
Yij = Yield of jth pond in the ith clusters (j = 1,2 _mi, i - 1,2_n)
Xij = Area of jth sampled pond in the ith cluster (j = 1,2 _ _ mi, i = 1,2 __n)

Area and Number of Ponds Estimate

Average ponds per clusters

$M = 1/n \Sigma Mi$

Total no. of ponds

$M = N/n \Sigma Mi$

Average area per cluster

$X = 1/n \Sigma Mi$

Total area

$X = N/n \Sigma Xi$

Yield statistics

Average yield per cluster

$Y = 1/n \Sigma Mi\ Yi$ where $Yi = 1/mi \Sigma Yij$

Total yield

$Y = N/n \Sigma Mi\ Yi$

$(Y) = N^2(1/n - 1/N)\ 1/n - 1(Xi-X)^2$

$V(X) = N^2(1/n - 1/N)\ 1/n - 1\ (XCi - X)^2$

$s^2b = 1/n - 1\ (Mi\ Yi - 1/n \Sigma MiYi)^2$

$s^2wi = 1/mi - 1\ (Yij - Yi)^2$

Preservation

In the field: Small specimens of fish from shallow waters can conveniently be collected by means of a small bag net. The size of the handle must be adjusted according to the requirements of each individual. As soon as the fish are collected they should be transferred to a weak solution of formalin, about 2 to 3 per cent (5 to 7 percent commercial formaldehyde). The fish die in this solution after dashing about for a couple of minutes and usually at the time of death all the fins become fully expanded, making it possible to count without much difficulty, the number of rays, etc., which are essential for the proper identification of many species. The fish should be left in this solution for 4 to 5 hours and then preserved in rectified or methylated spirit. If no spirit is available, they may be kept in a container with the fishes wrapped up in cotton wool and dampened with weak formalin solution. To get best result this solutions is neutralised with 5-10 g. of Borax per litre of water. The volume of the material to be preserved is never allowed to exceed that of preserving liquid. For proper fixation, the fish is kept in this solution for 4-5 hours. Medium sized fishes (10-30 cm) should have a narrow cut made on the abdominal wall at one side of mid-ventral line. Care should be taken to avoid a deep cut to expose or injure the alimentary canal or other organs. For longer specimen (more than 1 foot) undiluted formalin should be injected in several places and the belly should be slited in 2-3 places. The delicate fishes like *Chela*, Clupeoids etc. are injected with dilute formalin with particular care. In case of fishes with keeled abdomen, the incision is preferably made on the left side. In case of bigger sample, fins may be kept in stretched condition in concentrated formalin for a few hours. As soon as the fishes are caught, all colours, colour patterns, spots, blotches, number, design etc. are carefully noted in the field note book.

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INSECTS: LARGER ORDERS

S. K. TANDON*

Introduction

Insects comprise nearly 80% of the animal species living on this earth and are grouped into about 30 Orders, of which Coleoptera is the largest, followed by Lepidoptera, Diptera, Hemiptera and Hymenoptera and so on. India holds over 6% of insects comprising about 15,000 species of beetles (Coleoptera) in 103 families, 13,000 species of moths and butterflies (Lepidoptera) in 80 families, 6,000 species of mosquitoes, midges and flies (Diptera) in 87 families, 6,500 species of bugs, aphids, coccids, cicadas and hoppers (Hemiptera) in 77 families 5,000 species of bees, wasps, ants and hornets (Hymenoptera) in 57 families and 834 species of grasshopes and crickets (Orthoptera) in 22 families.

Habitats

Terrestrial Habitat : The majority of insects live in this habitat. Besides being exposed air as free-living insects, they occur on plants and on live or dead animals, externally or internally, as adults or immatures.

Subterranean Habitat: This comprises soil and sand. This habitat is invaded by apterygotes, mole-crickets, some ants and termites, and immatures of many groups like Diptera, Coleoptera etc.

Aquatic Habitat : The aquatic habitat is classified as (i) fresh-water and (ii) brackish water. The fresh-water includes both confined water in ponds, lakes, pools, ditches, etc. and the running water like mountain rivers, streams, etc. Water beetles, water bugs, a few Diptera and immatures of a few crickets, are aquatic, but immatures of some Diptera are strictly torrenticolours. A few adults and immatures of these insects inhabitant brackish water. Besides, a few aquatic insects like beetles and fly larvae live in hot springs.

Collection

Collecting insects is done in the field by adopting a suitable method depending upon the size and structure of the insect-body. It is, therefore, imperative for the collectors to have an idea of characteristics of that insect so as to overcome any probable damage of those characteristics. The methods are as follows.

Hand-picking: Small insects with soft body like bugs (aphids and coccids), beetles, orthopterans, hemipterans, and larvae living under bark, etc. dipteran parasites and miners and hymenopterans are best collected by hand either with the help of a fine hair-brush moistened with 70% ethyl alcohol or by forceps.

Beating bushes: Beating vegetation over a tray or a sheet of paper by a long stick is usually employed to detach insects like hemipterans, small beetles, etc. from foliage or twigs. An insect-net should be kept ready for capturing any crawling, jumping or flying insect.

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Sweeping by an insect-net : This is the most convenient method of collecting good number of free-living specimens in a short time but it is advisable to remove specimens from the net after sweeping twice or thrice so as to avoid any damage of specimens by repeated strikes. Sweeping over the herbage generally yields a good catch of different kinds of insects in an area.

Netting in the air: Free-living insects that are on the wing are best collected by the aerial neting. Generally, Lepidoptera, Hymenoptera, Orthoptera and Diptera are collected by this method.

Netting in water: Insects living in water are collected by a water-net of convenient size. Water-bugs, water-beetles and immatures of Diptera, etc., are collected by this method. However, immatures living deep in mud are required to be collected by other means along with mud and are to be sorted out in the laboratory.

Trapping: Different kinds of insects are trapped in a number of ways so as to study their distribution, seasonal incidence, migration and other biological phenomena. The light trap is used to collect photophilic insects among Diptera, Lepidoptera, Coleoptera, Hymenoptera, Hemiptera, Orthoptera, etc. The sticky trap is used to capture especially the flying insects like Diptera, Hemiptera and insects of soft body and small size. The Malaise trap is convenient for larger insects and is set in open field near forests. The pitfall and bait traps are essentially used with different types of baits for attracting insects that they like most. The former is used to capture mainly beetles, and the latter for Diptera, Lepidoptera, etc.

Collecting by funnel: The Berlese funnel is used to extract small insects like Hymenoptera (wingless forms), Diptera (wingless forms), etc. and immatures of Dipterra, etc. that live either in leaf-litter and sub-soil or in the nests of ants, termites, etc. or in the burrows of mammals like rodents.

Specimens collected by the above methods are killed either with the help of the killing jar designed for the purpose or directly preserving them ethyl alcohol, if the colour of the body as well as the bristles, scales, hairs, etc. does not bear any taxonomic value.

Equipments

Sweeping net: The sweeping net or the aerial net, comprise a metal ring attached to a handle of convenient size, and a cloth bag worn around the ring. Usually, the depth of the bag measures 75 cm. or twice the diameter of the ring of 20 cm. and the handle of 45 cm. The handle requires to be longer for the purpose of aerial netting. Likewise, the handle of the water-net may be still longer and the bag may be of nylon in lieu of cotton.

Traps: There are different types of traps as already mentioned. The pitfall trap or the bait trap is essentially a trap with a kind of bait that the insect like most. The light trap is essentially a box-type or a cylinder-type device with light arrangement for attracting insects from distant places. On certain occasions, the Malaise trap also becomes useful.

- *Aspirator*: This is operated to suck the small insects into it.
- *Brush, forceps and scissors*: For the purpose of hand-picking these are indispensable.
- *Axe, knife and hammer*: These are necessary tools for collecting insects that inhabit, soil, rotten wood or under bark.

- *Tray, white cloth and hand-lens*: These are required for collecting and sorting specimens.

- *Killing bottle*: This is used to kill insects captured by any of the means mentioned above. Usually the bottle contains a layer of NaCN or KCN covered with plaster of paris. A bottle containing a layer of cotton covered by a blotting paper and moistened with either chloroform or benzene is also used as a killing bottle.

- *Vials, chemicals and cotton*: Vials usually filled with 70% ethyl alcohol are kept ready in the field for capturing and preserving small insects or immatures.

- *Polythene packets and paper packets*: Paper packets are used to kept insects after killing and are put in polythene packets for temporary storage.

- *Field labels and note book*: These are very essential for references to the insect and its place of occurrence. Insects are to be kept in any hard box like cardboard box containing Para-di-chloro-benzene or simply naphthalene (never both together) for probable safety against fungal growth.

Group-wise Methods

- *Orthopteroids*: The grasshoppers, crickets, stick and leaf-insects and the cockroaches are found almost everywhere near vegetation. They are collected by the sweeping net and are preserved dry. Some soft-bodied grasshoppers may, however, be preserved in alcohol.

- *Hemiptera*: The bugs are mostly found near vegetation. They are with collected by sweeping net or beating the bush and lower branches over a piece of cloth, or picked up individually from the stem of trees. Those living in water are collected by water net. Some of the bugs are of sedentary habit, living in large colonies on the host plants sucking their sap, for example aphids, mealybugs and scale-insects. They are collected with the help of brush and preserved in spirit, whereas the larger bugs are preserved dry. It is important to note the name of the host plant.

- *Lepidoptera*: The butterflies and moths are collected by sweeping a new when they visit or fly about near flowers, damp places and decaying vegetation. They are generally killed by pinching their thorax hard and kept in separate paper packets or triangles to avoid shedding off of their wing scales. The moths are generally found flying at night and are attracted to light. One may collect the caterpillars, which do the major damage to plants, and breed them in laboratory for recording the pest species of different crops and plants.

- *Diptera*: The true flies, mosquitoes, midges, are generally small, delicate insects with only a single pair of wings, their hindwings being transformed into halters. They are strong fliers and some of them may be found almost everywhere. Some are of importance due to their nature of carrying diseases. Some others infest and damage vegetables and fruits. As they are very delicate, care has to be taken to preserve these insects so that the bristles and scales are not damaged. They should preferably be pinned or set on card, right in the field, to avoid damage. Larger insects may be kept in paper packets or triangles.

- *Hymenoptera*: The wasps, bees and ants are distinguished from other insects by their membranous wings and constricted abdomen. The ovipositor in these insects is modified for sawing, piercing and stinging, as such care should be taken in collecting these insects in the field. Ants, bees and some wasps are polymorphic, it is advisable to collect this different castes.

Parasitic species should be collected by rearing them from the eggs, larvae or pupae collected along with the hosts.

- *Coleoptera*: The beetles and weevils are found in a variety of habitats, in damp soil, mud, sandy ground, cowdung, under stones, logs, debris, live or dead wood, on vegetation and in water. Most can be collected by sweeping net or by picking individually, or by beating the bushes over a piece of white cloth. The aquatic beetles are collected by water net. They are killed in the killing jar, or some larger ones by putting them in hot water. All beetles are preserved dry by pinning or in case of smaller specimens by mounting on card.

Preservation in the Field

It has already been indicated that those insects that are collected from aquatic or Semi-aquatic conditions are to be preserved in 70% ethyl alcohol. The same course should be followed in case of immatures of any kind. However, those insects that have the colour and chaetotaxy accountable in taxonomy, should be kept dry in packets temporarily till they are brought to the laboratory for preservation by some other treatment. It should be borne in mind that the insects are to be dried well in the sun.

Field Labels and Notes

It is necessary for the collectors to attach the collection data, such as place of collection, substratum, ecological conditions, altitude of the place, date of collection, name of the host, if any, name of associated animal, if any, name of the collector, etc. to the insects they have collected in field.

Collectors are also required to take notes of the situation in details with particular reference to the data with the insects in packets in their day-to-day field note books. All these information would be of great help in determining a species, its environment and its distribution pattern.

Selected References can be found in the following chapter.

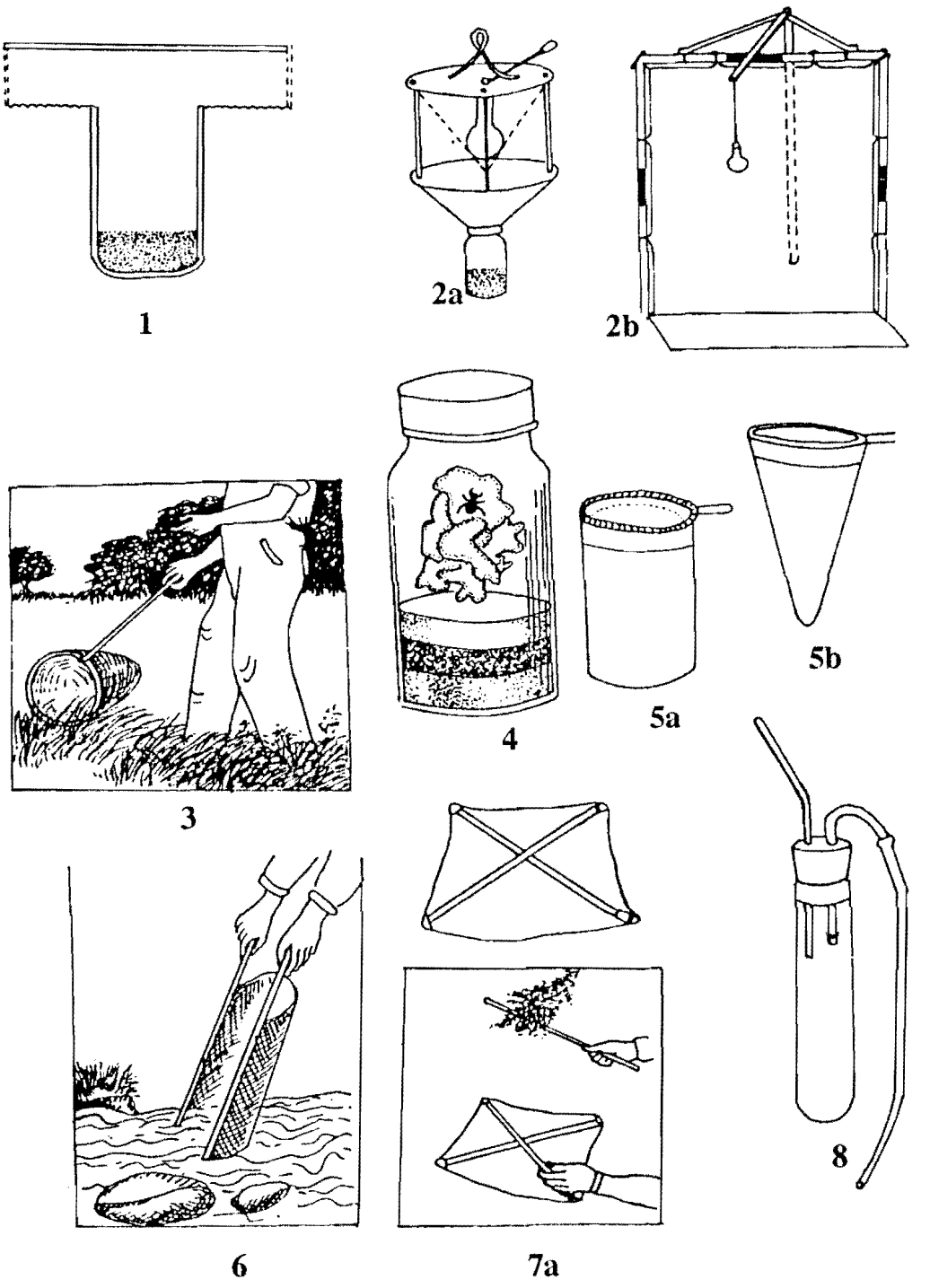

Fig.3 - **Collection Equipments for Insects**

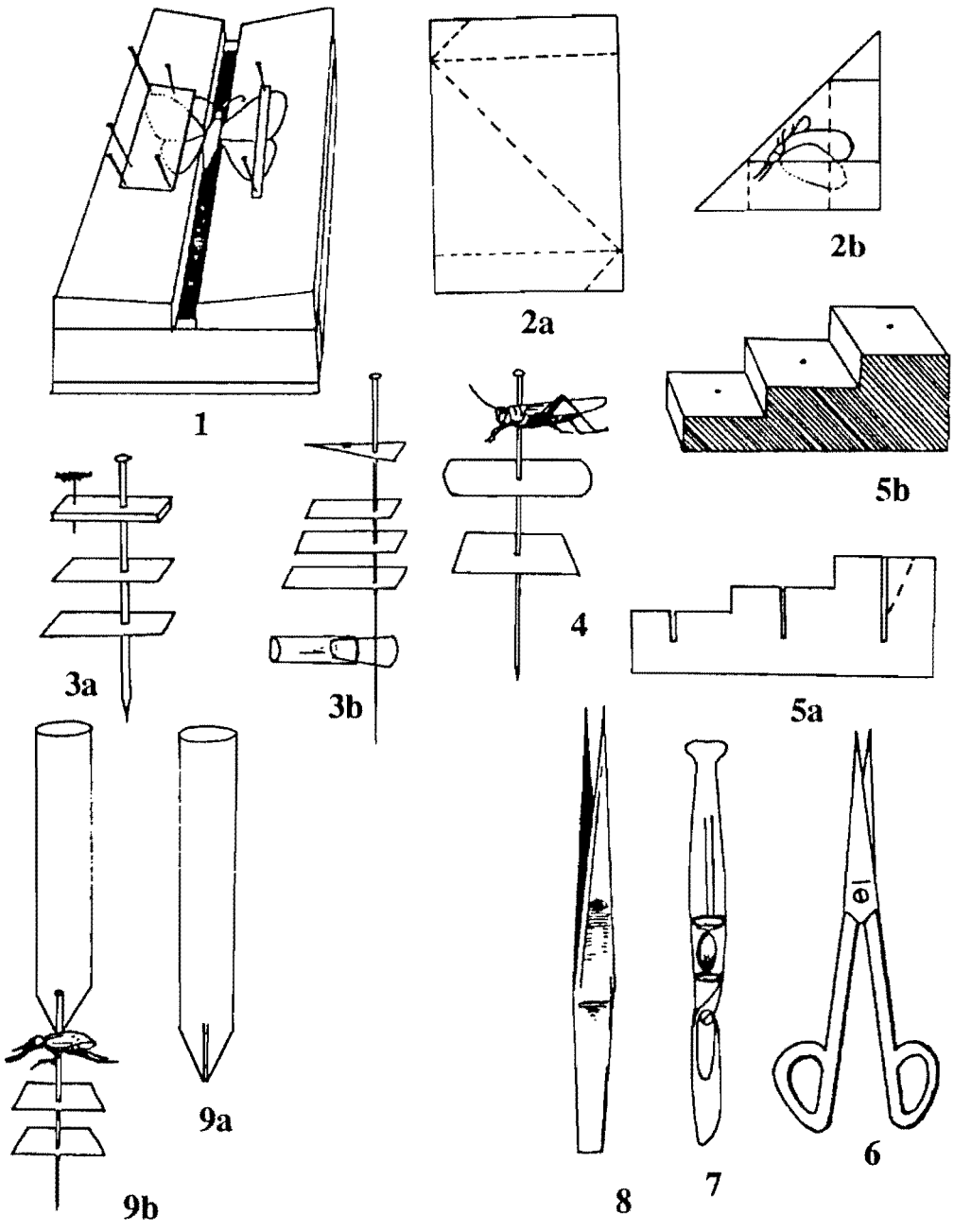

Fig.4 - **Setting and Pinning of Insects**

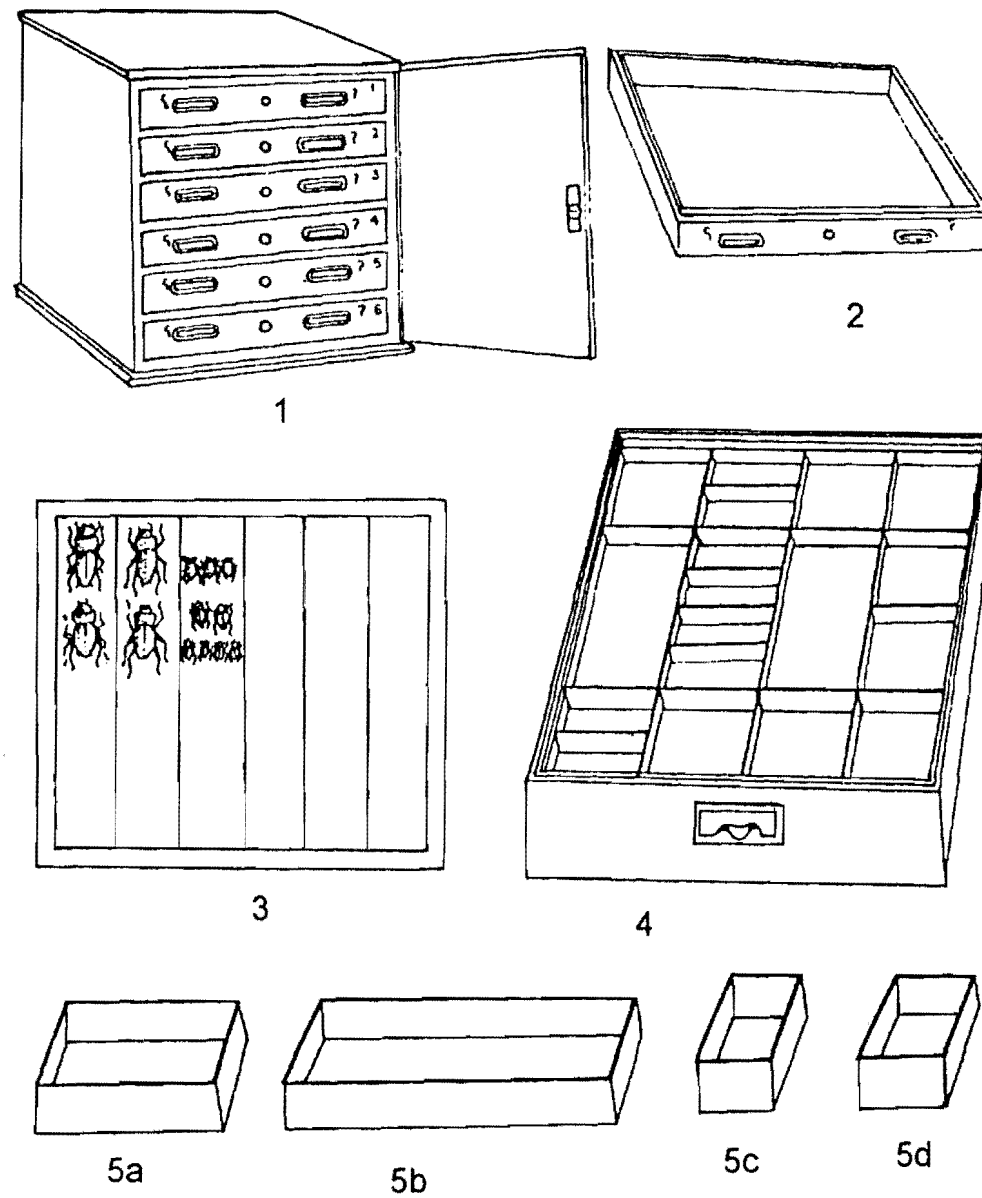

Fig.5 - **Storage of Insects**

INSECTS: SMALLER ORDERS

S. K. GHOSH*

Introduction

By a conservative estimate, the Class Insecta encompassed atleast 8,53,000 extant species from the world, classified under 29 Orders of which four Orders in the Apterygota (primitive and lacking wings) and 17 Orders in the Pterygota (with wings) are dealt with here, while the six Larger Orders of insects have been dealt in the previous chapter.

Insects can be found in forests, grasslands, deserts, cultivated lands, urban areas, bodies of fresh and salt water, and flying in the air. In other words, they occupy all major habitats. Specifically they are found in or on soil and water, decaying plant or animal matter and, living plants or animals. Many insects are able to occupy more than one habitat and feed on different foods during different stages of their lives.

In soil, insects are especially rich in variety in the litter of leaves and dead plant matter. Some penetrate the upper layers of soil by following natural crevices or entering burrows, while others are adapted to digging burrows themselves. Rocks and other covered objects provide shelter for ground dwelling forms. Decaying logs are occupied by many types of insects (e.g. Termites). The subcortical habitat beneath the bark of dead tree is favoured by insects.

Insects that feed on green plants are termed phytophagous. All parts of green plants are attacked : roots, stems, twigs, leaves, flowers, seeds, fruits and sap in the vascular system. Insects are main consumers of pollen and nectar in flowers. Insects also live on primitive plants such as fungi, algae, lichens, mosses and ferns.

Insects feed on many other kinds of terrestrial animals. Insects that kill other insects are termed entomophagous. Of these, the predators kill their prey more or less immediately (Odonata, Neuroptera) while parasitoids feed externally or internally on their host. Some ectoparasites live on the host (bitting and sucking lice; adult fleas). Some endoparasites (Strepsiptera) live internally on other insects (Hymenoptera, Orthoptera).

Some insects construct shelter about their bodies of rolled leaves or plant debris. Many that have a resting pupal stage in the life history construct a special silken cocoon or chamber in soil or decayed wood. Termites may construct elaborate nests that provide some degree of control over temperature, humidity and light, as well as to provide a defensive fortress against enemies.

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Table - 1. Habitats of Insects

Order	*Habitat*
Group I Apterygota	Wingless, Young's resemble adult
1. Thysanura	Book shelf, under stone, logs, debris.
2. Diplura	Soil, moss, rotten wood, damp places.
3. Protura	Moist soil, decaying leaves, under stone bark
4. Collembola	Soil humus layer, moss, caves, on snow, termite nest.
Group II Pterygota	Winged insects
5. Ephemeroptera	Near water
6. Odonata	Near water bodies, among tall grass and river bank
7. Plecoptera	Around water bodies
8. Phasmida	Foliage of trees and bushes
9. Dermaptera	Under stones, crevices, bark, garbage.
10. Embioptera	In their own silk tunnels, in ground, under stone, debris.
11. Dictyoptera	Green tropical vegetation, and domestic habitats.
12. Isoptera	Decaying logs, mound, bark, ant's nest, unders tone.
13. Zoraptera	Under bark and decaying wood.
14. Psocoptera	Foliage in bushes and shrubs, dust in uninhabited rooms, bark, book-bindings, in bird and rodent nests, among lichens.
15. Phthiraptera	Ectoparasite on birds and mammals.
16. Thysanoptera	Plants (foliages, flowers and galls).
17. Neuroptera	Shrubby bush, hill forest, cultivated crop field, around water bodies
18. Mecoptera	High altitude insect; in all types of vegetation. Near water and damp moss.
19. Trichoptera	Ectoparasite on birds and mammals.
20. Siphonaptera	Endoparasitic in Diptera, Hymenoptera,
21. Strepsiptera	Hemiptera, Orthoptera

Collection

Collection methods in detail are dealt with in the preceding chapter.

However, during the field work following equipment's are necessary; nets; for sweeping, aerial netting or aquatic net; bush, forceps, twigcutter and scissors; aspirator; axe, knife, hammer: for use in soil, termite mound, under bark or rotten logs; killing bottle; collection vials: assorted sizes are necessary for keeping insects of different sizes; hand lens: to examine materials in the field; paper packets: to keep the dry insects.

Chemicals: Napthaline balls or Para - dichloro - benzene for keeping in the storage boxes; and 80% alcohol (rectified spirit) for wet collections.

Cotton : For killing bottle and also for packing.
Taps, Enamel tray.
Petromax lamp or electric light: For using as light trap or light attractant, etc.

Collection of Ecto-and Endoparasites:

Siphonaptera: The insects may be collected either by fine forceps or by pressing the parasites gently with cotton soaked in chloroform or alcohol which may either kill or make it inactive for killing. When the parasites get detached from the host they will be preserved in 80% alcohol.

Phthiraptera: These insects may be collected in the following manner:

i) Picked up by fine forceps from the host's body.

ii) Body of the host (except the head; if alive) may be kept inside a polythene bag or fumigation jar with some chemicals preferably chloroform, either or carbon disulphide. The chemical will kill the parasite or force it to leave the body of the host. Then the parasite may be preserved in 80% alcohol.

iii) Chemical extraction is only useful in case of dead hosts, if the skin is not required to be preserved. The parasites are obtained by dissolving a piece of skin in 5-10% KOH or NaOH and filtering the dissolved contents.

Strepsiptera: Their hosts namely, Hemiptera or Hymenoptera, may be kept in capacity to capture males. Virgin females of stylops secrete sex pheromone which attracts male. Males may also be collected at light. Females may be collected by dissecting the stylopised insects.

Preservation

As soon as the specimens are captured and killed for the purpose of scientific study, every specimen must properly be stored right from the field to the place of work. During field surveys it is preferred to keep unpinned dry specimens in paper envelopes. These should be arranged loosely in a row in storing boxes with naphthalene powder. The small and soft bodied insects are sometime preserved in 80% alcohol within a small vial.

For permanent preservation, the dry specimen may be kept in any standard size insect box with necessary chemicals (Naphthalene powder, liquid benzene, camphorcarbolic acid mixture in 1:3 ratio) to check the growth of fungus or the damage caused by other insects, the insect boxes may be kept in insect cabinets. Specimens preserved in alcohol may be kept as such with locality label in each individual vial.

Preparation for microscopy

a) ***Genitalia:*** The study of genitalic structures specifically in males is one of the important subjects for proper identification of the specimen upto the level of species. For a closer examination of genitalia, the terminal abdominal segments of males are removed and boiled

in 5% KOH solution for 5-10 minutes. Then it is rinsed through water, alcohol and glycerol. The internal structures are removed for examination. After the examination, the genitalia may be mounted on slides or between the two cover slips if it is desired to keep the genitalia tagged with the concerned specimen. This technique is used specially in Neuroptera and Trichoptera.

b) ***Slide preparation*:** Preparation of slides for the whole mount of specimens to be studied under microscope may be different in various groups. However, the technique related to Order Thysanoptera for the preparation of slide is given here. The material may be kept in 5% KOH generally overnight. The treated material is rinsed through water and dehydrated in 50%, 70%, 90% and absolute alcohol and cleared in clove oil and mounted in Canada balsam. Utmost care should be taken so that the legs and wings are well spread. Otherwise the identification i snot possible.

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AQUATIC INSECTS

V. D. SRIVASTAVA*

Introduction

Four insect orders are termed as strictly aquatic, as all members of these need aquatic phase in life cycle as oviposition to last instar larvae, which are truly aquatic and utilise dissolved oxygen. These orders are Ephemeroptera, Odonata, Plecoptera and Trichoptera.

Nine insect orders are of whole not all members but only part are aquatic. Some of these are aquatic throughout their life or only part of it as immature. Four of these namely Diptera, Hemiptera, Coleoptera and Neuroptera have relatively fair representation in aquatic ecosystems; while other five orders viz., Megaloptera, Hymenoptera, Collembola, Orthoptera and Lepidoptera have only fringe representation in aquatic realm.

Aquatic insects have importance as fish food, fish predator, fish bait, bio-indicator of pollution and in biological control. Aquatic insects have by and large colonised fresh water ecosystems, with but a very few with meagre and fringe representation in estuarine or coastal water.

Aquatic habitats are bradly divisible in two categories:

i) Lentic (= placid): e.g. ponds, lakes, marshes, bogs etc.
ii) Lotic (= running): e.g. rivers, streams, springs etc.

Collection

Collection of Aquatic Insects need be pre-planned, based on mainly type of study to be undertaken. Site should be selected and its background clearly understood i.e. its topography, size of waterbody including depth, types of vegetation (rooted/ floating/ submerged etc.), vegetative covers (canopy providing shade and adding organic matter) and utilisation of particular wetland. Establishment of Collecting Stations should then be made for regular sampling throughout the period of the experiment. These Stations should be representative of all ecological niche of all zones of water body planned to be studied. Sampling Strategy should involve selection of proper sample size and sampling devices. These include nets and screens, water nets, metallic screen, sieve nets, and Needham's metallic apron nets; artificial substrata sampler, floating substrata sampler; traps like submerged fly traps, tent traps, inverted cone emergence traps and water light trap; dredged like Ekman's dredge, Peterson dredge, Surber sampler, Hess sampler, drag type sampler and square foot tray.

Preservation

Aquatic insects are preserved either dry or wet. Insects for dry preservation can be effectively killed in traditional "cyanide killing bottle". It should be noted that cyanide used is deadly poisonous and obviously should be used with utmost care. Safer killing bottles can be made by using benzene, ethyl acetate, carbon tetrachloride; these materials are poured on to plaster of paris, cotton or even tightly packed paper and the bottle may be re-charged as needed. Sufficiently thick circular paper disc should be placed above such material so as to avoid direct contact with insects being killed.

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Specimens thus killed are better kept in insect packets or 'paper triangles' made of butter paper. Preferably one specimen per packet with adequate particulars on label be inserted in each packet. These are kept using sufficient amount of chemicals to avoid museum specimen's pest such as Psocopterans, ants, moths, etc.

Dry killing preservation is advised for emerging adults of Odonata, Diptera, Ephemeroptera, Neuroptera, Trichoptera etc.; but for not their larval forms.

Wet preservation can be effectively used for the larval forms of above mentioned insect Orders. All Stages of these insects can be best killed and preserved in 70% - 80% ethyl alcohol. Sometimes adults of Ephemeroptera, Odonata, Plecoptera, Hemiptera, Coleoptra etc. are also wet preserved; and may serve parallel set of collection. This ensures certain pigments lodged in hypodermal layer preserved, which otherwise gets faint or decoloured in dry specimens.

Other details applicable to the aquatic insects are given in the preceding two chapters.

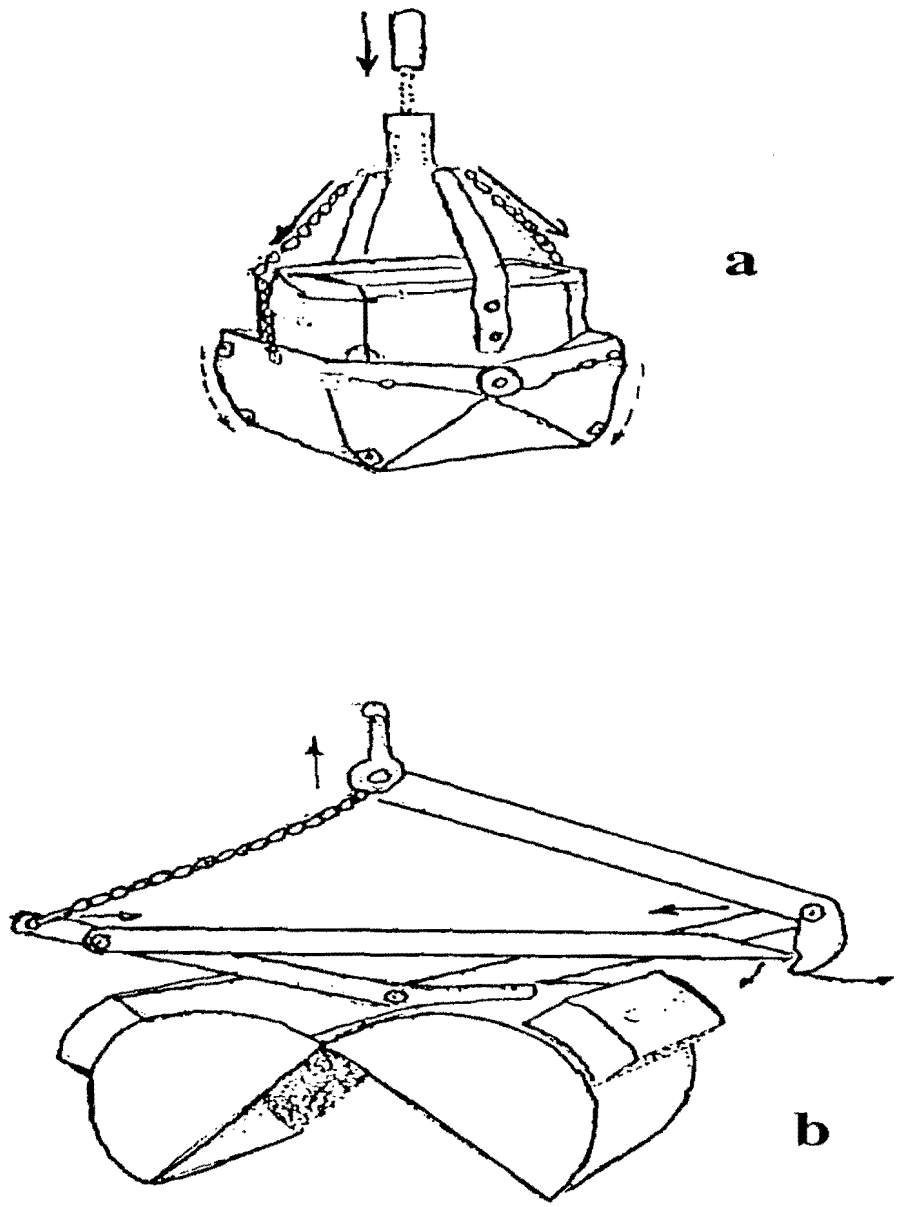

Fig. 6 - **Collection Equipments for Aquatic Insects**

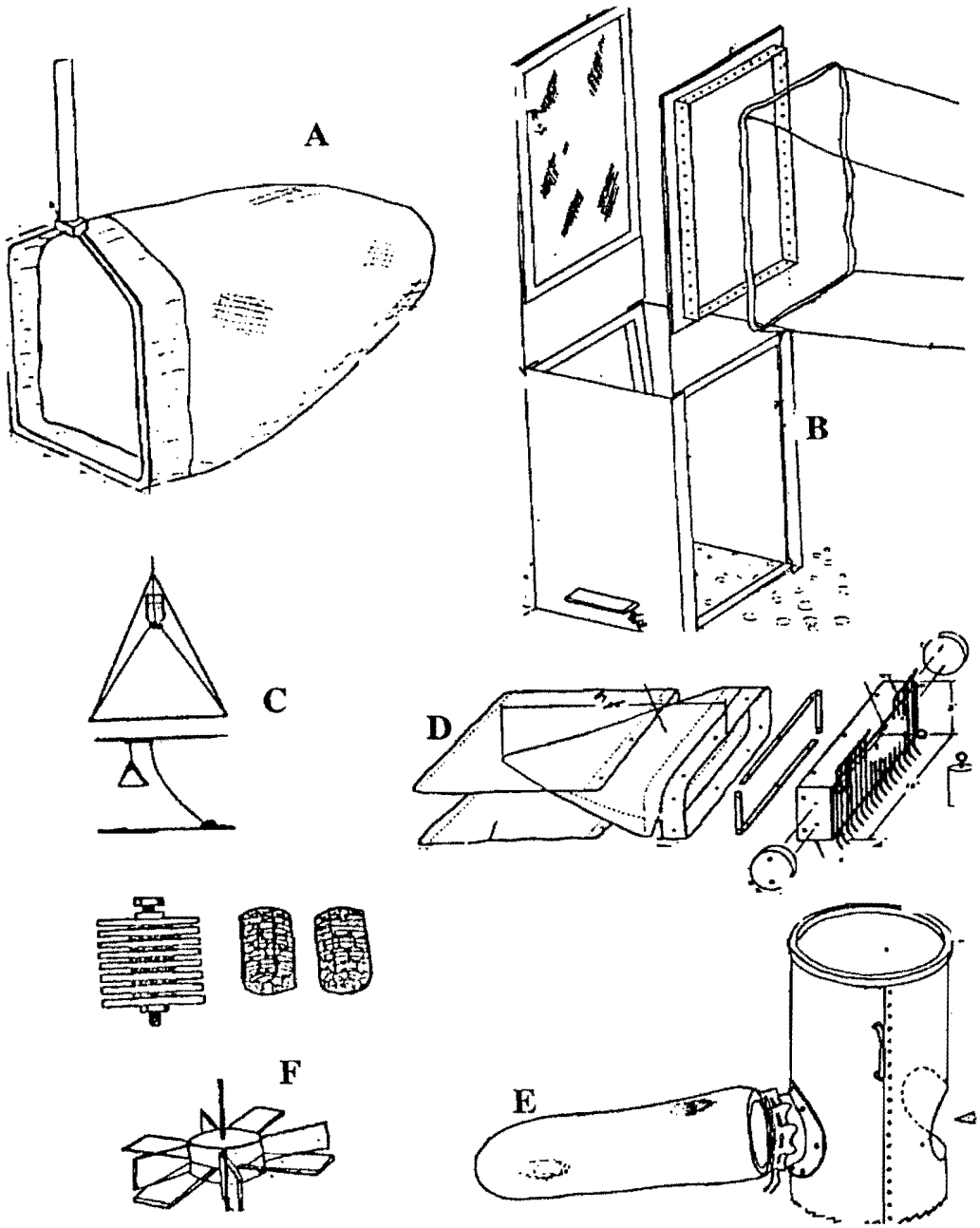

Fig. 7 - **Collection Equipments for Aquatic Insects**

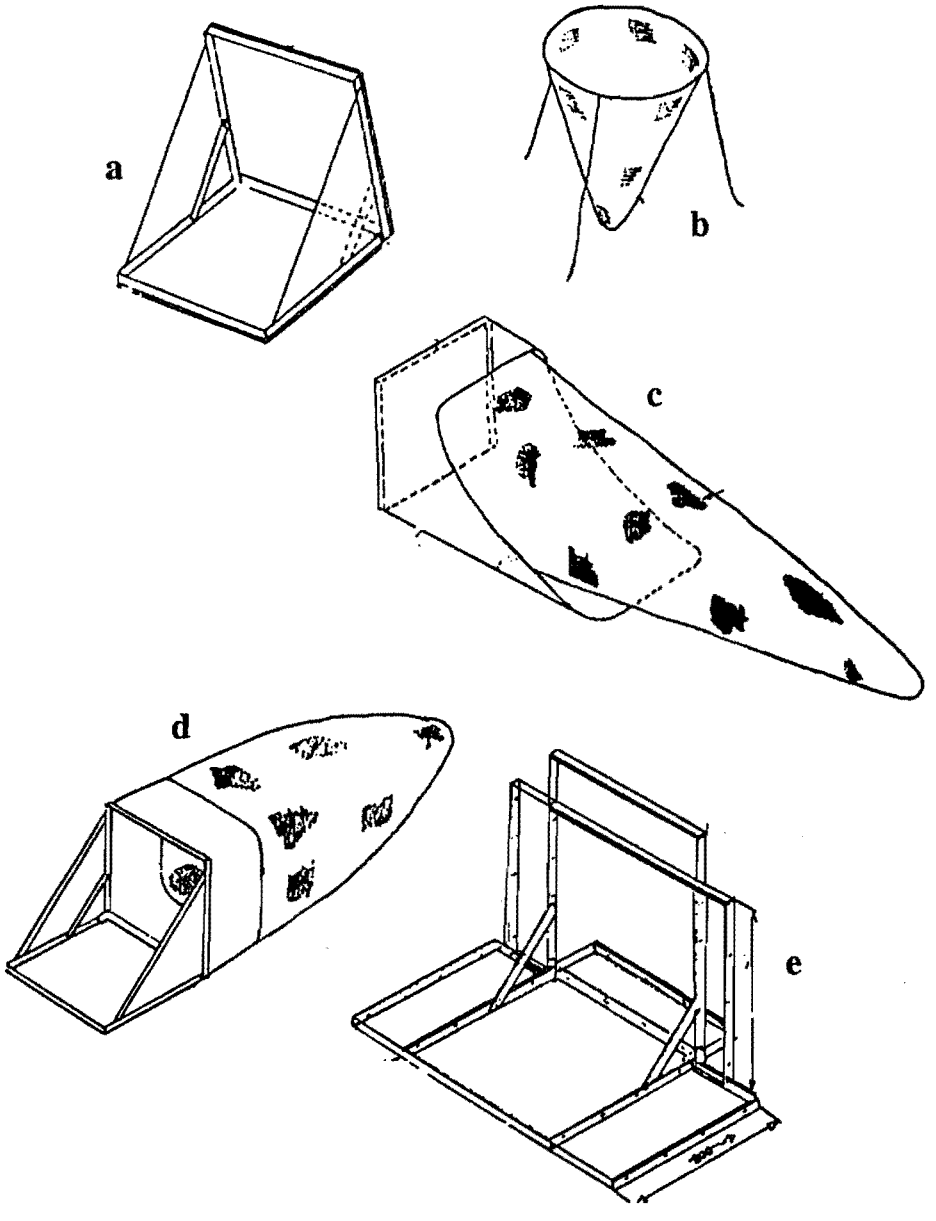

Fig. 8 - Collection Equipments for Aquatic Insects

ARACHNIDA AND ACARI

S. K. GUPTA*

Introduction

The Acari and other Arachnids occupy diverse habitats, as follows:

Soil mites: They occur abundantly in any soil rich with litter and organic debris and hence are common in forest soil, agricultural land, etc. Their population is rich in the upper strata of soil because of having richer organic matter in that layer. Therefore, the population decreases with the increase of depth. They are cosmopolitan including arctic and Antarctic regions.

Plant mites: A diverse group of mites occur in plants and occupy niches such as under surface of leaves, in the angles formed by veins as well as on upper surface, on twigs, crevices, of stems, auxiliary buds, under bark, within galls, inside malformed and deformed plant parts and on fruits as well. They are distributed world wide and belong to two major groups, viz. plant feeding and predators. There are few, which are fungivirous.

Water mites: These mites are found in all types of aquatic and semi-aquatic habitats which include lakes, ponds, swamps, marshes, creeks, springs, rivers, water soaked mosses and algae on the cliffs near the water falls and seepage areas. The water may be stagnant, running, fresh, salty, cold or hot. Many interesting species occur in hot springs as well as in Tundra pools in Frozen condition. The subterranean water mites are found in sandy soil or gravel soil, and soil near river edge. Most of these mites are free living in adult and nymphal stages, being effective predators and parasitic in larval stages.

Nest mites: A varied groups of mites and ticks occur in nests and nest debris of birds and mammals either as blood suckers, predators, scavengers, coprophagous, mycitophagous, etc. They are cosmopolitan in distribution.

Dust mites: A varied group of mites occur in nests and nest debris of birds and mammals either as blood suckers, predators, scavengers, coprophagous, mycitophagous, etc. They are cosmopolitan in distribution.

Parasitic mites: Some mites such as Macronyssidae, Trombiculidae, etc. occur on external parts of body of animals including man as facultative or obligatory parasites; pyemotids occur in haystacks as parasites of insects; *Cheyletiella parasitivorax* and many Listrophoridae occur in hair follicles; Myobiidae occurs at the bases of hairs, hair follicles and fur; Analgesidae and Syringophillidae on feathers of birds; Rhinonyssidae inhabit respiratory tracts of birds and reptiles; *Halarachna* occurs within nasal cavities of seal; Sarcoptidae and Psoroptidae occur within skin; Demodicidae occur within burrows of hair follicles; *Cytodites* occur within air sacs of birds, Listrophoridae inhabit subcutaneous tissues forming nodules while there are some which are found in urinary, intestinal and respiratory tracts of man and *Acarapsis woodi* occurs in respiratory tracts of honey bees. There are large groups of mites that infest invertebrates like insects, millipeds, centipedes and spiders.

Ticks: Most of the Ixodid ticks and some Argasid ticksare external parasites of birds, mammals and reptiles and some Argasid ticks are also found under bark.

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Spiders: These are cosmopolitan in habit, occur almost everywhere or near water, inside or in ground, from sea shore to top of mountains and also from desert to snow - clad mountains.

Collection

Plant Mites

Hand picking : Though it is strenuous but yet sometimes is profitable as varied groups of plant mites can be collected by this method. The infested leaves are directly examined under IOX lens in the field or under stereobinocular microscope in laboratory and mites are collected with a fine sable hair brush moistened with alcohol. A total of 30-40 leaves/ plant should be examined to have representative collection.

Sweeping: Low herbaceous plants, grass land, etc. when swept with a butterfly net, a large number of mites will be collected on the inner wall of the net and can be picked up by brush.

Beating: A white enamel tray with cotton pad on the inner surface is kept under the portion of plant from where the mites are to be collected. A total of 15-20 beatings/ plant part will dislodge the mites and after their falling in cotton, they get themselves entangled. Later, these are picked up by a fine brush.

Aspirating: A Singer type aspirator commonly used in insect collection may be used and plant mites, even if they are small, can be collected directly in the collection tube containing alcohol.

Brushing: Often the leaves/plant parts which are to be brushed are put into brushing machine and on its operation, the mites are brushed and dislodged into Vaseline coated plastic discs from where the mites are picked up by a fine sable hair brush. Each leaf is to be brushed forward and backward at least for two times.

Sweeping & teasing: Infected plant parts like inflorescence when scrapped or galls when teased, the mites come out and are collected.

Flotation method: Infested leaves are put in a jar containing water and teepol (a detergent) and shaken vigorously for 20 times which will dislodge all the mites and then filtered. The residue containing mites is taken out by a brush. Later, the same is taken in a watch glass and alcohol is added to it for separating out the mites.

Special methods for Eriophyids : The gently opened galls are put in a open glass jar in the airy room away from direct sun light. With the gradual drying of galls, the mites emerge out and start crawling on the inside wall of the jar which is previously wiped with glycerol 5 cm. below the mouth of the jar to avoid their escape. Later, previously warmed chloropicrin acid is poured over the dried plant parts and the jar is shaken vigorously. After the plants get settled, the liquid is decanted and filtered for getting the mites.

Water Mites

Dipping: This method is very frequently used for collection of water mites. The collection tools like water enamel bowl and wide mouthed pan with long handle are immersed in water and lifted with water in quick succession at least for 20 times. The water sample is observed under a binocular microscope and the mites are collected.

Hand picking: The parasitic and aquatic mites can be collected from the hosts by direct by examining it with a hand lens and collecting with a brush.

Netting: Nylon nets of narrow mesh fitted with an iron ring with a long handle are very useful tool for collection of free living mites. Another type of net is the Berge net which is a funnel shaped net, with a fine sieve fitted with a long wire enabling to put the net into deep water. The contents of Berge net are examined in white porcelain tray.

Soil Mites

Direct method: Sample of soil, soil litter, etc, are taken in a white enamel tray and examined under a stereobinocular microscope. The mites are picked up by a fine sable hair brush.

Heat desiccation method: Soil samples with rich organic matter and litter with plant material may be collected in polythene bags. The mouths of the bags should be kept open to allow the excess moisture to evaporate away. The samples are put into a Tullgren's apparatus containing a battery of funnels with light source (40W bulb) above and a collecting tube containing alcohol fitted with the stems of the funnels. The samples are put into a sieve fitted inside the funnels in upside down position. With continuous heating for 2-4 days depending upon the moisture content of the soil samples, a large number of mites will be seen to have been collected in the collection tubes.

Flotation method: The soil sample is stirred in $MgSO_4$ solution of high specific gravity (1:2) and at the same time a stream of air is bubbled into the suspension from below. After settling, liberated animals and the organic matter float to the surface and are collected on a filter paper or on a sieve. A slight modification of this method has been done by introducing primary washing and sieving to remove stones and coarse plant material and by introducing the benzol water method for partially separating the animals from the plant debris in the "float". The float is vigorously shaken with a mixture of benzol and water in a flask, the animals accumulate above the plant material on the benzol-water interface.

Stored product mites, Nest mites, Dung mites, and Litchen mites: The heat desiccation as described earlier for soil mites may be used for collecting this group of mites also.

Dust mites : The dust mites are collected in the following stages: (a) first of all a small quantity (2 gms) of dust sample is washed with kerosene oil in a beaker and stirred with a magnetic stirrer for about 15 minutes at slow speed. The liquid is filtered through a 500 micron sieve, (b) the material on the sieve is washed in a beaker by kerosene oil jet and filtered through 500 micron sieve and the filtrate is added to first. (c) filtrates of (a) and (b) containing fine dust with mites in kerosene oil sedimented and the supernatant is filtered through Buchner funnel, (d) the sediment washed with kerosene oil and carbon tetrachloride (1:3) (sp. gr. 1.4) and centrifuged, then filtered. Lastly, the sediments is mixed with para carbon tetrachloride (sp.gr. 1.6) and the process is repeated. However, most of the mites are collected in steps (c) and (d).

Parasitic Mites

Direct examination: The infested animals are directly examined under a microscope or keeping the host for 2-4 days in a screen cage over a pan of water. As the parasites are not obligate, they will drop from the host after feeding. Parasites can also be collected by combing or brushing of hosts.

Flushing: Intranasal mites are recovered by flushing nasal cavities with a stream of water under high pressure. The tip of 20 gauze needle is to be cut off 2 mm from the base. The end is grounded to a smooth rounded tip. The dead animal is firmly grasped by the throat in order to close the oesophagus or trachea. The needle is attached to a 5 ml syringe filled with water and introduced into one of the nasal passages and is collected as it comes out from the other nostril. Many mites may be collected from the extracted fluid.

Autopsy: Splitting of the bills of dead host between nares also facilitates the recovery of mites. Nasal cavities of dogs are examined by submerging the carcasses of birds, small rodents, etc. in soap solution and then shaken vigorously. The mites leave the hosts and can be collected from the decanted washings. In case of repellent, the host is put in a cylinder with head and neck protruding through a hole in the upper lid. Chloroform fumes or dry dye or pyrethrum is pushed inside the cylinder and the bird is encouraged to flutter. All the mites drop off on a white paper kept at the bottom of the cylinder.

Applications of detergents/repellents: Ectoparasitic acari may be collected by submerging the carcasses of birds, small rodents, etc. in soap solution and then shaken vigorously. The mites leave the hosts and can be collected from the decanted washings. In case of repellent, the host is put in a cylinder with head and neck protruding through a hole in the upper lid. Chloroform fumes or dry dye or pyrethrumis pushed inside the cylinder and the bird is encouraged to flutter. All the mites drop off on a white paper kept at the bottom of the cylinder.

Application of acaricides: Parasitic mites may be recovered from the skin of dead hosts by Hopkin's dissolution method. Fresh or dry skin pieces should be kept in 5-10% KOH/NaOH solution over a water bath till hairs dissolve completely. The contents of the beaker is then filtered while hot through fine stainless steel gauge. The solid residue in the gauge is then washed well in a petridish and examined for parasites. In a slightly modified method, the residue is treated well with $ZnSO_4$ solution. Parasites float on the surface of the solution and can be easily collected.

Light trap method: This is used for free living mites attracted to light.

Ticks: These are collected by directly examining the hosts and picking the ticks by forceps. Before doing that it will be better if the host is first brushed with glycerol or paraffin which will relax the ticks and prevent their damage. The unattached ticks can also be collected from grassy land by flag dragging or sweeping.

Spiders: The collection of spiders is very easy. One of the commonest methods of collecting in large numbers is by sweeping over bushes, grasses, etc. with a sweep net and picking out the spiders by hand. Another effective method is to hold an inverted umbrella underneath the bushes and to shake the plants vigorously. Spiders along with insects and mites will fall on the umbrella.

Scropions: These are usually collected by long forceps.

Horse shoe crabs: These are collected by fishermen's nets from sea.

Opiliones: These are collected by hand picking.

Pseudoscropioins: These are collected by direct picking by forceps and from soil, debris etc. by heat desiccation method.

Preservation

Liquid preservatives: Usually all the Acari and other Arachnids are preserved in 70% ethyl alcohol in a suitable vial. In case of Acari, a few drops of glycerine may be added to it to avoid drying of specimens. Often Oudeman's fluid (70% alcohol - 87 parts, glycerol - 5 parts, lactic acid - 8 parts) or Keonike's fluid (glacial acetic acid - 10 parts, glycerol - 50 parts, distilled water - 40 parts) may be used as preservative for Acari. the mites/ ticks are usually kept in tubes which are put in a big jar in inverted position and the jars should contain enough alcohol to keep the tubes immersed.

Eriophyid mites are not preserved in alcohol but are better preserved in dry condition by wrapping the infested leaves/galls in tissue paper and from where the mites are brought into normal condition by simmering in Keifer's preparatory solution (Resorcinol - 50 gms, diglycolic acid - 25 ml + some iodine).

The spiders, Horse- shoe crabs, Opiliones, scorpions and pseudoscorpions are preserved in 70% alcohol.

Slide Preparation

Temporary: The temporary slide preparation is usually done by keeping the specimens in a drop of lactic acid on a microslide and a small broken piece of coverslip is put over it. The slide is then gently warmed which will help in better orientation with stretched appendages. For small and soft bodied specimens, glass bits may be kept around the specimens and the coverslip should be put over it to avoid direct pressure of the coverslip on the specimens.

Permanent: Among the several mounting media which are used for mounting of mites, Hoyer's medium (distilled water - 50 ml, gum arabic - 30 gms, chloral hydrate - 200 gms., glycerol - 20 ml.) and Heinze's medium (polyvinyl alcohol - 10 gms, distilled water - 40 ml, lactic acid - 35 ml, glycerol - 10 ml, phenol aqueous solution - 26 ml, chloral hydrate - 100 gms) are commonly used.

For animal and plant parasitic mites, the correct identification of the host is most essential.

Unless otherwise desired by specialists, the specimens should better be sent in alcohol preserved condition enabling the specialists to mount the specimens in desired position for identification.

Quantitative Method of Sampling

For most of the arachnids and many acarines no standardised technique is available. For plant mites, the techniques which are available are described below:

Direct counting: This is commonest of all methods. The sampled leaves numbering 30-40/plant are put into a petridish containing chloroform soaked cotton to immobilise the mites. Later, the leaves are put under stereobinocular microscope one by one and the mites are counted from the entire leaf or from a definite leaf area (often 2.5 sq cm area) and from a definite leaf area (say basal, middle and upper part of leaf) depending upon the intensity of population and dispersion pattern. Some authors found that comparing the plotted values based upon actual counting of mites with the number of leaves having no mites, one can have a rough estimate of mite population.

Imprinting method: A Whatman filter paper No. 1 is put on the under surface of the leaf and a wooden roller is rolled rapidly with pressure sufficient enough to crushed leave characteristic stains on the filter paper making permanent record of the population. Later the spots are counted. Experienced persons can differentiate the stains of adult and eggs.

Flotation method, Jarring method and Brushing Method: Already discussed under Collection method.

Sampling of predatory Mites: By and large, the above methods will also hold good for predatory mites. However, some predatory mites inhabit twigs and crevices of stems. Therefore, only sampling of leaves may not be enough and sampling from other plant parts may be necessary. This can be done by dislodging them or by suction methods. Also without disturbing the predators, a visual scan over a certain number of plants and trees, if made, for a desired period of time, a reasonably clear picture about the population of predatory mites can be had.

Sampling of Soil Mites: Normally a core size of 5 cm diameter and 5 cm depth is taken out by a soil corer and the same is extracted to get the quantitative data on soil population.

Sampling of Ticks: This is done by flag dragging for a definite period (1-2 hrs.) covering a unit area and by adopting this technique a fairly clear picture of population of ticks will be available.

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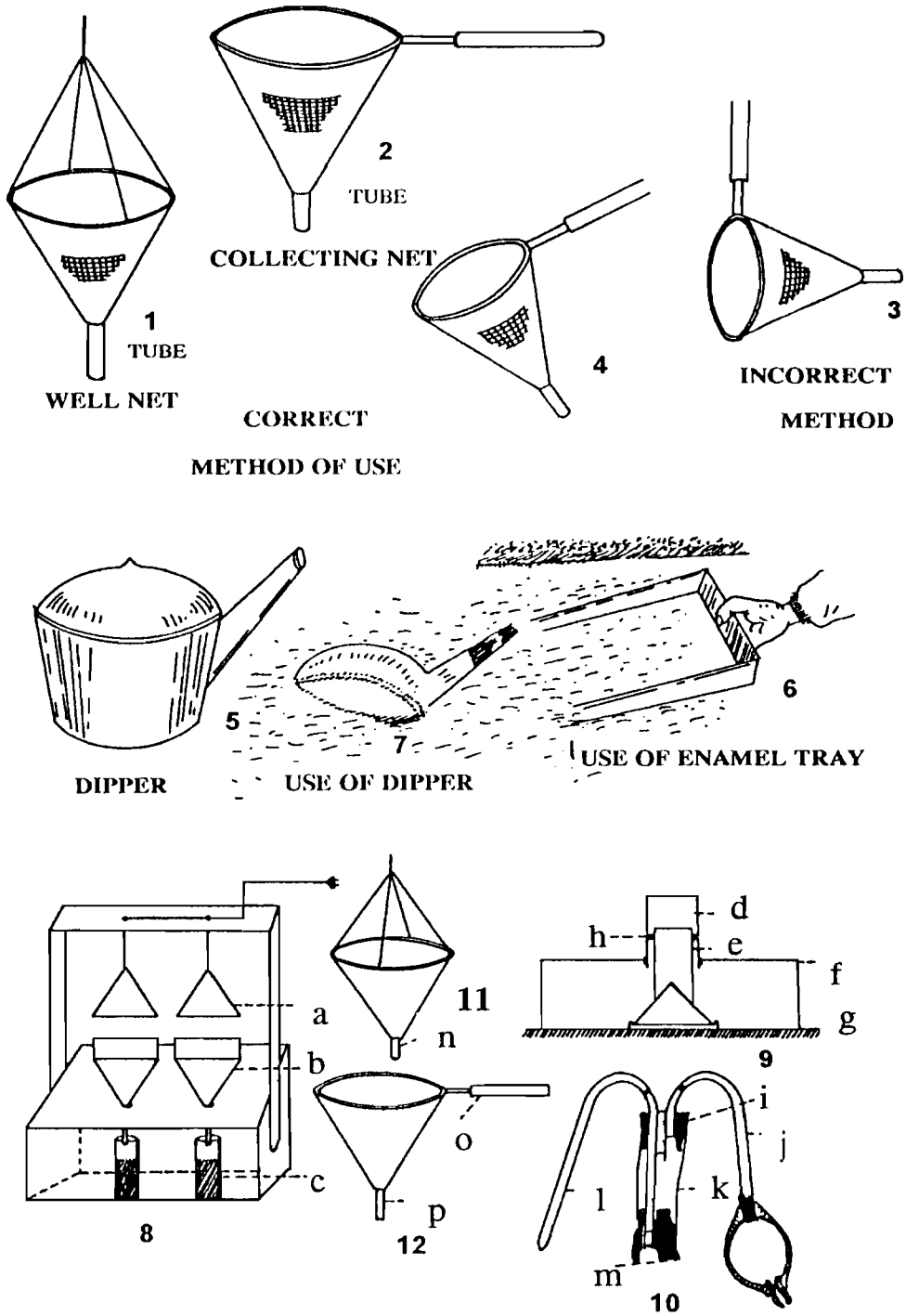
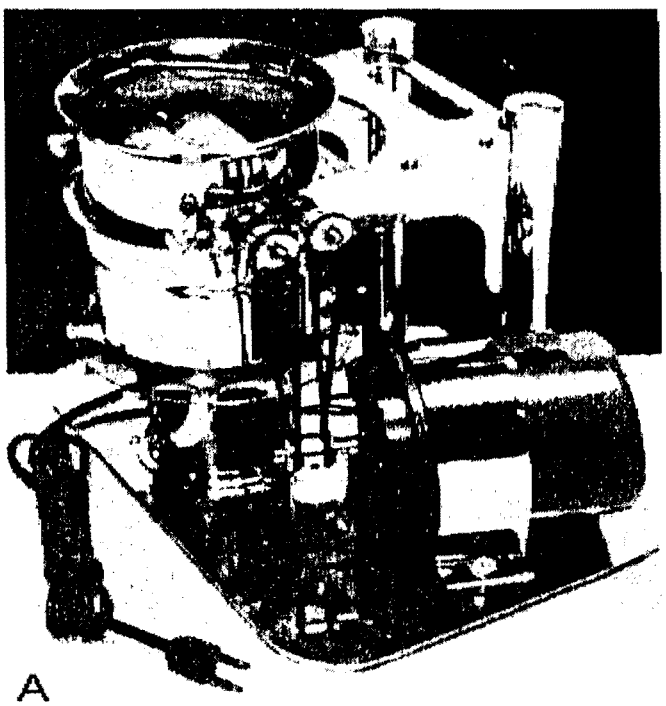


Fig. 9 - **Collection Equipments for Acari**



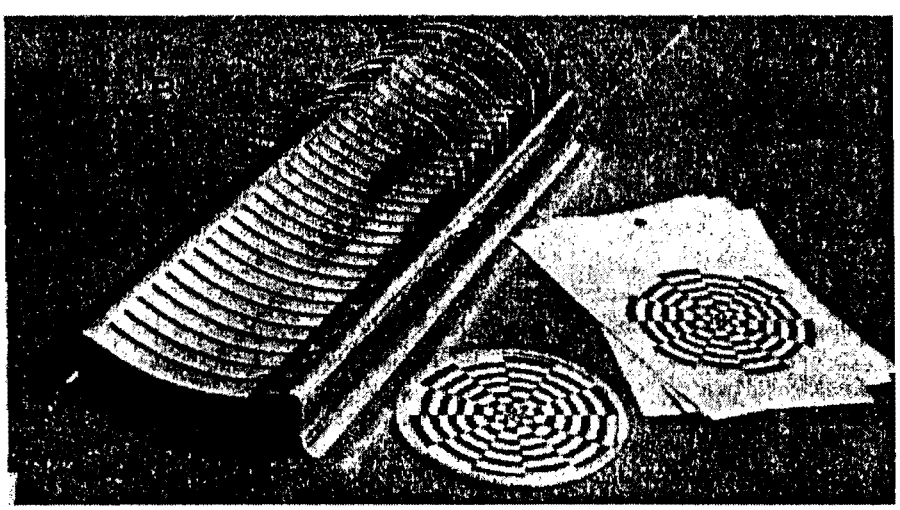


Fig. 10 - **Mite Brushing Machine**

CRUSTACEA

P. KRISHNAMOORTHY**

Introduction

Crustacean habitats include benthic environments of the coastal, deep oceans and estuarine regions. In addition, they also inhabit semiterrestrial, terrestrial and freshwater environments of the ponds and lakes. They also live in pelagic environments of the temporary ponds and permanent lakes and in the epipelagic to bathypelagic zones of the oceans.

Crustaceans have adapted themselves to live in a variety of ecological conditions. They can be broadly grouped under marine, freshwater and terrestrial crustaceans.

Marine

Crustaceans are more abundant in marine environment than in freshwater or on land. Majority of the crustaceans occur in the littoral and shallow waters. Most crustaceans found in this habitat are either sedentary forms which live permanently fixed to the ground or burrowing forms. Various species of isopods, crabs, anomurans and alpheids live on and under the rocks and boulders as well in the crevices. The diversity and abundance is more in the rocky intertidal zone along the coastal and in the coral reef ecosystem. Most of the burrowing forms live in the sandy substratum. Sandy coast also support a variety of small microscopic interstitial fauna. In addition, large number of the brachyuarns, macruarans and anomurans occur between the lowest low tide level to 100 m depth of various substrata. Crustceans also occur in the pelagic region. It is however, copepods which form the major group in this region.

Freshwater

In fresh water the crustaceans are more abundant in the littoral regions with a variety of macrophytes. Temporary ponds harbour a variety of small sized zooplankton community in which Cladocera is the dominant group. Freshwater crabs and prawns are found in the hillstreams, rivers, ponds, tanks and also in paddy fields. In addition, amphipods and isopods also live in this squatic habitat.

Terrestrial

Despite great diversity in morphological features, only a few terrestrial forms have also far evolved and among them the isopodgroup, viz. Oniscidae and Porcellionidae, have become completely terrestrial. It is found under the leaves, flower pots, bricks and stones, in the garden and among the decaying vegetable matter.

Collection

The usual instruments like hammer, chisel, scalpel, large wooden forceps, sieve, shovel, spade and hand net are required to collect the intertidal organisms. Two kinds of collecting gear i.e. push - nets for collecting shrimps and other organisms from grass flats and a yabby pump for borrowing decapods are used in the intertidal region. For collecting shallow and deepwater benthic forms,

** Zoological Survey of India, Madras.

various types of dredges, trawls and nets are used. The floating planktonic crustaceans can be collected by different types of plankton nets and recorders. These equipment's are to be operated from mechanized boats with hauling facilities. Terrestrial and parasitic crustaceans can be collected with simple instruments like forceps, knife, mounting needle and brush.

Table - 2. Habitats of Crustacea

Group	*Habitat*
BRANCHIOPODA	
Notostraca	Freshwater
Cladocera	Freshwater (95%) Brakishwater (3%) Marine (2%)
Conchostraca	Freshwater
Anostraca	Freshwater and Inland Salt water
Copepoda	Marine & Freshwater
Cirripedia	Marine & Brackish water
Ostracoda	Marine & Freshwater
MALACOSTRACA	
Stomatopoda	Marine
Amphipoda	Marine, Freshwater and Terrestrial
Isopoda	Marine, Freshwater and Terrestrial
Cumacea	Marine
Decapoda	Marine, Freshwater and Terrestrial

Sampling

Aquatic

Most of the marine forms inhabit the littoral region in the sea. During low tide many slow moving intertidal forms can be collected easily with a forceps. A flat stone or a bunch of weeds suddenly turned over will generally reveal a variety of crustaceans. Vigorous rinsing of sea weed tuft in a tray containing few drops of formalin will make these animals to come out and they can be easily picked out with a small forceps or by a pipette. Rockpools harbour rich fauna of decapods and stomatopods and these are collected with the help of a small hand net. Isopods and other small decapods live in narrow crevices of rocks and these can be made to come out of their hideouts by squirting in a week solution of formalin or by dropping a little bleaching powder. Small shrimps and other crustaceans living on attached vegetation in shallow water are collected with a push net. Many borrowing forms can be collected by digging out the sand. The sand is put into a sieve and filtered leaving the animals

in the sieves. Small burrowing macrurans are collected with the help of a yabby pump. By this pump the deep burrowing animals are removed without much damage. Most of the crawling animals like prawns, crabs and stomatopods which live near or at the bottom in shallow waters are collected by various type soft dredges and trawls. For collecting planktonic organisms different types of plankton nets are used. These equipment's are to be operated from mechanised boats with winch facilities.

A small type of Naturalist dredge is used to collect the bottom fauna present in lakes and rivers. This can easily be operated from a small boat. If it is a small stream, bottom sample can be obtained with a shovel and by sieving it, benthic animals are collected. For collecting the planktonic forms, an ordinary plankton net of small mesh size is used. The week inhabiting cladocera and ostracoda may be collected with a hand net by sweeping through the weeds.

Terrestrial

Terrestrial isopods which live in damp places in the soil under decaying vegetation are collected by digging the upper layers of the soil with a shovel and by examining the under surface of stones. The isopods may be picked individually with a forceps or with the help of a brush moistened with alcohol.

Parasitic

These are found mostly in the eyelids, nasal, cavities, buccal cavity body surface and fins of fishes. These parasites can be removed from their host's body with the help of fine forceps, camel hair brush or by a needle. For proper identification it is better to collect the parasites with the hosts.

Preservation

Before preservation, the animals should be washed thoroughly to remove the dirt and unwanted material attached to it. Preservation of specimens it is carried out in three stages: Narcotisation, killing and preservation.

Narcotisation makes the animals less sensitive so that they will not be damaged or distorted by violent contraction, when killed. Some of the standard narcotising agents are magnesium chloride. Menthol, Chloral hydrate and Alcohol. The animals are kept in a container with seawater and a small quantity of any of these narcotising agent is added gradually at frequent intervals. The time taken and quantity of the narcotising agent to be used, depend on the size of the animal and quantity of sea water.

Once the animal become unresponsive, a proper killing agent is added, after draining out the narcotising agent. Formaldehyde is the best fixative for most crustaceans. The time for which they must remain in the solution varies with their size.

Permanent Preservation

The fixed organisms are thoroughly washed, treated with different grades of ethyl alcohol 30% to 90% and finally preserved in 70-90% alcohol. Planktonic crustaceans are best preserved in 4-5% formalin. The specimens after preservation and labelling are stored in tubes or jars depending upon the size of the specimen. If the specimens are kept in tubes, the tubes should be plugged with cotton and stored upside down in a jar filled with sufficient quantity of preservative. For specimens stored in spirit, corks should not be used as they discolour the spirit and specimens. If the samples are to be stored in formalin, periodic checks of pH must be made for every 3-6 months. If the pH of the preservative is found too less, 0.2 grams of Borax should be added for every 100 ml of preservative.

With the exception of certain parasites that require histological studies for identification, taxonomic identifications are mainly attempted on external morphological characters. The stains most commonly used in mounts of crustacean appendages or whole specimens are lignin pink, mallory's acid fuchsin red, fast green and chorazol black.

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MOLLUSCA

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Introduction

Molluscs are broadly classified into marine, freshwater and terrestrial or land forms. In the molluscan biodiversity, marine species are at the top. Out of 290 families represented in India, 242 families are found in marine habitat, 22 freshwater and 26 land families respectively.

In the marine ecosystem, the littoral region is very rich in molluscans. Coral reefs offer rich fauna. The population in marine environment is mainly controlled by salinity, temperature, tides and water currents.

In freshwater ecosystem, the mollusc populations are controlled by the water level, temperature, vegetation, nature of the soil etc. During summer when the ponds dry up, molluscous bury themselves into the soil and hibernate. Monsoon is the best season for collection of these animals.

In terrestrial ecosystem the snails prefer shady, moist and damp places and under surfaces of the stones and under litter.

A conservative estimate includes 66,535 species for the world. India has 5,070 species at present (7.62%). Out of seven Classes present in the world, India is represented with five Classes. Out of 462 families in the world, India is having a total of 290 families.

Collection

Equipments

The following equipment's are necessary for collection of molluscous:

Forceps, scalpel, shovel or spade, knife, hammer, chisel, brush, petridishes (assorted), specimen tubes (assorted), jars of either glass or polythene (assorted): sieves of different mesh size, enamel trays, enamel bowls, plastic bucket, metallic frame either rounded or polythene (assorted); sieves of different mesh size, enamel trays, enamel bowls, plastic bucket, metallic frame either rounded or square (for quantitative studies), polythene bags, gum-boots, rubber gloves, field note book, labelling paper, suitable net, thermometer etc.

Quantitative Method of Sampling

To study the population the following techniques are used:

1. Quadrate method: by using the metallic frame of 1 sq.m. which is dropped in the area of study and snails collected are counted.

* Zoological Survey of India, Madras.

2. Standard scoop or dredge method: By scooping or dredging of a standardise scoop or dredge is passed over the required area and number of snails collected are counted.

3. Counting per unit time: By number of snails collected systematically with sieves by one or more trained collectors in a measured area.

4. Palm leaf method: By placing palm leaves at a regular interval along the shore or bank, after exposing for certain period of time, remove the leaves and snails attached on it are to be counted.

Preservation

Aquatic: Animals collected are placed in an enamel tray filled with either sea water or pond water as the case may be, making sure that the specimens completely immersed in water. The surface of the water to be sprinkled with fine powder of Methanol or Magnesium sulphate or Chloral hydrate and covered it by lid. Narcotisation takes place depending upon the nature of the specimens. It may vary from 2 to 24 hours. After 12 hours the collection should be examined to ensure that decomposition has not taken place. When completely narcotised, the specimens lie in fully extended condition. Then add either 4% formalin or ethyl alcohol in drops of 3-4 at a time, repeat the process every 1/2 hour until the animal dies. This method is good for nudibranchs. Larger forms like cephalopods, especially opisthobranchs like sea-hare etc. should be injected with a fixative by the help of a syringe.

Freshwater snails and bivalves can be relaxed by plunging them into boiling water. 0.5% solution of Propylene Phenoxetol is good for relaxation.

Land: Asphyxiation' is the best method to kill the land snails and slugs. A glass jar or bottle is fully filled with water and specimens are to be plunged into it and the mouth of the container is closed with lid making it air tight. Add deoxygenated water or few drops of spirit at intervals may help in killing. The specimens may be extended after 20 to 24 hours. Thus narcotised specimens are to be washed in running water to remove the mucous secreted by them. When the animals die or are completely motionless, they should be treated in ascending grades of alcohol and finally preserved in 70% alcohol. Slugs are to be preserved in 4% formalin.

Specimens needed for anatomical studies etc. are to be fixed either in Bouin's fluid or Alcohol - Formalin - Acetic Acid (AFA) combination. It may take 20-24 hours and then remove and washed thoroughly. It is better to preserve some dry shells separately. The animal can be removed with the help of a bent forceps and boil the shells or remove any remaining parts. These are to be cleaned with a brush and dried in the air. In case of operculates, operculum should be kept closed by pasting it into a cotton plug inserted into aperture.

Marine: Shells like cones, cowries, olive shells, moon shells, which are highly polished shall never be plunged directly into bodied water. By burying the shells into soft dry sand and leave them intact for a week or so yield good results.

Packing: Dry shells are to be packed with sufficient cotton padding to avoid damage and the boxes are to be labelled.

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LOWER INVERTEBRATES

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Introduction

Ecologically lower invertebrates may be broadly categorised as follows:

1. Freeliving
   - i. Freshwater/estuarine/marine water forms:
   - ii. Soil inhabiting forms;
   - iii. Moss/sphagnum/plant inhabiting forms
2. Parasitic
   - i. Ectoparasite
   - ii. Endoparasite
     - a. Lumenicolous
     - b. Histozoic
     - c. Coelozoic

Habitats

1. Freeliving forms

Freeliving Protozoa and Nematode live in every possible niche of all habitats from deepest ocean beds to highest mountain tops and from hot springs to Antarctic snows. However, amongst Platyhelminthes only one group, viz., Turbellaria, are principally freeliving while the rest i.e. Monogenea, Trematoda and Cestoidea are exclusively parasitic.

For freeliving lower invertebrates following habitats are likely to be valuable and rich sources of material.

- i. Any fresh water body with sufficient vegetation, rich decaying organic matter, scum, debris, bottom ooze, etc.
- ii. Estuarine and marine waters and their bottom ooze;
- iii. Almost every type of soil in every kind of environment from pit rich soil of bogs to dry sand of desert;
- iv. Sphagnum, mosses, moist stones, roots and shoots.

2. Parasitic forms

- i. Entire outer surface of hosts, e.g., oral region, gills opercula and fins of fishes;
- ii. Tissues and organs of vertebrate and invertebrates hosts;
- iii. Gut contents, organ smears, faecal matter, etc.;
- iv. Plant roots and shoots

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Collection

Equipments

In addition to conventional laboratory articles, such as glass vials, specimen tubes and jars, petri dishes, beakers, culture dishes, embryo flask, finger bowls, micropipettes and rubber teats, dissecting box, spirit lamp, chemical balance, measuring cylinder, filter and tissue papers, glass marking pencil, etc., following items are specially required: microscope, stereoscopic binocular, centrifuge with buckets and tubes, microslides, cover slip (No.1 thickness), cavity block, coplin jars, slide box and cabinet, hot plate, autoclave, incubator, microtome; microsiever, Bearmann funnel, plankton net and dip net (for collection of freeliving Protozoa).

Sampling

1. Freeliving Forms

a. Protozoa

i. Freshwater: Water samples are to be collected in wide mouthed glass jars, keeping some algae, water weeds, flocculent matter and bottom ooze. Qualitative sample collection may be conveniently made by keeping only small amount of vegetation in a sampling jar and filling up remaining part of the jar with water of the surrounding area. Water samples with vegetation and bottom ooze collected from different parts and depths of water body are to be kept in different jars for collecting maximum number of species. Plankton nets and sieves of different mesh sizes can also be used for sample collection.

After bringing the sampling jars in the laboratory, lids are to be immediately removed and the jars should be placed near a well-lighted window. The, some Protozoa can be collected through micropipette in a few hours from the side of the jar facing the strongest light. But, only after 24-28 hours many species of Protozoa can be collected, since then only they become concentrated near the top and near the edges of leaves, stem and roots of vegetation kept in the jar. Some Protozoa, more particularly testacid rhizopods are mostly available in the bottom ooze. Samples are to be examined regularly under microscope for three to four weeks since Protozoa occur in succession and one species abundant and dominant one day may be very meagre or absent next day.

ii. Marine: Samples are to be collected by plankton nets, towing slowly behind the boat or by sampling bottles. Protozoa occurring in them are to be extracted following earlier mentioned methods. Some samples may be preserved in 4 per cent formaldehyde solution in the field and brought to the laboratory for collection of Protozoa.

iii. Soil: Soil samples are to be taken from different habitats making 12-17 cm deep boring. Samples are to be mixed, sieved through 3-mm mesh size and stored in sterile bottles. The, samples are to be brought to the laboratory for microscopic examination and/or culture.

iv. Moss inhabiting: Moss samples from different habitats are to be collected in polythene bags and brought to the laboratory. A portion of each sample is to be kept in petri dishes or in 50 ml/100 ml glass beakers and sprinkled regularly with distilled water. After 24-28 hours aqueous drops are to be drawn through micropipettes by squeezing the moss sample, kept on microslides and examined under the microscope. The Protozoa occurring in the sample are to be fixed and preserved. Usually moss samples need processing within a few days after bringing them to laboratory. However, if these samples can be stored in cool places preferably in a refrigerator at 5° C some Protozoa may be recovered even after one year.

b. Turbellaria (Platyhelminthes)

Turbellarians can be collected by hand picking or by water net since they are mostly found under stones in ponds and streams, on aquatic weeds and on damp soil.

c. Plant and Soil Nematode

i. Collection: Soil samples from around host plants are to be collected with the aid of shovel or any sharp cutting tool from a depth of 5-15 cm in most fields and from more depth in semi dry and dry fields. It is better to collect 4-5 subsamples to form a bulk sample. Plant roots are also to be collected for extraction of plant nematodes. These samples are to be kept in polythene bags whose open ends are to be covered with a rubber band. In the field these bags are to be stored in a cool place out of direct rays of sun. In the laboratory these samples are to be stored in a cool place out of direct rays of sun. In the laboratory these samples are to be stored in refrigerator at about 5°C until processed. Processing of samples should be done as soon as possible.

ii. Processing: A combination of sieving and recondition technique and Bearmann funnel technique is discussed as follows :

Soil sample measuring 500 gm is to be placed in bucket 'A' of 15 litre capacity, filled up to one-third of its volume with water. Soil and water in the bucket are to be thoroughly mixed by hand to make a homogeneous suspension. Then the bucket is to be left undisturbed for 20-30 seconds to allow heavy soil particles to settle down. This suspension is to be filtered through a coarse sieves (2 mm pores) and collected in another bucket 'B'. The entire process is to be repeated 2-3 times.

The suspension in bucket 'B' is also to be made homogeneous by hand and to be allowed to settle down for 10-20 sec and then to be passed through a sieve (if necessary, a set of 2 sieves) of 300 mesh size with 53 μ pores where nematodes will be retained. The nematodes and the fine silt caught on the sieve are to be washed off and to be collected in a beaker of 250 ml.

The aliquot collected in the beaker is to be poured gently on moist double tissue paper placed on a small supporting coarse sieve (with 2-mm pores) which is to be placed into a modified B Bearmann funnel. The funnel must hold sufficient water to remain in contact with the surface of the sieve. A rubber tube of suitable length and diameter is to be attached to the stem of the funnel and to be closed by a string

of screw clip. The funnel is then left undisturbed for 24 hours so that most nematodes pass through the tissue paper and settle at the bottom of funnel stem. The nematodes are then collected in a test tube by loosening the grip of the clip.

Root - shoot samples: Infested roots and shoots are to be kept in a refrigerator at 5° C to 10° C and examined as early as possible. Roots should be washed gently to remove as much soil as possible with the aid of scissors. A few such pieces are to be placed in open petri dish and teased apart with stout needles. Nematodes released from the plant tissue float out and can be collected in a cavity block with the aid of handling needles.

Chopped pieces of soft roots may be placed in a blender, operated electrically for 1-2 min. Then nematodes are to be collected by using decanting and sieving technique explained above.

2. Parasitic Forms

Entire outer surface of hosts (e.g oral region, gills operculas and fins of fishes) is to be thoroughly searched for ectoparasites or encysted endoparasite. Then the host animal is to be sacrificed and its different organs are to be cut open for the collection of endoparasite.

Before removing viscera body cavity and coelomic fluids are to be examined with the aid of microscope/stereoscopic binocular for parasites or their cysts. Subsequently all internal organs are to be removed and each organ is to be placed separately on petri dish/suitable container in normal saline. Then each organ is to be thoroughly searched under light microscope for parasites. Gut contents, faecal matter, blood films and organ smears are also to be examined.

Preservation

(A) Field Techniques

a. Protozoa

i. Freeliving, lumenicolous and coelozoic forms :

Fixation: The most commonly used fixative is Schaudinn's fluid (cold saturated mercuric chloride 66 ml, absolute alcohol 33 ml, glacial acetic acid 1 ml). Before fixation one small drop of sample is to be poured on the middle of the slide by means of micropipette. Protozoa to be fixed should be observed under the microscope. When the sample becomes semidried and specimen becomes almost motionless with its normal shape then one or two drops of Schaudinn's fluid are to be poured on the sample and kept for 5-15 minutes. After fixation it is necessary to wash the slide with 70 per cent alcohol to remove all traces of mercuric chloride present in the fixative.

The other fixative commonly used are Carnoy's fluid, Bouin's fluid and Zenker's fluid.

Staining and differentiation: Heidenhain's iron-haematoxylin is the most suitable and widely used stain. It requires a mordant, ammonia-ferric sulphate (iron alum) and a dye haematoxylin. To use this stain the fixed specimen/smear is to be brought to water through descending series of alcohol (from 70% to 30% and then water), kept in 3 per cent iron alum for 3 hours or more, rinsed in distilled water and differentiated (destined) in 1 per cent iron alum under microscope until the proper intensity of colour is reached. Then it is to be kept under running water for 5 minutes and gradually dehydrated.

Dehydration and mounting: Dehydration is done through ascending series of alcohol (30% - 70% - 90% - absolute alcohol). The slide is then cleared with xylol and mounted in a neutral medium, such as DPX or Canada balsam.

ii. Histozoic and coelozoic forms

The blood films and smears of different organs of host animals are to be drawn on grease free microslides, air-dried and brought to the laboratory for staining and further study. Facial samples of different boasts are to be collected in a vial containing 2.5 per cent potassium dichromate for coccidian parasites. For histological preparation suspected organs are to be fixed in Bouin's fixative and brought to the laboratory for further processing.

b. Platyhelminthes

i. Freeliving forms (Turbellarians)

The specimens are to be kept in petri dish with some water. A few crystals of menthol, chloral hydrate or camphor are to be sprinkled over water for Narcotisation and relaxing of specimens. Such effect can also be achieved by keeping the specimens in 1 per cent nitric acid solution for a few seconds. After the specimens have relaxed, water is to be drained off and the fixative is to be poured gently over specimens and left for 24 hours. Small specimens can be fixed by pressing them between two slides which are to be tied with thread and left in fixative for 24 hours. Then the specimens are to be washed and stored in 70-90% ethyl alcohol.

A.F.A. (Absolute alcohol 90 parts, 40% formaldehyde 5 parts and glacial acetic acid 5 parts) is the commonly used fixative.

ii. Parasitic forms

These animals usually contract and become thickened during fixation. To avoid this, it is preferred to place them between two slides or under coverslip, depending upon the thickness of the worm. The slides are to be suitably pressed, tied with threads and kept in appropriate fixatives. Pressing of specimen should be done in such a way that there is not much distortion of shape of parasites and normal position of their internal organs should not be disturbed.

Small specimens may be killed by shaking them vigorously in hot water or in 4% hot formalin. Specimens killed in hot water are to be put in fixatives whereas formalin killed specimens need not to be placed in any other fixative. Fixatives used are hot Bouin's fluid, A.F.A. and 3-5% formalin.

c. Nematoda

i. Plant and Soil Forms:

The suspension containing nematodes is to be placed in a test tube and left undisturbed for 2-3 hours. Then most of the water is to be removed carefully from the tube and nematodes settling at the bottom are to be placed in fixative for at least 24 hours. Nematodes may also be stored in fixative for longer period. F.A. (40% formalin 10ml, glacial acetic acid 2-5 ml and distilled water 100ml) is very efficient fixative for plant and soil nematodes.

ii. Animal parasitic Forms:

These may be fixed either in hot 70% alcohol or in 3-5% hot formalin. The fixed material may be stored in tubes filled with fixative mentioned above.

(B) Permanent Preservation Techniques

Protozoa and nematodes from soil/moss samples brought to the laboratory are to be extracted for specimens as soon as possible following standard procedure elaborated earlier. Blood slides and organ smears of Protozoa parasites prepared in the field are also to be fixed and stained for the preparation of permanent slides. Permanent slides of parasitic Protozoa, Platyhelminthes and Nematoda are to be prepared as follows:

Parasitic Protozoa: Histozoic and Coelozoic Forms

Air dried blood films brought from the field are to be fixed in acetone-free 100 per cent methyl alcohol for 5 minutes and then allowed to dry. Then, Giemsa's stain is to be used for staining blood films. One drop of stain is to be diluted in 1 ml of 7.0 - 7.2 pH distilled water or buffer. The stain is to be poured on slides with smear side up, placed on a staining rack and left for 45 minutes. Then, the slide is to be washed with neutral distilled water or buffer solution and dried. Unmounted slides are to be wrapped in wrapping papers and preserved. If necessary, slides may be mounted in neutral mounting medium, such as DPX. Slides of suspected organ-imprint smears brought from the field are to be fixed and stained as described for blood films above.

Histological preparations of tissues sections of suspected organs are to be made by fixing them in Bouin's or Carnoy's fluid, embedding in paraffin, making microtome sections, stretching prices of sections on slides over a hot plate, processing through xylol, absolute alcohol and down to water, staining with Haematoxylin (Delafield's or Heidenhain's) and mounting in DPX. For a detailed standard histological procedure see Pearse, (1960).

b. Platyhelminthes

Fixed specimens brought from the field are to be stained and mounted in the laboratory. Borax carmine is widely used for staining Platyhelminthes. This stain is prepared by adding 4% borax solution to 3% carmine solution and boiling it for 30 minutes. It is then cooled and filtered and an equal amount to 70% ethyl alcohol is added. The specimens are stained for 10 to 30 minutes depending on the size and thickness of specimens and differentiated in acid alcohol. After the staining the specimens are dehydrated through ascending series of alcohol (30% - 50% - 70% - 80% - 90%- absolute alcohol), giving at least 3 to 4 changes in 30-40 minutes. Then specimens are cleared in xylol and transferred to clove oil or Beach Cedarwood oil for clearing. After clearing, the specimens are again brought back to remove oil and mounted in Canada balsam or any other suitable mountain.

c. Nematode

i. Plant and Soil Nematodes

For permanent preparation nematodes are to be transferred from fixative to a solution of 5 parts of glycerine and 95 parts of 30% alcohol in glass cavity. A fungicide (thymol, copper sulphate) may be added in small quantity for avoiding growth of fungi. The glass cavity containing nematodes in the solution is to be placed in a desiccator for 2-3 weeks for dehydration of nematodes at room temperature. The cover glass over the glass cavity should remain open slightly in the desiccator for the convenience of slow dehydration of nematodes.

Then, the nematodes are to be mounted in pure anhydrous glycerine. For such mounts aluminium double-coverslip slides are preferred because nematodes can be studied with greater clarity from either side of the slide. For mounting a small drop of anhydrous glycerine is to be put on the square coverslip placed on the hole of aluminium slide. The mounting drop should be such size that it just spreads to the edges of coverslip. After sorting out nematodes of about equal thickness under stereoscopic binocular microscope the same are to be transferred to the mounting drop with the aid of hair needle. These may be arranged in the centre of the drop and then glass wool of suitable size are to be placed in the drop to avoid flattening of nematodes after the permanent mount. Finally a clean round coverslip is to be gently warmed over a flame and to be placed over the mounting drop. After this, sealing material is to be applied on the outer edges of the round coverslip with the help of a brush. 'Zut' of 'Glyceel' is the best sealing material in absence of which ordinary nail polish may be used.

ii. Animal Parasitic Forms

These parasites are difficult to stain. So, these are generally studied in temporary mounts in glycerine. For clearing, the nematodes are to be transferred from fixative to glycerine alcohol (70% alcohol 90% parts and glycerine 10 parts). Then these nematodes are to be covered with a piece of filter paper to avoid dust and allow evaporation until these are brought gradually into pure glycerine. The glycerine also serves as a clearing agent. Paper labels are to be put in vials and on slides.

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PART III

SELECTED ECOSYSTEM FAUNA

URBAN BIODIVERSITY

A. K. GHOSH*

The urban biodiversity features still remain one of the most inadequately known areas of study. The current trends of study of biodiversity within identified protected areas is yet to extend areas beyond PA's; even when such studies are planned, the focal point largely revolve around rural scenario. Urban conglomerate being already built-up areas and urban development being restricted to physical planning (to a great extent) biodiversity profile of cities remain in obscurity.

Genesis of Urban Settlement

However, in tropical developing countries, the present day towns and cities must have evolved from within areas which were forested, agricultural land or reclaimed wetlands and swamps. In all such set up, the adjoining peri-urban fringe areas and remnants of original landscape provide opportunities to study biotic elements still surviving under human impact. Besides such relict elements, urban planning may also lead to massive plantation of exotic and endemic plants, creation of grassland and tanks/pools. Each one of these manmade feature attract in turn different biotic components both freshwater and terrestrial, invertebrates and vertebrates.

A study of urban biodiversity in one of the largest cities in India - Calcutta, shows that inspite of highest population density per sq.km., the city still possess more than 100 species of butterflies and 150 species of birds. The habitat offered by an admixture of exotic and endemic plants harbour every major group of insects; soil substratum providing interesting data on soil mites and earthworms. The aquatic habitats likewise offer records of freshwater sponges, plankton, aquatic insects, crustaceans and molluscs. As many as 25 species of mammals were recorded from the peripheral limit of the city, even after 270 years of its establishment, including a new species of marsh-mongoose. Such impressive data obviously can be taken as an example to justify the study of urban biodiversity. Again in India, a comparison of biotic elements recorded from Delhi, Madras, Bombay and Calcutta show unique distinctiveness depending on topographical and habitat features.

Priority Areas of Study

In order to study urban diversity the following steps may be recommended. The exercise must be based on existing maps, preferably at 1:50,000 scale.

i. Survey of trees and other plants within city limit (to find out the habitat these canopies can offer for roosting and feeding of birds/attract bees and butterflies).

ii. Survey of all listed parks and gardens within municipal areas to document ground dwelling and tree dwelling species.

iii. Survey of major identified wetlands/canal/river to document aquatic biota (fishes, crustacea, mollusca, insecta, birds) which in turn (in combination with water quality parameters) may helped to act as bioindicator.

iv. Survey of soil fauna (mites, earthworms, nematodes and rodents in subterranean habitat); and finally

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v. Integrate conservation of biodiversity with Urban Planning process.

The data collected would also be useful to develop a better understanding of urban ecology and functioning of urban environment system under stress.

It is well known that many urban areas even attract long range migrant species within wetland habitats. As such seasonal studies should be planned to document resident and seasonal migrant species.

Urban biodiversity, in the final analysis can lead to setting up of urban sanctuaries (not parks and gardens) with an undisturbed area of original landscape, as a protected area, offering urban dwellers a direct access to the past environment and an unique opportunity for creating awareness for conservation.

MARINE FAUNA

D. R. K. SASTRY*

Introduction

The marine environment consists of a Benthic Division of the sea bottom and a Pelagic Division of the overlying waters. The latter can be designated by a nearshore Neritic and an offshore oceanic region. Vertically the Pelagic Division can further be divided into a upper Euphotic zone of sufficient sunlight for photosynthesis, a deep Aphotic zone of utter darkness and an intermediate Despotic zone of diffused sunlight.

The Benthic Division is further divided into the following regions :

Supralittoral - Above the level of Extreme High Water Springs

Littoral or Intertidal - Between Extreme High Water Springs and Extreme Low Water Springs.

Sublittoral - Continental Sheld below the Extreme Low Water Springs.

Bathyal - Continental slope Continental rise.

Abyssal - General level of the sea bottom.

Hadal - Deeper regions of trenches in the abyssal plain.

The Pelagic Division can be considered uniform with its parameters such as temperature and salinity slightly varying over extensive areas. In contrast, the Benthic Division offers varied habitats consisting of rocks, corals, mangroves, sand, mud, silt, oozes etc. and often a combination of two or more of these. In addition, each habitat may be varied in several respects. For example, a rocky shore may consist of boulders, shingles or platforms and provide exposed, protected, semi-exposed, undersurface, crevice or rock pool habitats.

Ecology

The medium for all the marine organisms is principally the sea water, whether they are pelagic or benthic in habit. The Pelagic Zone supports two types of organisms the Nekton consisting of large organisms with powerful swimming habit such as fishes, whales etc. and the Plankton including microscopic organisms with feeble swimming or drifting habit such as medusa, copepoda, arrow worms, and larval forms of almost all marine organisms. In addition a third category, Neuston is often distinguished for forms like *Janthina, Physalia, Velella, Porpita etc.*, which drift at the air-water interface.

The benthic organisms, on the other hand, exhibit various habits to suit the different habitat they occupy. The hard substrate are inhabited by permanently attached forms such as hydroid colonies, serpulids, barnacles, oysters; organisms with firmly holding mechanisms such as mussels, limpets, sea

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urchins; organisms of crawling habit with firm hold such as crabs; and boring organisms such as sipunculans and some molluscs. The algal covering provides sheltered habitat for many polychaetes, amphipids, isopods, crabs, shrimps and molluscs. Many organisms occupy crevices during exposed low tide periods. The soft substrate are inhabitated by crawling, quick burrowing, semi-buried or tube-dwelling organisms such as polychaetes, crustaceans, bivalves, gastropods and sea stars. Mangroves are the habitat for several attached, crawling and boring organisms. Corals and gorgonaceans harbour a variety of symbiotic, epizootic and boring organisms. Estimates of taxa of some invertebrate groups in the world and coastal South Asia are given in Table - 1.

Table 1 Estimates of Species of Some Marine Invertebrates

Taxonomic group	Approximate number of marine species					
	World	India	Maldives	Pakistan	Sri Lanka	Bangladesh
Porifera	5000	500	-	-	-	-
Cnidaria	10000	800	-	-	-	-
Ctenophora	100	10	-	-	-	-
Gastrotricha	25000	67	-	-	-	-
Kinorhyncha	100	10	-	-	-	-
Sipuncula	202	38	15	-	9	1
Echiura	127	33	5	-	1	-
Annelida						
Oligochaeta	3	1	1	-	1	-
Hirudinea	28	12	-	1	2	1
Polychaeta	8000	400	44	7	94	-
Archiannelida	125	21	-	-	-	-
Phoronida	11	3	-	-	-	-
Bryozoa	4000	170	5	9	20	10
Brachiopoda	280	5	-	-	-	-
Chaetognatha	100	30	-	-	-	-
Echinodermata	6225	765	200	23	200	12

For convenience of designation, the fauna of marine environment is identified with their habitat and habit, such as intertidal sandy benthos, offshore plankton bathypelagic, abyssal benthos etc. From the stand point of size, the organisms are designated as Macrofauna - large animals which can be easily seen with naked eye: Microfauna - small organisms which measure only a few millimetres; and Meiofauna - organisms measuring less than a millimetre and retained by a 50 μ sieve. The equipment and method of collection according vary with the habitat and size of the organisms under consideration.

Benthic Fauna

The macrofauna of supralittoral and intertidal benthos can be best collected during low tide periods when the region is well exposed. The organisms can be collected with the help of simple instruments such as scalpel, forceps, hammer and chisel, hand shovel etc. by carefully searching the various habitats. Boring organisms of hard substrate are extracted with hammer and chisel or a sharp scalpel by carefully chipping the rock or wood, piece by piece, without damaging the organisms. A hand net can be used in rock pools and submerged algal beds. Sand dwelling micro-organisms can sometimes be easily located by the markings on the surface. Usually the substratum upto 30-cm is transferred into a sieve and thoroughly washed.

The algal tufts can be carefully scrapped from the substratum with a scalpel or mason's scraper and vigorously rinsed in a bucket of sea water with a few drops of formalin, to dislodge the organisms. Frequently, a careful examination under a stereomicroscope is necessary to collect all the organisms. The epizootic and symbiotic organisms of corals and other hosts which lie in shallow waters, escape at the slightest disturbance. Hence the coral colony or the host organisms need to be enclosed in a polythene bag with its mouth tied at the base, before separation from the substratum.

For collecting meiofauna, the substratum upto 30 cm depth is transferred into a bucket of sea water and vigorously stirred with a glass rod to dislodge the specimens from the sand grains. The supernatant is decanted and filtered or the organisms allowed to settle in suitable containers.

The following special equipment's are required for collecting subtidal benthos:

i) Beam Trawl - It is essentially a collecting bag-net kept open by a horizontal metallic beam. It is dragged on the surface of a soft bottom from a slow moving mechanised vessel. The organisms crawling on the surface or swimming slowly near the surface are collected by the net.

ii) Naturalist's dredge - A dredge consists of a rectangular mouth frame and a collection device of upper and lower metallic sheets with wire mesh on the sides and at the rear. The horizontal beams of the mouth frame are edged with projecting balder in order to shovel the substratum into the collecting device along with its inhabitants. The dredge is to be used on hard substrata with small pebbles orgravel while the beam trawl is meant only for soft bottom benthos.

iii) Van Veen Grab - The grab essentially consists of two hollow metallic jaws hinged together and kept open by a locking mechanism while lowering from a stationery boat. On touching the bottom, the jaws dig into the substratum and the locking mechanism is released. On hauling up, the jaws snap shut enclosing the substratum along with the inhabiting organisms. As it collects a constant and specified quantity of the sample depending on the size of the jaws, the samples can be used for quantitative estimates of the fauna.

For quantitative estimates of the intertidal and sublittoral fauna with scuba, the organisms in a quadrant of one metre square are counted. for burrowing soft bottom benthos the substratum upto 30 cm is transferred into a sieve and carefully washed. For quantitative estimation of soft bottom microfauna a hand core of 30 cm length is to be used.

Pelagic Fauna

For collecting powerful swimming Nekton such as squids, fishes, and whales various types of nets such as shore seines, purse seine, otter trawl, midwater trawl, gill net, stake net and special methods such as long lines, harpooning, pole & line etc. are used.

For collection of planktonic organisms, a standard plankton net is used. It consists of a conical net of bolting silk or organdie or required mesh. A metallic ring at the wider end keeps the mouth of the net open. A small cylindrical container at the narrower end collects the planktonic organisms sieved by the net. The net is hauled by the metallic ring from a slow moving mechanised vessel horizontally in the surface layers. Horizontal sampling is done at night or early hours when the planktonic organisms are expected to be maximum in the surface layers during their diurnal vertical migrations. The net is hauled vertically from a stationary boat to collect plankton from deeper regions, by attaching a weight at the end of the towing rope. By adjusting the weight and length of the rope paid, the net is used for vertical hauling from specified depths to the surface.

The plankton samples from vertical hauls from same depth and horizontal hauls for the same period can be used for a comparison of faunal abundance. However, a flow meter attached at the centre of the ring gives the exact quantity of seawater filtered by the net and thus the organisms can be more accurately expressed by number per cubic metre of water column. There are also special instruments with built-in flow meters and automatic closing mechanisms, which can be operated serially at different depths and for specified periods of time.

Preservation

Plankton

Immediately after hauling up, the plankton net is thoroughly washed with seawater to dislodge the organisms entangled in the meshes of the net and the sample in the collection bucket is transferred into a suitable container. The plankton sample is immediately and directly fixed by adding sufficient quantity of neutral or buffered seawater formalin to make a final concentration of 4% formaldehyde.

Nekton

The organisms can be directly killed and preserved in sufficient quantity (about nine times the size of the organisms) of 4% neutral formaldehyde or 90% alcohol, after a preliminary cleaning with seawater.

Meiofauna

The meiofauna sample is taken into a beaker containing 6% magnesium chloride solution for Narcotisation. Later the organisms are filtered and fixed in 4% neutral formaldehyde or 70% alcohol containing 2% glycerine.

Benthos

Preservation of benthic organisms consists of the following stages.

i. Cleaning - The benthic organisms generally contain particles of the substratum, shell pieces, other debris, mucus etc. adhering to them. The organisms are thoroughly washed in fresh seawater and cleared with a brush if necessary.

ii. Narcotisation - The cleaned organisms are transferred to wide shallow basins or petri dishes containing sea water and a suitable narcotising agent is added in order the specimens to relax to their normal habit. The usual narcotising agents are 70% alcohol, 4% neutral formaldehyde or 15 chloral hydrate solutions added drop by drop at interval or powdered menthol or magnesium chloride crystals sprinkled on the surface of sea water. Containers are closed with a lid and kept undisturbed. In about 12 hours the organisms are narcotised and do not respond to touch with a needle or brush.

iii. Fixing: Soon after the organisms get narcotised, they are to be transferred to a fixing agent to kill them and stop deterioration of the tissues due to physiological or bacterial action. The most widely used fixing agents are 4% neutral seawater formaldehyde or 95% alcohol. The animals are kept in the fixing agent for not less than a day or longer if required as per the size of the organisms. For larger organisms, the body is slit or the fixing agent is injected into the tissues for quick penetration of the fixing agent.

iv. Preservation: The fixed organisms are transferred to suitable containers with sufficient quantity, usually nine times the size of the organisms, of a permanent preservative either 4% buffered or neutral sea water formaldehyde or 95% alcohol, similar to the fixing agent used. Formalin fixed organisms are to be thoroughly washed if they are to be preserved in alcohol.

While the above stages are necessary for almost all the groups of organisms, exceptions should be or can be followed for the following groups:

Porifera: After cleaning in seawater, sponges can be directly preserved in 95% alcohol or fixed in alcohol and preserved dry. Formaldehyde should not be used.

Cnidaria: Alcyonaceans should be narcotised, fixed and preserved only in alcohol. Corals and goraonacens are to be fixed in alcohol and can be preserved in alcohol or in a dry condition. Other cnidarians are to be narcotised, fixed and preserved only in formalin and alcohol should into be used.

Echinodermata: Holothurians are to be narcotised, fixed and preserved only in alcohol and formalin should not be used. Other echinoderms can be narcotised and fixed in formalin or alcohol, but should be preserved only in alcohol or in a dry state. Echinoids do not require Narcotisation.

Organisms required for dissection or Histological study should be preserved in special fixatives meant for the purpose such as Bouin's, Zenker's, in Formal-Acetic acid - Alcohol etc.

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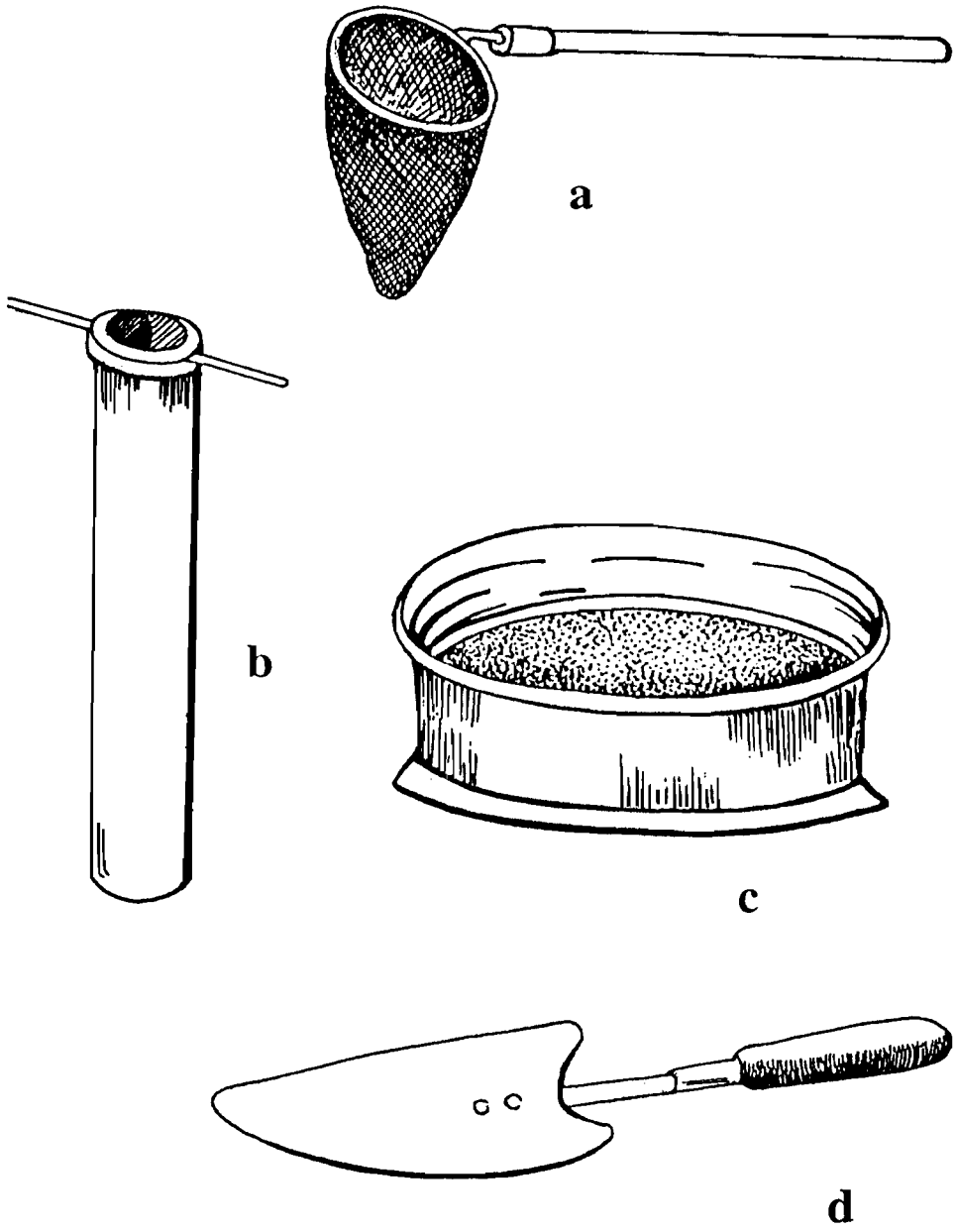

Fig. 11 - **Collection Equipments for Marine Organisms**

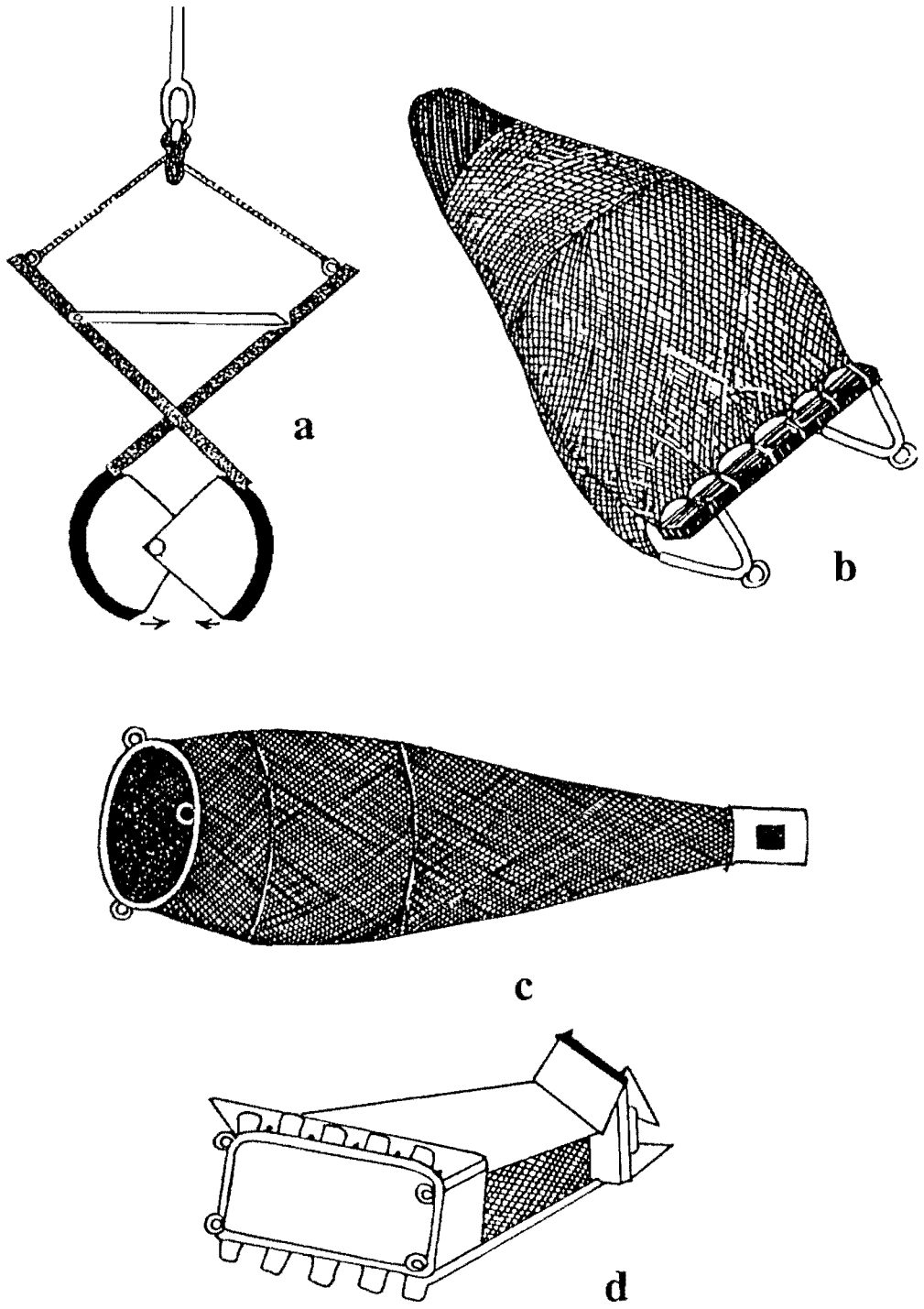

Fig. 12 - **Collection Equipments for Marine Organisms**

SOIL FAUNA

A. K. HAZRA*

Introduction

Major soil inhabiting biota are as follows:

Heterotrophic microflora (Bacteria and Fungi) : Soil bacteria occur in clumped colonies around individual soil particles forming 0.03 per cent weight of the top soil and the roots and root hair of points are generally enclosed in bacterial film and they are more dense in the rhizosphere. About four billion bacteria estimated per gm. of soil and in goods soils bacterial matter has been known to weigh about 5600 lb/ acre. More than 20 million actinomycetes occur in one gram of dry soil. More than 200 species of moulds and fungi occur in the soil, the estimated number being one million fungi in one gram of dry soil.

Soil Protozoa and Soil Nematods have been described elsewhere in this Manual.

Earthworms (Oligochaeta : Haplotaxida : Annelida) : Large size oilgochaetes. Since time immemorial earth worms are known to be friends of farmers. They are cylindrical, bilaterally symmetrical coelomate worms with internal and external metameric segmentation. They lack any appendages and suckers but possess a few hook-like chaetae embedded in skin with which they gain hold on substratum. Earthworms are represented by 508 species in the Indian Subcontinent as compared to 3320 species in the world.

Potworms: (Oligochaeta : Haplotaxida : Enchytraeidae : Annelida) Enchytraeids) : Small worm like oligochaetes. They are widely distributed and found in all parts of the world in habitats ranging from marine and freshwater littoral to benthos. In India only 21 species are known; in comparison 500 species are recorded from the world.

Mollusca: The soil molluscs comprise those animals known as sludge and snails and they live either on the surface or in the top 30 cm of soil. In snails the size of the shell is dependent on the availability of calcium and the thin-shelled species viz. *Vitrina sp.* can be found on acid soil. The details are dealt with elsewhere in this Manual.

Isopoda: The members of this group ranging in size from 2-20 mm, are terrestrial crustaceans. They are wide spread. In world: 400 species. In India: 200 species.

Thysanura: Popularly known as "silverfish" or "bristle tails", the abdomen is composed of 10-11 segments. They have very long, many segmented antennae, cerci and median tail. Generally found in bark of trees, forest floor, termites and ant nests. In world: 1250 species, in India: 23 species. However, more than 150 species are expected from India.

Diplura: These are soft bodied wingless soil wingless soil inhabiting insects. Body without scales, eyes absent, cerci long and filiform. In India: 16 species. In world: 355 species. More than 100 species are likely to be discovered from the Indian subcontinent.

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* Zoological Survey of India, Calcutta

Protura: The body is cylindrical, antennae and eyes absent, first pair of leg of these insects are used as tactile organs, cerci absent. They are found even from 20-cm depth of soil. In India: 20 species. In world: 260 species. More than 50 species are expected yet to be discovered from the India.

Collembola: Popularly known as "springtail" due to the presence of spring like, forked jumping organ, below the fourth abdominal segment. these insects are unique in having only six abdominal segments. They are found to occur from intertidal seashore to snow peak mountains. These of insects play a significant role in the break down of leaf litter. In India: 200 species. In world: 5000 species. More than 200 species are expected to be discovered from the Indian subcontinent.

Staphylinidae and Scarabaeidae : These families of Coleoptera are commonly associated with soil, both in larval and adult stages. Staphylinidae: In world: 30,000 species. In India: 154 species. Scarabaeidae: In India: 160 species. In world: 27,500 species.

Millipedes (Diplopoda): Millipedes are joint footed, many segmented animals having double pairs of legs on each segment of body. In India: 162 species. In world : 7500 species. Less studied group in Indian subcontinent.

Pseudoscropions (Pseudscorpionida : Arachnida): These are dorsoventrally flattened animals, which resemble scorpions in general form of their pedipalps and body, except that the hind part of the body is not narrow. In India: 100 species. In world: 230 species. Not properly studied group in India.

Soil Mesostigmata (Acari): The adult bears a number of distinct chestnut-brown shields on dorsum and venter. They are most abundant in soil. They play major role in the soil humification. In India: 110 species. In world: 4000 species. Many more species are expected from India.

Oribatids (Cryptostigmata): Most abundant soil inhabitant group. Popularly known as "beetle mites". The adult mites often posses a discrete tracheal system consisting of a series of ducts which open laterally between coxae II-III. In India: 328 species. In world: 6000 species. It is expected that about 500 more species are yet to be discovered from Indian subcontinent.

Collection

Heterotrophic microflora; Random sampling of soil by means of soil corers from different depths of soils are to be collected and kept in polythene packets with proper levelling for estimation of soil bacteria and fungi.

Soil Protozoa and Nematodes: Described elsewhere in this Manual.

Earthworms: These are collected by digging soil with shovel or spade. They are also collected by hand sorting from mosses, soil and dung. Dilute solution of formalin employed for collection from the soil (.55% solution prepared by adding 40% formaldehyde solution in 4.5 litres of water). Potassium permanganate solution (1.5 gm/litre) is also often used to collect earthworms from soil.

Potworms (enchytraeids): These soft bodied minute animals are collected by means of soil corer and extracted in special kind of extractors (Nielsen extractor and wet funnel extractor).

Molluscs, Slugs: Both soil washing and flooding methods are used. Flotation method is used for snails.

Soil-Arthropods: These are collected generally by two methods:

i. Direct

ii. Extracting from soil samples.

i. Direct: a) Beating the soil and litter, (b) Hand picking for bigger forms, and c) By aspirator.

ii. Extraction from Soil samples:

They are normally extracted in two main ways:

a. *Dynamic or behavioural method*: For soil arthropods the typical method of extraction is by causing a dry zone to move slowly down a soil sample. This tends to drive animals into moisture and cooler parts of the sample and from there into a sampling tube. This method is used by employing a tullgren funnel and it is obvious that the samples have to be processed immediately since the animals have to be alive otherwise they will not be extracted.

b. *Mechanical Method*: This is soil washing system that is based on the different specific gravity's of the soil and its fauna. This involves separating the organic debris from the inorganic matter and fauna by washing with water through a series of sieves. The fauna is then separated from the inorganic matter by floating off animals in magnesium sulphate solution of the correct specific gravity (1.17-1.20). This letter method has the advantage that the samples can be cold stored for some months before extraction as the animals are just as efficiently extracted dead or alive.

Equipments

Heterotrophic microflora

Soil augur, b) autoclave, c) Distilled water plant, d) Incubator, e) Refrigerator, f)

a) Test tubes, g) Petridishes, h) Aseptic chamber, i) Bunsen burner, j) L-shaped rod for inoculation, etc.

Earthworms: a) Showvel, b) Spade, c) Long forceps, d) Enamel tray

Pot worms: a) The split corer, b) Nielson's extractor, c) Wet funnel extractor.

Molluscs: a) Shovel, b) Spade, c) Forceps

Soil Arthropods

General: a) Stainless steel corer b) core holders, c) Spade, d) Shovel, e) Enamel tray, f) Aspirator, g) Forceps, h) Brush, i) Hand lens.

For Extraction

Dynamic methods: Simple plastic funnel extractor, Rothamsted funnel extractor without light source or heat source, Murphy split funnel extractor, Kemson infra-red extractor, Macfedyen high gradient extractor and Macfadyen air-conditioned funnel extractor.

Mechanical Methods

a. Salt and Hollick floatation method : Sieves of different mesh sizes, Ladell container and cylindrical shaker.

b. Greas film-method extracto: Rectangular plastic tanks (7.5 cm x 10 cm x 20 cm), and Mechanical shaker.

b) Quantitative Method of sampling

The number of soil animals present in a defined area are estimated by direct method. Taking soil sample in a measured corer, extracting the animals and then multiplying the number caught by a factor to give the number per unit area or volume of soil.

The plot in which the quantitative assessment of soil fauna is to be made is divided into several quadrants. Having decided on appropriate extraction method, the investigator must than determine the size and number of sampling units required for population estimates of known accuracy for each animal group from each quadrate.

Heterotrophic Microflora

The quantitative and qualitative study of soil micro-organisms is carried out by the dilution plate count technique.

Preparation of Soil Suspension

Dilution is made to get the organisms in a countable range (9: ratio, 10 fold dilution is preferable)

a. One gm soil of 9 ml of double distilled water
b. One ml of diluted soil solution to 9 ml water (to get 10^{-2} dilution)
c. From this suspension 1 ml. soil solution is taken and added to 9 ml water (to get 10^{-3} dilution and dilution is done upto 10^{-6}).

Preparation of Petridish

A solid medium is commonly used in a plastic petridish usually the size which has a lid diameter of about 9 cm. Before use the medium must be sterile.

Composition of Basal Media

Peptone 1%; NaCl 0.5%; beef extract 0.5 - 1% (This forms nutrient broth).

Nutrient broth jelled with 1.5-2% agar. For counting of soil fungi Wakman's special medium is used. Composition: (for 1000 ml) Glucose - 10 gm; Peptone - 5 gm. Potassium dihydrogen phosphate-1 gm.

$Mg.So_4$ - 0.5 gm. Agar - 25 gm, water - 1 lit pH is to be adjusted to 4.00 by addition of NH_2SO_4 or H_3PO_4.

Inoculation: A spread plate is made by spreading a small volume of liquid inoculum (0.1 ml) over the surface of the solid medium by means of a sterile L-shaped glass rod.

Incubation: Plates should be incubated for 24 hrs. at 37° C for bacteria and at 30° C for fungus.

Counting: The total number of cells in a sample is called *total cell count* - it can be estimated by direct counting in a typical counting chamber (Haemocytometer). The number of living cell is viable cell count - it can be estimated from the number of colonies.

Earthworms:

The quantitative estimation of earthworms can be done by the following method.

In an arable soil the average number of worms estimated per m^{-2} from 50 units of each size were 25 x 25 x 84.5, 50 x 50 x 77.7; 50 x 100 x 62.8; 100 x 100 x 62.5. A tool for taking soil samples of 25 x 25 cm ($1/6m^2$) to a depth of 20 cm can be used in soils which are not too stony. Svendsen used a steel scoop of semicircular cross section to cut cores 283 sq.cm ($1/35m^{-2}$) in area and 20 cm deep. The minimum sample area which will prove satisfactory will depend largely on the density of the population to be sampled.

Enchytraeids (Potworms)

A reliable method exists for the quantitative removal of enchytraeids from soil. The principles of the method of the method and the construction is as follows:

In the Nielsen's extraction method the worms move upwards into the layer of cooled sand at the top of the core under the influence of temperature gradients. After about two hours all the worms will have moved into the sand layer which is then removed and the worms washed from it with a gentle stream of water.

Removal of Soil Cores for Extraction

The enchytraeids are soft bodied and some care is needed in taking soil cores, especially from heavy-clay soils as such soil tend to stick in ordinary cylindrical corers and great force is needed to remove the core. This can impair extraction efficiency, both by damaging the worms and by compacting the soil. These risks are avoided by the use of a longitudinally split corer. This system has the great advantage that the corer insert can be opened to remove the core and that the core can very readily be divided into the natural divisions of the soil profile before extraction.

Molluscs

The quantitative estimation of snail population can be conducted by counting the numbers within measured quadrant. This method is good for the surface dwelling forms in grassland habitat.

A special method has been evolved to separate species that live in litter. Snail's shells are separated from the litter by a flotation method but by this method overestimates of the living population might be made as recently dead animals would be extracted indistinguishably from live animals. Snail shells

flat in water because air becomes trapped in the shell whorls. Separation of plant material from snails is accomplished by soaking and agitating the sample in a solution of molluscicide. The dead animals will sink to the bottom with the soil, leaving the plant material and empty shells floating on the top. This flotsam can be decanted off and the reminder is then thoroughly dried. This process fills the shells with air so that when the sample is once again placed in water all the shells float, the soil sinking to the bottom.

Soil Arthropods

The quantitative estimation for the soil micro-and macrathopods are made by counting individuals in a certain number of sample units. Therefore, as a preliminary step, one shall decide the dimension and number of sample unit to be taken, the frequency of sampling and the method of extraction to be used.

Sample Size

Samples are mostly taken at a constant volume and area by means of cylindrical corer. Generally these cores have an area between 10 and 20 cm^{-3} and penetrate the soil to a depth of 2-6 cm. Such corers are suitable for relatively compact soil, but have to be used carefully in sampling forest litter. In this case, sample units should be obtained either by keeping the volume constant or by taking a constant area using a quadrant or frame. In general, the area of the sample unit is known with a fair degree of accuracy, and the precision of the estimate for density is determined essentially by the precision of the estimate of the mean.

Methods of Extraction

A review of all the methods for extracting edifice Arthropods from their substratum would be too wide a subject. A comparative study of the efficiency of different types of extractor has been made by Edwards and Fletcher, 1971. The most efficient extractor for quantitative estimation of soil arthropod fauna is by: The Macfadyen air-conditioned funnel extractor. The apparatus is completely enclosed in a rectangular cabinet made of chip board, supported on angle-iron frame work, and lined with expanded polystyrene. The funnels are arranged in rows or three, fit tightly into holes in removal chip-board trays and are 25 cm high x 15 cm diameter with an included angle of 32 narrowing to an opening 1.2 cm diameter. Collecting tubes 2.5 cm diam x 5 cm deep are held in position at bottom of the funnels by rubber sleeves. A door in the front gives access to the funnels and sample containers, 10.5 cm diam. x 5 cm deep, are suspended above funnels, and mouthed in a baffle board cover with aluminum foil to minimise heat transfer. The sample containers are supported by a metal rod across the top of each funnel and held firmly in position by holes in the baffle. The upper part of the cabinet containing the heater assembly is supported on pulleys and can be raised to insert samples and lowered onto the sample containers which it flush to the surface of the baffle board. The heaters are of length 20 gauge inichrome wires 7.5 ohm stretched between ceramic insulators. Each heater serves three funnels which are arranged in paris each side of vertical ducts. A fan circulates moist air in the lower compartment with ducts arranged so that air currents pass over a cold water bath, through the coils of an evaporator from a refrigerator, to the lower surface of the soil sample. The air then passes through holes in a false back plate and back to the fan. The air passages in the false back are of equal length, to ensure an even air flow to all funnels. The evaporator coils lead to long lengths of flexible copper pipe to complete the circuit between the compressor and condenser. The compressor is driven by a 1/4-h.p capacitor start induction motor (230 v. 50 cycles single phase) and a thermostat controls the air temperature in the cabinet.

The soil samples are taken in the field in the sample containers, which have a sieve of 10 mesh 23, S.W.G., held in position by crosswise supports from the sides. Intact samples are inverted and left in the apparatus for nine days and animals are collected in 70% v. aqueous ethyl alcohol and glycerol (ratio 2:1). For the first seven days the air in the lower cabinet is regularly moistened, using an atomiser driven by a compressor to maintain a relative humidity greater than 85% and cooled (8-10°C) while the temperature in the heating compartment increased to 450°C. The refrigerator unit is there switched off on the 7th day and relative humidity allowed to fall.

Preservation

Heterotrophic Microflora

The soil samples collected in the field for studying microflora are kept in polythene packets with proper labelling. After sampling it is recommended that soils should be kept as cool as is practical.

The plant can be preserved at 4-6°C, until needed.

Earthwork & Potworms

The oligochaetes are to be narcotised by using fresh water. They are washed and placed in a dish containing freshwater. After a while, they will tend to extend and are then killed by pouring concentrated formalin to the water. Earthworms can be killed also by dropping them in 70% ethyl alcohol, when the worms become motionless they are removed from alcohol and placed on a piece of blotting paper or any other absorbent paper in straight position for fixation. After fixation they are preserved in vials or bottles filled with 70% ethyl alcohol or 10-15 formalin if the size is bigger.

Soil Arthropods

The soil samples collected from the field for extraction of soil insects, Acarina, Arachnid etc. are to be kept cool, minimum moisture of soil to be maintained, the mouths of polythene packets containing soil samples are to be kept open for oxygenation while in the field.

The collected soil inhabiting specimens are best preserved in 70% ethyl alcohol in tight vials or bottles.

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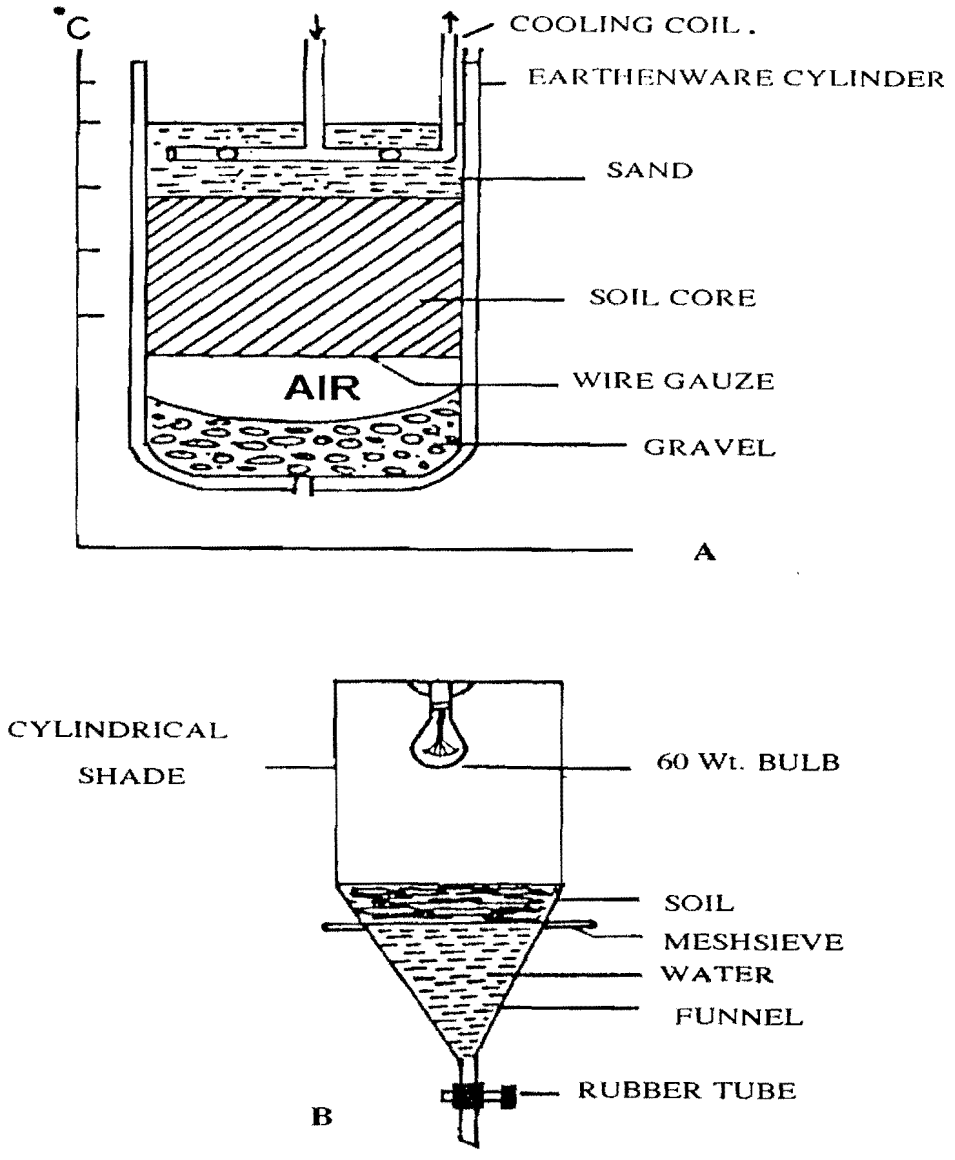

Fig. 13 - Extraction Apparatuses for Soil Pot-worms

PART IV

ANALYSIS OF DATA

VERTEBRATES

P. K. DAS*

Introduction

Various groups of vertebrates play a very important role in the economic prosperity of different countries. Ana analysis of classification follows:

Cyclostomes, comprising lampreys and hagfishes, belong to primitive group of vertebrates, which are not known to occur in countries of South Asia. This group of vertebrates does not have jaws. But all vertebrates of South Asia have upper and lower jaws. Therefore, vertebrates of this region are all included under the Subphylum Gnathostomata (mouth bounded by jaws) of the phylum Chordate.

Characters

All vertebrates have a vertebral column each. This structure is otherwise known as the backbone, spinal column or simply, spine. The vertebral column is made up of a number of segmented vertebrae. It extends from tip of the tail to the base of the skull. A vertebrate animal has distinct head, inside which lies the skull. The skull essentially consists of the cranium or brain-case, paired sense organs (eyes, ears nostrils) and the lower jaw. A post-anal (originating from beyond the anal/cloacal aperture) tail is another most important character of vertebrates. The tail may be absent in adults, but is always present in the early part of the life-history.

Paired appendages of vertebrate animals are never more than two pairs. In fishes, these paired appendages are the two pairs of paired fins. In tetrapod, a pair of fore limbs and one of hind limbs represent the paired appendages. Digits of vertebrates are not more than five on each limb. In many instances, these can be even less than five. The skin of vertebrates are covered with scales, feathers or hairs. In some groups, however, the skin is said to be naked, i.e. not covered with any one of these. From the point of origin, scales of vertebrates fall under two distinct categories, e.g., superficial (epidermal) and deep-seated (dermal). It is worth mentioning here that in many vertebrates, the males differ from their females in size, structure and/or coloration.

A schematic classification is given below

Phylum Chordate

Subphylum Gnathostomota [4, 778 species in Indian area].
Superclass Pisces [2, 546 species in Indian waters]
Class Chondrichthyes [10 orders, 28 families, 67 genera and 131 species in Indian waters].

Class Osteichthyes [30 orders, 226 families, 902 genera and 2,415 species in Indian water]
Superclass Tetrapoda [2,232 species in Indian area].
Class amphibia [3 orders, 9 families, 32 genera and 204 species in Indian area]

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Class Reptilia [3 orders, 26 families, 137 genera
and 428 species in Indian area]
Class Aves [20 orders, 78 families, 405
genera and 1,232 species in Indian area]
Class Mammalia [13 orders, 45 families, 169 genera
and 372 species in Indian area].

Fishes

All fishes are accommodated in the Superclass Pisces, which have certain common characteristics, as follows:

Fishes are provided with two paired fins, which are supported by stick-like structures called fin-rays. Being primary aquatic vertebrates, fishes breathe through gills placed on either side of the posterior part of the head. Besides the paired fins, fishes have also a number of unpaired fins whose number, position, shapes, etc., vary in different groups of fishes, but all are placed medially and vertically, on the dorsal and ventral aspects of the animal as also at the end of the tail.

Most fishes have scales. Scales of fishes are deep-seated, i.e., these are embedded in the deeper layer of the skin known as the dermis. Hence, scales of fishes are called dermal scales. Many fishes, however, are scale-less. All fishes live in water. They are to be found in all kinds of fresh water bodies, in the saline water of seas as also in the brackish water of the estuary.

Fishes are broadly classified into two classes, viz, Chondrichthyes and Osteichthyes.

Sharks, rays skates and their allies as also chimaera are included in the Class Chondrichthyes. Skeleton of this kind of fishes is made up of cartilage which is softer as compared to the done. Their skin is covered with minute scales, called placode scales which are structurally similar to their teeth. The gill-slits of this class of fishes open separately to the exterior. A cartilaginous fish has its mouth placed ventrally. Chimaera are on odd group of cartilaginous fishes which are not only scale-less in the adult stage but they also have coverings of their gills like the bony fishes. The coverings of the gills are known as opercula (Operculum in singular). Cartilaginous fishes are marine. Some of these fishes are, however, also found in estuarine waters.

Fishes belonging to the class Osteichthyes have their skeleton made up of bones. All the food fishes of commercial importance belong to this class. This happens to be the largest class of vertebrates. They live in every kind of fresh water body, in the sea and also in the estuaries. Primitive bony fishes like the Coelacanth (Latimeria), lung fishes, Bichir, sturgeons, gar pikes and the Bowfin are absent from South Asia. Bony fishes of this region are usually covered with two types of scales - the cycloid or the smooth scale and catenoid or the spiny scale. Many fishes, including the carps have cycloid scale. The exposed end of this type of scale is not provided with spines. The exposed end of the catenoid scale is provided with tiny spines. Perches, murrels, etc., have this type of scale. However, quite a large number of bony fishes are devoid of scales. Cat-fishes as a group are scale-less, many other fishes are also scale-less. The gill-slits of bony fishes do not open separately to the exterior. The gills of one side are covered by a bony operculum. Water entering the gills come out through an aperture between the operculum and antero-lateral part of the trunk. Mouth of bony fishes is usually terminal. Their bony fin-rays are often modified into sharp and even serrated spines. Many have lateral line sense organs - two longitudinal lines (some times broken) along the sides of the tail and the trunk.

Amphibians

Frogs and toads, salamanders and newt and caecilians are included in the Class Amphibia. As the name signifies, these animals lead a double life in the sense that in the early part of their life (larval life) they live in water and breathe through gills. Their adults, however, breathe through lungs, be they in water or on land.

Amphibians are recognised by their naked and moist skin. Their skin is not covered with scales, feathers of hairs. Their limbs are provided with five, four or less number of toes, which do not have claw. Unlike fishes, nostrils of amphibians are connected to the mouth cavity and are provided with valves, which exclude water and aid in breathing. Many amphibians including the frogs and toads have protrusile tongue. Most amphibians have fine teeth.

Depending on the presence or absence of tail in the adult and on the presence or absence of limbs, Class Amphibia is divided into three distinct orders, namely

Order Gymnophiona - both pairs of limbs totally absent, body slender and elongated, skin with transverse furrows, tail short eyes degrade, a small tentacle between eye and nostril. Caecilians belong to this group of amphibians.

Order Caudate - both pairs of limbs and a tail present in the only Indian representative (Himalayan Newt) of this order.

Order Anura - adults tail-less (but their larva, called tadpole, has a tail), both pairs of limbs are present - the hind limbs are usually stronger and longer, often the toes of hind limb are connected with one another with thin layer of skin. Frogs and toads belong to this order. Incidentally, largest number of amphibians belong to this group.

Reptiles

Lizards, snakes turtles and tortoises, and crocodiles and gavial constitute the reptilian fauna of South Asia. The tuatara or sphenodon, famous for its antiquity, is not found in this region.

Reptiles belong to the Class Reptilia. Unlike amphibians, the skin is dry and is covered with scales, which are different from those of fishes. These scales originate from the superficial layer (epidermis) of the skin, sometimes sautés from the body-covering. Typically, they have two pairs of limbs, each limb with five toes, which end in a claw each. In some, however, the limbs are absent.

Class Reptilia is divided into three distinct orders:

Order Crocodile - comprising the crocodiles and the gavial. These animals have elongated body with long jaws and laterally compressed, long tail. Their skin is thick, leathery and is covered with sautés. The sautés on the back and belly are larger and rectangular. Toes are with claws and have webs between them. The ear-opening is protected by a flap of skin, jaws are armed with bluntly conical teeth. Two species of crocodiles and one species of gavial are found within the Indian limits.

All turtles and tortoises belong to the Order Testudines. These are characterised with broad body encased in a firm shell of rounded dorsal carapace and a flat ventral plastron, joined at sides, and covered by polygonal sautés or leathery skin; jaws tooth-less and covered with horny sheaths; in aquatic forms the to are webbed, while in marine turtles the limbs are modified into flippers.

The third i.e. Order Squamata embraces two distinct groups of reptiles, namely, the lizards and their allies (Suborder Sauria) and the snakes (Suborder Serpents). Lizards have both pairs of limbs, but in some both the limbs are absent. Their tongue is usually entire. In snakes, both pairs of limbs are absent. The tongue of snake is slender and bifid. Their eyes are covered with transparent scale. Earopenings are absent in snakes.

Birds

All birds belong to the Class Aves. Birds can be recognised by their specialised body-covering, the feaster. Featherless portion of the leg of birds is covered with cornified skin. Jaws of birds are toothless and are covered with horny seath forming the bill or beak. Fore limbs are modified into wings while the toes end in claws, and in swimming birds, are provided with webs.

Flightless birds, namely, ostrich, rhea, emu, cassowary, kiwi, and penguins are not found in South Asia.

Mammals

As animals with feathers are called birds so are animals with hairs called mammals. All mammals belong to the Class Mammalia.

The egg-laying mammals (Duck-billed platypus, spiny anteaters) and the pouched mammals (kangaroos and allies) as also sloths, giant anteaters, armadillos, seals and sea-lions, Arad Vark and hyraxes are not found in South Asia.

In some groups of South Asian mammals the hairs are modified variously, e.g., modified as spines (in hedge-hogs), quills (in porcupines) and large scale-like horny plates (in scaly anteaters). In whales and dolphins only few hairs are to be found on the muzzle, the rest of body is devoid of hair. Toes of mammals are provided with claws, nails or hoofs. Forelimbs of bats are modified into wings while the hind limbs are absent in whales and dolphins and in sea-cows. Fore limbs of these latter group of mammals are modified into paddle-like structures. Most mammals have pinnae, which are quite large in hares and rabbits and in some bats, while in house shrews these are quite small. In whales these are absent. Teeth of mammals are of several distinct types and are embedded in sockets of jaws. In toothed whales, however, all teeth are of the same size, while teeth are absent in adult anteaters.

Identification

Identification of vertebrates, like other groups of animals, is done on the basis of their morphological characters. Certain vertebrates, especially the larger reptiles (crocodiles, gavial), large mammals (tiger, panther, elephant, lion, different species of deer, rhinoceros, monkeys, wild ass, etc.) and many species of birds can be authentically identified in the field by simply observing them. Characteristic calls of certain mammals (e.g, Hoolock Gibbon) and many birds act as tools for their positive identification even if these animals are not visible to the observer. In most cases, however, study of the morphological characters present in the fixed and preserved specimens (either while or parts thereof) are essential in the authentic identification of different species of vertebrates.

Characters, which aid in the positive identification of an animal, are called its taxonomic characters. These taxonomic characters are to be found in specimens preserved for the purpose of identification. Sometimes certain biological characters (e.g., the characteristic calls of the Hoolock Gibbon, the

House Crow, etc.) also are to be regarded as taxonomic characters, because these also help in the positive identification of the species.

Taxonomic characters vary in different groups of vertebrate. Characters most usually utilised in the identification of different groups of vertebrates are enumerated in the following paragraph.

Fishes

Shape and size of the body; number and position of gill-slits; number, position and fusion of different fins are some of the most useful characters utilised in the identification of cartilaginous fishes.

Shape and size of the body; number and relative positions of different fins; number of fin-rays in different fins; number of fin-rays modified into spines; fusion of nearby fins; number of scales and width at certain specific points of the body, etc., are some of the characters used in the identification of bony fishes.

Amphibians

Presence or absence of limbs; presence or absence of tail in the adult; number of toes on limbs; degree of webbing between toes; presence or absence of discs on toes; size and shape of warts are some of the most useful characters used in the identification of amphibians.

Reptiles

Shape of the body; nature of body-covering (scales, sautés or shell); Presence or absence of limbs; webbing of toes: presence or absence of teeth on jaws; position and structure of teeth; number and relative size of scales in particular positions of the body; nature of tongue are some of the most important taxonomic characters used in the identification of different groups of reptiles.

Birds

Size and shape of the body; structure and dimensions of the bill; number of tail-feathers and length of tail; number of primary and secondary feathers on wing as well as its length; structure and length of tibia; and above all coloration, both colour pattern and absolute colour of different feathers are some of the characters used in the identification of birds.

Mammals

Measurements of various parts of the body; structures and measurement of horns and antlers; number and structure of different teeth; dimensions of different parts of the skull and colour of the fur in different parts of the body are some of the characters used in the identification of mammals.

Documentation

Various faunal works provide keys up to species (up to subspecies in case of birds and mammals). These also provide descriptions of every species involved. Handbooks provide illustrations (both black and white and coloured) in addition to keys. Other publications may contain descriptions, illustrations and/or keys for commoner species of the group of a particular area.

The data obtained by authentic identification of all the specimens obtained during collecting trips at least up to the species, with name(s) of author(s) and year of publications, can be utilised in the preparation of the final list of a particulars group of a country. Besides the specimens actually present before the expert, those present in other museums of natural history and in those of a college or a university whose particulars have not yet been published, should also be taken into consideration.

Likewise, sight records (from the field data of competent persons) as also good photographs with unmistakable identifying characters should also be taken into account in compiling the final list.

Another source of very important information concerning the preparation of the final list is the published literature on the subject. Various catalogues, revisionary studies, descriptions of new taxa, etc. should be consulted for this purpose. To avoid duplicate entries, the question of synonymy should especially be looked into.

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INSECTS

J. K. JONATHAN*

Introduction

Insects constitute the largest animal group in animal kingdom, adapted to all possible ecological niches. Insects, as contrary to popular conception, are more beneficial than injurious; they produce honey, bee wax, silk and dyes. Besides these, they act as pollinators, predators and parasitic on other harmful insects, and serve as food for many higher animals like fishes, toads, lizards and birds.

The Class Insecta is one of the major classes of phylum Arthropoda.

Phylum Arthropoda - derived from Greek (Arthros - jointed, podus - foot) jointed legs or jointed appendages. Some important characteristic feature are :

a. There is no internal skeleton of bones as in birds and instead the cuticle (skin) is hardened into a chitinous exoskeleton to which the muscles are attached. The exoskeleton or cuticle protects as well as supports the soft parts of the animal. This cuticle is shed in immature stages number of times to allow the animal to grow. The phenomenon is called moulting or ecdysis.

b. In order to make it possible for the arthropod to move, the cuticle is divided into a large number of separate plates or armour. Body is divided into a number of segments. Each segment has body parts are between the segments are attached jointed limbs or appendages or legs. Wings arise from the lateral side of the body.

A centipede differs from this general plan-having large number of segments, each with a pair legs and no wings. On the other hand, to identify a spider according to this plan would obviously call for a great deal of alteration, merging some segments together and remodelling other. In addition to these two characters, the method of breathing by means or *tracheae*, which open to the air by a series of opening called spiracles. (Respiration through general surface, by gills in aquatic forms, tracheae or book lungs in terrestrial forms).

Phylum Arthropoda is usually divided into five major classes

1. Crustacea - Crabs, lobsters, shrimps etc.
2. Diplopoda - Millipedes
3. Chilopoda - Centipedes
4. Arachnida - Spiders, mites, scorpions, ticks
5. Insecta - Insects

* Zoological Survey of India, Calcutta.

Characters of Class Insecta (or Hexapoda)
(Hexa = six, poda = feet, six legged)

1. The segments of the body are arranged into three groups

   a. The head consisting of 6 segments - all fused together. The corresponding appendages are modified into one pair of *antennae,* three pairs of *mouth-parts.*

   b. The thorax - consisting of three segments, nearly always with the corresponding three pairs of legs and often with wings on one or both the second and third thoracic segments.

   c. The abdomen, generally legs, but the appendages of the last two or three segments modified into genitalia.

2. Six (three pairs) of true jointed legs, though some times false legs, or Pseudopods are present (as in caterpillars).

3. Wings are found in insects, though not all insects have them.

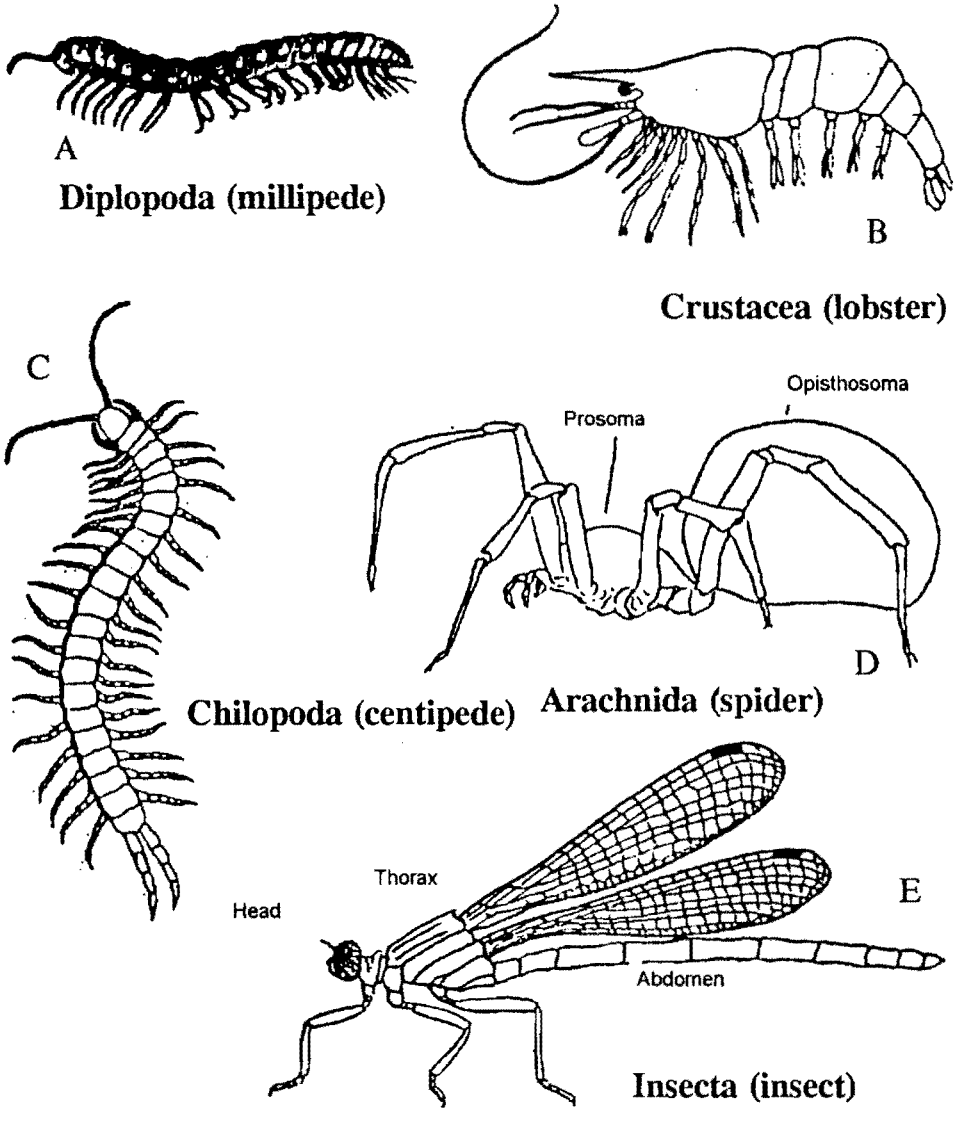

Fig. 14 - **Diversity in Phylum Arthropoda**

Classification of Class Insecta

Sub-class Apterygota (Wingless Insects)
(Young ones resembles adults)

S. No.	Order	Common Name	Size	Habitat	Approx. number of species in the world
1.	Thysanura	Bristle tails	6 mm - 12 cm	Book shelf, under stone	700
2.	Diplura		Very small to 4.6 cm	Soil, Moss, rotten wood in damp habitat	350
3.	Protura	Telson tails	less than 2 mm.	Moist soil, decaying leaves under stone, bark	Less than 100
4.	Collembola	Spring tails	0.2 - 6 mm	Soil, Humus layer, Moss, Caves, on snow, in termite nests	2000

Classification of Class Insecta

Sub-class Ptergota (Winged Insects)

(Wings develop as pads, externally, larvae resemble adults but differ in habitat (Exopterygota : Orders 5-17). Wings developed internally, larvae usually differ from adults and have a resting pupil stage (Endopterygota : orders 18-26).

S. No	Order	Common Name	Size	Habitat	Approx. number of species in the world
5.	Dictyoptera	Cockroaches	6 mm - 7.2 cm	Green Tropical vegetation, Domestic habitats	22,500
6.	Orthroptera	Grasshoppers, Stick insects, Crickets	2.4 - 3.1 cm	Open grassland, Foliage and trees	14,400
7.	Dermaptera	Earwigs	5 mm - 4.8 cm	Under stone, crevices, Garbage	1,100
8.	Plecoptera	Stoneflies	4-5 cm	Around water source	1,500
9.	Isoptera	Termites		Decaying logs, Mound, Ant's nests, under stone	1,900
10.	Embioptera	Webspinners	1.2 cm	Live in silk tunnel, in ground under stone, debris	150
11.	Ephemeroptera	Mayflies	3 mm - 3 cm	Near water	1,500
12.	Odonata	Dragonflies, Damsel flies	2-15 cm	Near water source, among tall grass and river bank	5,000
13.	Zoraptera (Corrodentia)		3-7 mm	Under bark, decaying wood	20
14.	Psocoptera (Corrodentia)	Book lice	2.5 mm	As above	1,100
15.	Phthiraptera (Mallophaga, Anoplura)	Sucking lice, biting lice	5-6 mm	On birds and mammals	3,100

S. No	Order	Common Name	Size	Habitat	Approx. number of species in the world
16.	Thysanoptera	Thrips	5 mm	Arial parts of flowe and plant galls	3170
17.	Hemiptera	Nugs	2-9 cm	Plant parts and water bodies, or parasitic on birds & mammals	55000
18.	Neuroptera	Alder flies, Lace wings	2 mm - 4 cm	Shurbby bush, hill forests, fresh water sponges	5000
19.	Mecoptera	Scorpion flies	1.5-2 mm	Vegetation but feed on insects	350
20.	Trichoptera	Caddis flies	12-75 mm	Near water and damp moss	4450
21.	Lepidoptera	Butterflies, Moths	20 mm - 27mm	Vegetation, damp soil flowers	112000
22.	Coleoptera	Beetles	0.5 mm - 15 cm	Wood, bark, fungus, and nests, Vegetation, flowers ect.	277000
23.	Strepsiptera	Stylops	1.5 - 4.0 mm	parasitic on Hemiptera and Orthoptera	300
24.	Hymenoptera	Bees and Wasps	2.0 - 36 mm	Tree-nests, flowers, vegetation	103000
25.	Diptera	Flies and Mosquitoes		Vegetation, Garbage dump, near or in water, within leaves, parasitic on insects	85000
26.	Siphonaptera	Fleas	1.5 - 4.0 mm	On bodies of host viz. birds and mammals	1100

Identification

As a first step in identifying an insect specimen, it may be useful to separate or distinguish them in different orders.

The insects can easily be distinguished into two groups:

A. Winged insects
B. Wingless insects

A. Winged Insects

1. Insects with 4-wings (2 pairs) 2
- Insects with 2-wings (1 pair). True flies. (Fig.16, P) ..... Order Diptera

2. Wings covered with scales. Butterfly and moths. (Fig.16,A) ..... Order Lepidoptera
Wings not covered with scales 3

3. Only hind wings used for flight. fore wings partly or entirely horny, used as covers of hind wings 4
- Both pairs of wings membranous and used for flight. 7

4. Mouth parts tube-like a adopted for piercing and sucking. Bugs. (Fig 16, B) ..... Order Hemiptera
- Mouth parts adapted for biting and chewing 5

5. Wing venation of fore and hind wings similar. Fore wings stiffer and serves as covers. Grasshoppers. (Fig.16, C) ..... Order Hemiptera
- fore wings without veins, modified into hard covers of hind wings .....

6. Fore wings without short. Tip of the abdomen with forceps. Earwings (Fig.16,D) Order Dermaptera
- Fore wings nearly always long, covering abdomen, enclosing hind wings. Beetles. (Fig.16,B) ..... Order Coleptera.

7. Wings narrow without veins, fringed with long hairs. Small insects 95 mm). Thrips. ``(fig.16,F) Order Thysanoptera Wings fully developed 8

8. Hind wings much smaller than fore wings 9
- Hind wings similar in size to fore wings 12

9. Abdomen with 2 or 3 long tail-like appendages. Mayflies. (Fig. 16, G ..... Order Ephemeroptera
- Abdomen without tail-like appendages 10

10. Wings hairy. Caddisflies, (Figs. 16.H) Order Trichoptera
- Wings not hairy .... 11.....

11. Body size less than 6 mm. Tarsi 2 or 3 segmented. Book lice. (Fig. 16.1) .... Order Pscocoptera
- Body often large, Wasp or Bee-like. Tarsi 4 or 5 segmented. Wasps, Ants and Sawflies. (Fig. 16,J) Order Hymenoptera

12. Tarsi 3 or 4 segmented 13
- Tarsi 5 segmented 15

13. Wings with a few cross-veins. Hind wings greatly expanded posteriorly. Stoneflies. (Fig.16,K Order Plecoptera
- Wings usually with several cross-veins. Fore and hind wings very much similar in size 14

14. Small insects with long antennae. Wings folded flat over body. Termites. (Fig.16.L) .... Order Plecoptera
- Large insects short antennae. Wings held away from body when at rest. Dragonflies. (Fig.16,M) .... Order Odonata

15. Mouth-parts beak-shaped. Scorpionflies. (Fig.16,N).. Order Mecoptera
- Mouth-parts short. Lacewings. (Fig.16,O) ..... Order Neuroptera

(B) Wingless Insects

1. Parasitic forms, living on warm-blooded animals, or found closely associated with them 2
- Not parasitic on warm-blooded animals, free living, or parasitic on other insects, snails etc 4

2. Body flattened from side to side, bristly, with strong legs for jumping; Found on birds and mammals. Fleas. (Fig.15,A) ..... Order Siphonaptera
- Body either rounded or flattened dorso-ventrally 3

3. Mouth-parts chewing-type. Chewing lice (Fig.15,B) Order Mallophaga
- Mouth-parts sucking type. Sucking lice. (Fig.15,C) ..... Order Anoplura

4. Mouth-parts not visible 5
- Mouth parts clearly visible 6

5. Abdomen with a forked appendage "spring" near tip of abdomen, Spring-tail (Fig.15,D) .... Order Collembola
- Abdomen without spring, but with long cerciat tip. Bristle tail. (Fig.15,E) .....(Order Thysanura

6. Mouth-parts sucking-type 7
- Mouth-parts chewing type 8.

7. Snout or proboscis small. Body long and narrow. Claws absent.

Thrips. (Fig.15,F) Order Thysanoptera
- Snout long, jointed Body oval, Claws present, Appphids, (Fig.15,) G) .... Order Homoipetra

8. Abdomen strongly constricted at base into a 'waist'. Ants; wingless velvet ants. (Fig.15,H) .... Order Hymenoptera
- Abdomen not constricted into a waist 9

9. Minute soft insects. Book lice. (Fig 15,I) ..... Order Psocoptera
- Small to large, usually hard bodied insects. 10

10. Tarsi 4 segmented. Pale, soft insects living in wood or soil, Terimintes (Fig.15,J) ..... Order Isoptera
- Tarsi 5 segmented. Highly coloured insects, living in open, but not in wood. Stick-and leaf Insects (Fig.15,K) ..... Order Orthopetra

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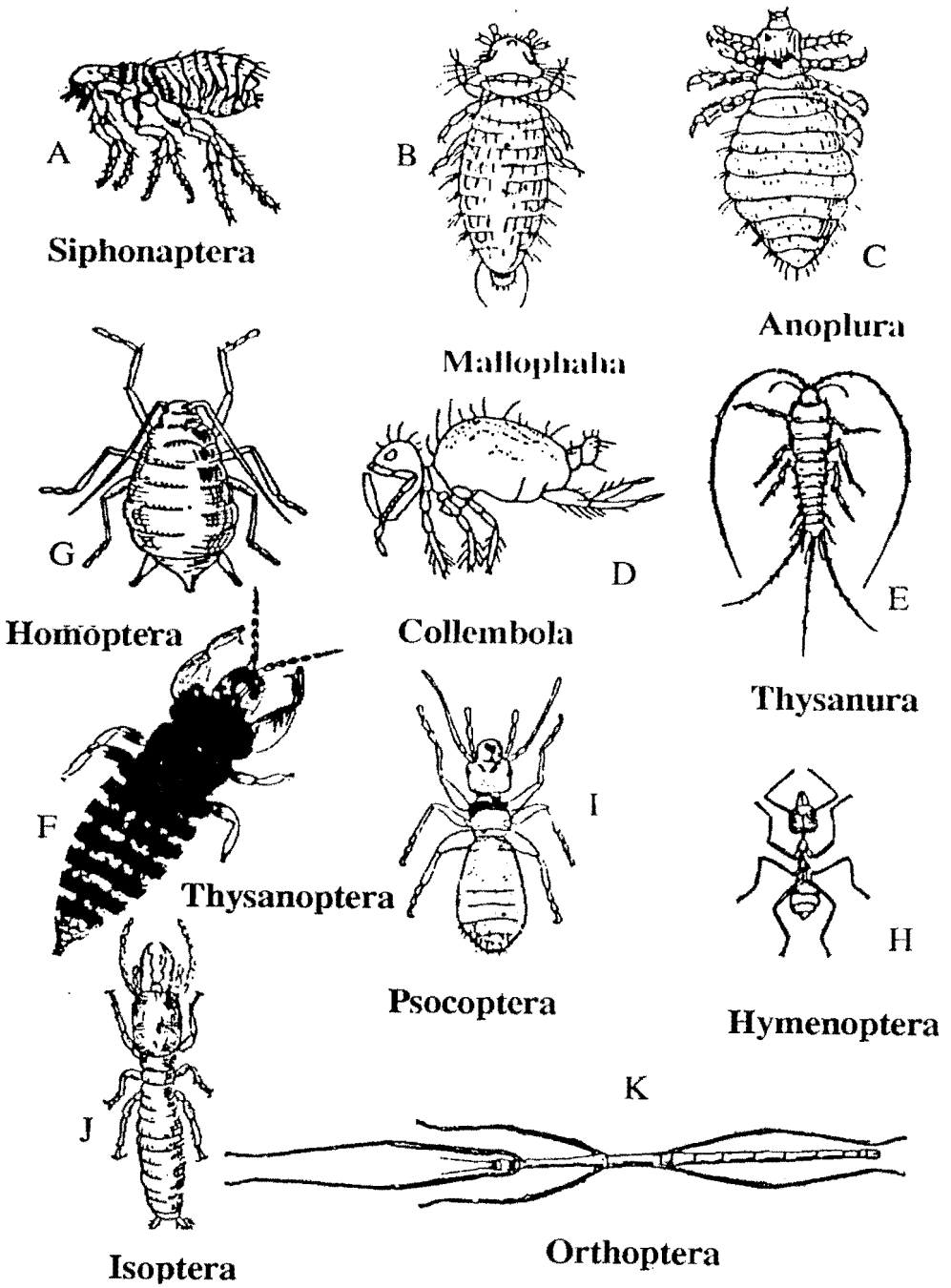

Fig. 15 - **Insect Orders (Wingless forms)**

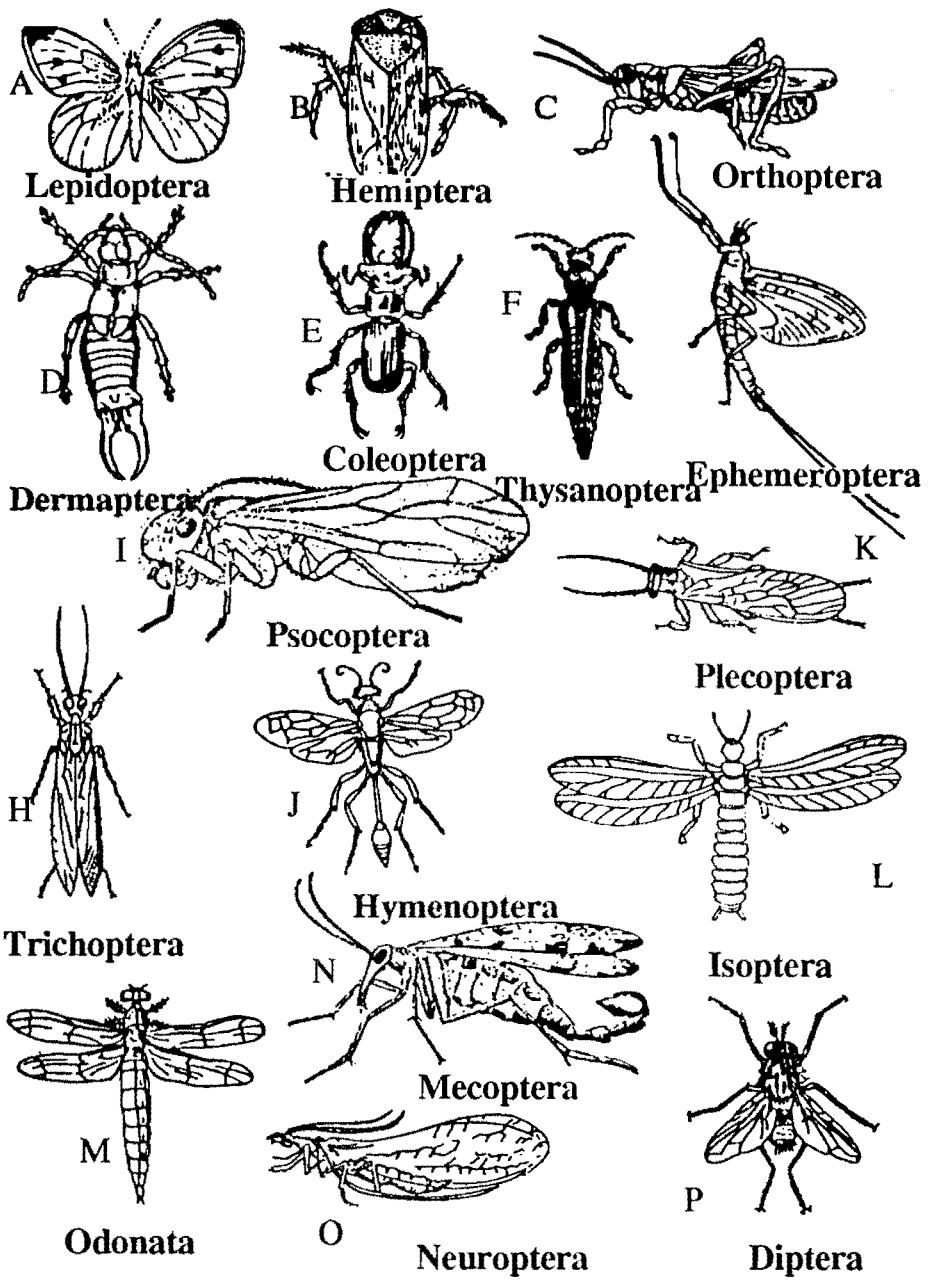

Fig. 16 - **Insect Orders (Winged forms)**

INVERTEBRATES

(Excluding Arthropods except Crustacean)

N. V. SUBBA RAO

Introduction

Animal Kingdom is classified into 32 phyla and out of these except the phylum Chordata all others fall under the category of Invertebrata. This general grouping of animals into Vertebrata and Invertebrata is based on the presence or absence of a vertebral column. Some of these invertebrate phyla are very large and include a few thousands to million of species. Phylum Arthropoda is the largest followed by Mollusca, Nematoda, Platyhelminthes etc. Only Crustacea among arthropods is included here. World over a total of about 1.17 million species of invertebrates are known and about 74,000 species are so far known from India (out of a total of about 82,000 species).

All the invertebrate phyla have not received equal attention from zoologists. Considerable basic data exist on sponges (Porifera), Cnidaria, especially corals, Platyhelminthes, Mollusca, Annelida, Echinodermata etc. and on Crustacea as shown in Table - 1.

Table - 1. Invertebrate Phyla

	Name of the phylum	Common name	No. of species of India
1.	Porifera	Sponges	519
2.	Cnidaria	Jelly fishes, Sea anemones, Sea fans, Corals etc.	850
3.	Ctenophora	Comb Jellies	10
4.	Platyhelminthes	Flat worms, Liverfluke, Tape worm	1650
5.	Rotifera	Wheel animalcules	310
6.	Gastrotricha		88
7.	Kinorhyncha		
8.	Nematoda	Round worms	2850
9.	Acathocephala	Spiny headed worms,	110
10.	Mollusca	Snails, Slugs, Clams, Mussels, Oysters, Cuttle fish etc.	5050
11.	Annelida	Earth worms, Bristle Worms, Leeches	480

12.	Onychophora	Peripatus	1
13.	Arthropoda	Insects, Spiders, Ticks, Mites, Prawns, Crabs etc.	60383
14.	Sipuncula	Peanut worms	38
15.	Echiura	Spoon worms	33
16.	Phoronida		3
17.	Bryozoa	Sea mats	170
18.	Entoprocta		10
19.	Brachiopoda	Lamp shells	3
20.	Tardigrada		
21.	Chaetognatha	Arrow worms	30
22.	Echinodermata	Star fishes, Sea urchins, Brittle stars, Sea lilies Sea cucumbers	765
	Total		**73,842**

The following phyla are either insufficiently known or so far not reported from India and adjacent countries. Hence, these are omitted from the discussion.

1.	Mesozoa	:	Endoparsites of marine invertebrates.
2.	Nemertinea	:	Ribbon worms, Majority marines.
3.	Nematomorpha	:	Horsehair worms. 20 to 93 cm Freshwater, terrestrial and a few marine
4.	Gnathostomulida	:	Very small. In anaerobic marine mud.
5.	Loricifera	:	Marine. In interstitial sand.
6.	Priapulida	:	Marine.
7.	Pentastomida	:	Parasites of tropical reptiles
8.	Pogonophora	:	Beard worms. From depths of oceans.

In general most of the collections include free living forms belonging to Porifera, Cnidaria, Mollusca, Annelida, Crustacea (of Arthropoda) and Echinodermata. Other groups can be collected by only experts or those who have some knowledge on the subjects as special techniques are involved in their collection.

At the global level, phyla with more than 10,000 species are Cnidaria, Platyhelminthes, Nematoda, Annelida, Mollusca and Arthropoda. Phyla Porifera, Rotifera, Bryozoa and Echinodermata include between 2000 and 6000 estimated species. The rest of the phyla are small and include only a few to hundred species.

Classification

Animals fall into two broad subkingdoms, Parazoa of Eumetazoa. The former include sponges (Porifera) with no digestive cavity and with the body wall pierced by pores. The latter category include animals with a digestive cavity and unperforated body wall.

Invertebrates range in size from a few millimetres to more than a metre in length and can be differentiated into microscopic forms. Microscopic forms mostly belong to free living Rotifera. Gastrotricha, Kinorhyncha, Chaetognatha and parasitic Platyhelminthes. Nematoda, Acanthocephala, and Planktonic groups under class Crustacea. But majority of the species are macroscopic and can be seen with a naked eye. Systematic position of a group at higher level can be decided by looking at 5 characters given below:

1. Symmetry: Animals generally have either bilateral or radial symmetry. Cnidarians and adult echinoderms belong to the latter category. In these planes around a median axis through the mouth divide the animal into radial sectors. Members of most other phyla are bilaterally symmetrical. In this length wise vertical plane divides the animal into two equal and opposite halves. These animals have anterior and posterior ends, and dorsal and ventral surfaces.

2. Segmentation: Linear repetition of body parts is known as segmentation (Metamerism). Each segment is called a somite. The segmentation is conspicuous both externally and internally in annelids, but it is mostly external in Crustacea (Arthropoda). Majority of the phyla are without segmentation and they have to be differentiated using other characters.

3. Appendages: Many animals have appendages that protrude out. These appendages are useful in locomotion, feeding and defence. There are tentacles around the mouth region as in Cnidarians, snails (Mollusca), Phoronida, Bryozoa, Entoprocta, Spiuncula, Brachiopoda etc. Some may have a proboscis, which may be either retractile or not as in Spiuncula and Echiura respectively. Crustacea have two pairs of antennae and jointed legs.

4. Skeleton: Majority of the invertebrates have a skeleton for support or protection. These skeletal materials may be in the form of a shell or carapace, which may be internal (starfish, echinoderm), cephalopods (Mollusca) or external (Coral, Cnidaria) Mollusca, crab (Crusacea). In some the skeletal material is in the form of spicules embedded in the some the skeletal material is in the form of spicules embedded in the body (sponges Porifera, Holothurida). The skeleton may be either organic or inorganic.

5. Body cavity: Majority possess a body cavity, which is known as coelom. Animals are divided into three categories based on the structure of coelom. They are acoelomate, where body cavity is absent, pseudoceolomate, in which the cavity is not lined by peritoneum and coelomate in which a true body cavity exists and lined by peritoneum. This structure of the body plan is however, noticeable only on making a transverse section of the body.

Table 2. Some Characteristics of the Principal Phyla of Invertebrates

Symmetry	Digestive trace	Excretory organs	Coelom	Circulatory system	Respiratory organs	Segmentation		
							Phylum	Distinctive features (Exceptions omitted)
variable	Incomplete	-	-	-	-	-	PORIFERA	Body perforated by pores & canals
Radial		-	-	-	-	-	CNIDARIA	Nematocysts; digestive sac-like
Biradial		+	-		-	-	CTENOPHORA	Comb plates for locomotion
		+	-	+	-	-	PLATYHELMINTHES	Flat, soft; digestive trace branched or absent
		+	-	+	-	-	NEMERTINEA	Slender, soft, ciliated soft proboscis
		+	Ps	-	-	-	ROTIFERA	Microscopic cilia on oral disc
		+	Ps	-	-	-	NEMATODA	Only longitudinal muscles, tough cuticle, no cilia
		-	+	-	-	-	BRYOZOA	Grow as moss-like or encrusting colonies
		+	+	+	-	-	BRACHIOPODA	Dorsal and ventral shell; a fleshy stalk
Variable		+	+	+	+	-	MOLLUSCA	External limy shell of 1 2 or 8 parts, or none; segmentation rate, fleshy lobe, the mantle, covering body
		+	+	+	+	+	ANNELIDA	Slender, of many alike segments; fine setae as appendages
		+	+	+	+	+	ARTHROPODA	Segmented, with jointed appendages; exoskeleton containing chitin
		+	+	-	-	-	CHAETOGNATHA	Small; arrow-shaped, transparent, lateral fins
Radial		-	+	+	+	-	ECHINODERMATA	Adults symmetry 5-part radial; tube feet; spiny endoskeleton, larvae bilateral

+ Present, - Absent; Ps Pseudocoel (After Storer & Usinger)

Systematic position of different families may run into a few thousands; hence, not given here. For data on families, genera and species one has to consult monographs pertaining to the particular phylum. Parker (1982) gives a detailed account upto families for various living organisms.

Identification

In India, terrestrial invertebrates are represented by about 4400 species. Of these majority belong to parasitic groups of Platyhelminthes and Nematoda, and a few free living forms of the latter. On the basis of ecological characters, organisms can be grouped as follows: 1. Land 2. Fresh water 3. Marine and estuarine and 4. Parasitic.

Land:

The following data are useful in further identification of the material:

(i) General shape of the body: The body may be worm-like and elongated as in Turbellaria (Platyhelminthes), Nematoda, Annelia and Onychophora.

(ii) Appendages: These include jointed or unjointed legs in Crustacea and Onychophora respectively. There may be a pair of tentacles in the anterio region as in the case of Mollusca (Gastropoda).

Thus, the terrestrial biota can be identified as given below:

A. Worm like body - Turbellaria; Nematoda; Annelida; and Onychophora.

B. Dorsoventrally flattened body - Mollusca (Gastropoda); and Isopoda (Crustacea).

A. 1. Epidermis ciliated, often pigmented with brilliant markings, no anus - Turbellaria (Platyhelminthes).
2. Body slender, very elastic, with a long reversible proboscis and with anus Nematoda.
3. Body segmented .....(Annelida) - Oligochae and Hirudinea.
4. Presence of 15 to 44 pairs of stumpy, unjointed legs .. Onychophora (only one very rare species known).

B. 1. Body soft, with a pair of tentacles in the anterior region and a creeping foot -
(i) without shell..... slugs (Mollusca)
(ii) with shell ..... snails (Mollusca).
2. Body covered with a cuticle, eyes sessile and appendages on the lower side, rolls into a ball when touched ..... Isopoda (Crustacea).

Further identification down to the family level has to be carried out with the help of literature on the particular group.

Freshwater:

It has more phyletic diversity but less species diversity. About 2000 species are estimated to occur in India. The following are the useful characters:

i) Sessile: The animals live attached to some object in water.
ii) Size: A number of organisms are microscopic. The microscopic forms may possess shell or carapace.
iii) Body segmented: Annelida.
iv) Sessile or free swimming with tentacles around the mouth: Cnidaria - Hydrozoa.
v) Sessile, attached to substratum or other objects: Porifera, Bryozoa.

Freshwater biota may be identified as follows:

There are seven phyla, which have their representatives in the fresh water ecosystem.

1. Microscopic: Cnidaria (Hydrozoa), Rotifera, Crustacea (Cladocera, Ostracoda, Copepoda, Conchostraca).

2. Animal can be seen with a naked eye: (i) with shell and creeping foot - Mollusca (ii) with a carapace and appendages - Crustacea (Conchostraca, Decapoda).

3. Animal segmented (i) Without bristles: Oligochaeta.
(ii) With bristles: Polychaeta.

4. Animal with spicules and attached to substratum - Porifera.

5. Animal mat-like or encrusted - Bryozoa.

Marine:

Except phylum Onychophora all other invertebrate phyla are either exclusively or partly marine.

Indonesian region is one of the world's richest area in the number of species. It is considered as a faunistic centre from where other centres in the Indo-Pacific region have got their faunas. The islands of this region-Borneo, Sumatra, the Celebes and others represent the remains of the great sunken land mass that once connected Australia with southern Asia. The areas near this faunistic centre have rich fauna. Thus, Andaman and Nicobar Islands have rich marine fauna and to a lesser degree along the coast of Bangladesh, Sri Lanka and Maldives.

Within the marine ecosystem the groups are evenly distributed. Plankton collections include only adults of few specialized groups such as Cnidaria, Ctenophora, Heteropods and Pteropods (Mollusca), Crustacea and Chaetognatha.

Meiofauna has species of Hydrozoa (Cnidaria), Archiannelida (Annelida), Gastrotricha, Kinorhyncha, Nematoda, Tardigrada, Crustacea and Mollusca (a few opisthobranch species without a shell).

Benthic samples include a number invertebrates such as sponges (Porifera), sea anemomes, corals (Cnidaria), Entoprocta, Bryozoa, Phoronida, Brachiopoda, Mollusca, Polyhaeta (Annelida), Sipuncula, Echiura, Crustacea and Echinodermata.

The benthic fauna can be differentiated as follows:

1. Animals with a shell or skeleton - Mollusca, Brachiopoda and Echinodermata. Some of the molluscs without a shell can be recognised by their conspicuous colours, a flat creeping foot and two or four tentacles at the anterior end.

2. Animals sessile, mouth surrounded by tenacles... Cnidaria, Bryozoa, Phoronida, Spiuncula.

3. Animals sessile, without any tentacles, Body porous ... Sponges (Porifera).

4. Body with appendages, which are typically biramous, with two pairs of antenna Crustacea.

5. Body segmented with fine bristle-like surface Polychaeta (Annelida).

Keys as Aid to Identification

Keys are available for a few well known groups, such as fresh water sponges, Polychaeta, Oligochaeta (in part) and Hirudinea of the phylum Annelida; and for fresh water molluscs, Copepoda (Crustacea) etc. in the *Fauna of India* volumes. Gosner (1971) had provided keys up to orders for marine and estuarine invertebrates. Although not dealing with Indian fauna, the book provides a valuable guide to the identification of marine invertebrates.

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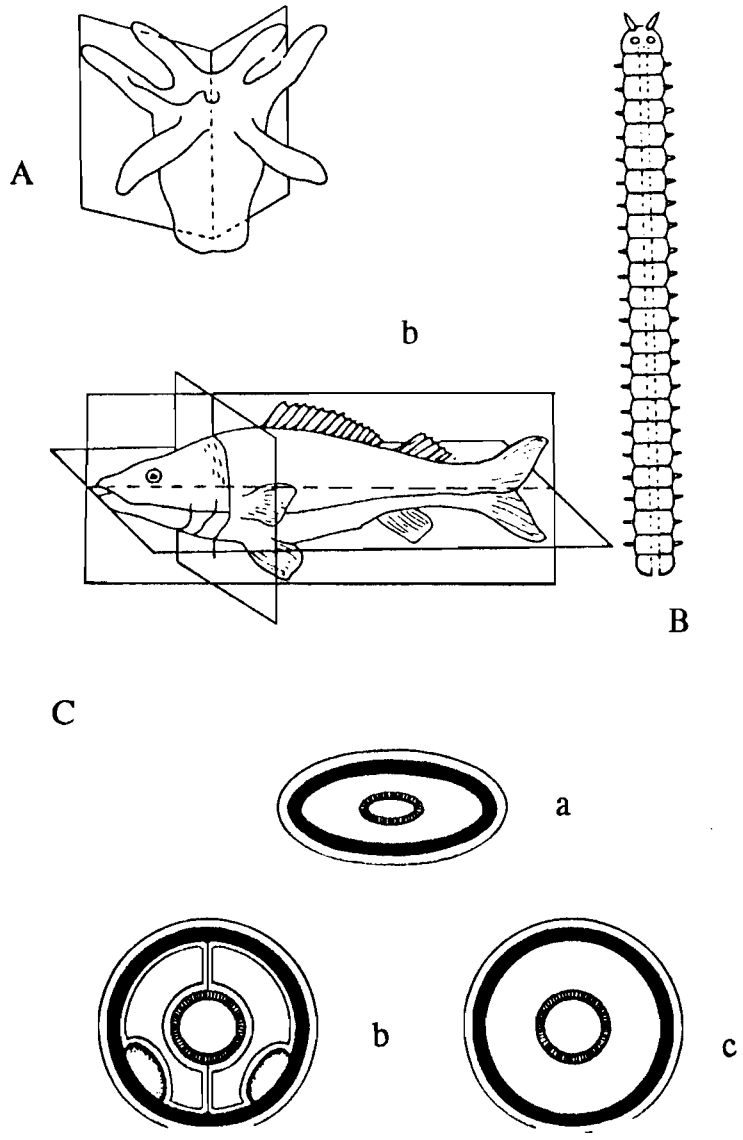

Fig. 17 - **Plans of Animal Bodies**

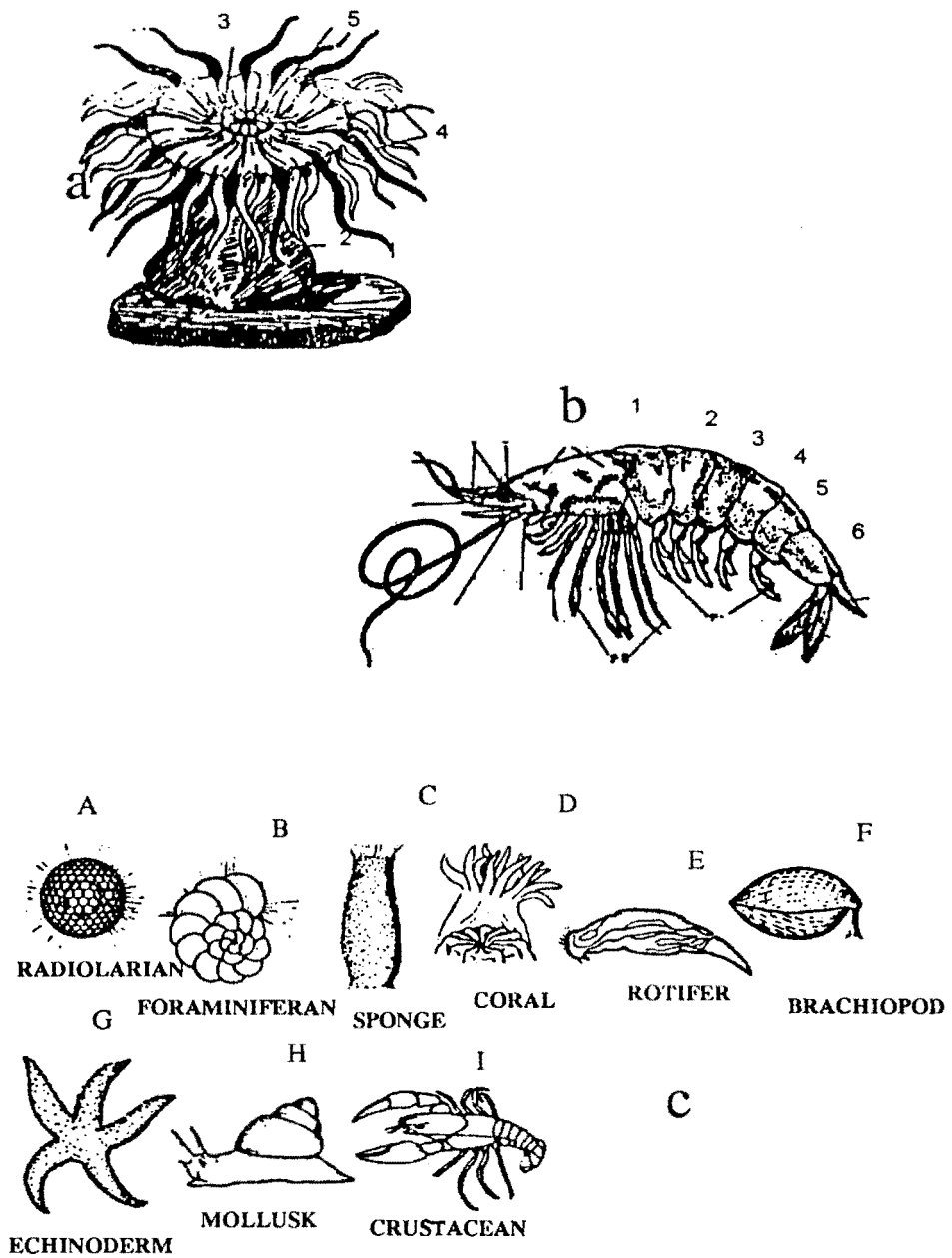

Fig. 18 - **Diversity in Invertebrates**

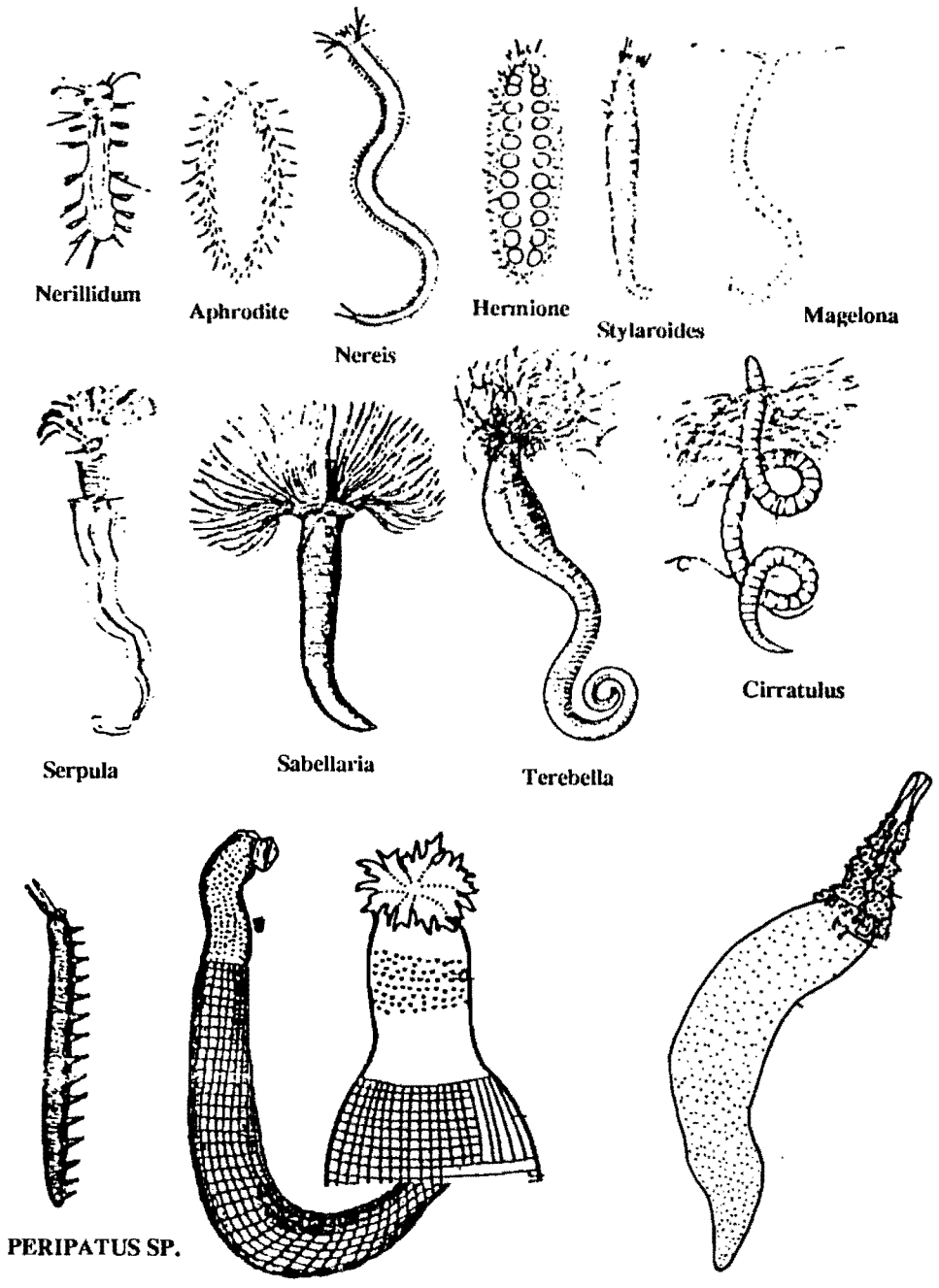

Fig. 19 - **Diversity in Invertebrates**

PART V

SELECTED TECHNIQUES

REMOTE SENSING TECHNIQUE FOR ASSESSING THE COASTAL AND MARINE ENVIRONMENTS

P. CHAKRABORTI*

Introduction

Remote sensing is the science or technology to collect thematic and geometric information of an object without having any physical contact with the object. In the present context, the term Remote Sensing is restricted to identify various objects on the earth's surface using visible (0.4 to 0.7 μ.), infrared (0.7 to 5μ) thermal infrared (8 to 14μ) and microwave (0.1 to 30 cm.) regions of solar radiation (EMR) which is reflected, emitted or scattered by the objects depending on its physical and chemical properties. The remote sensing techniques are classified into two types depending on the source of energy which illuminate the object under study viz.i) passive remote sensing and ii) active remote sensing. In passive remote sensing system, the naturally radiated or reflected energy from the earth's surface is measured by the sensors operating in different spectral bands on board the air-borne/space-borne platforms. An active remote sensing system supplies its own source of energy to illuminate the objects and measures the reflected energy returned to the system.

System Overview

The essential pre-requisites for collection of remote sensing data are:

A. Platform and B. Sensor

Platform:

Ground borne	High altitude (> 15 km from earth's surface)
Air borne	Low altitude (<15 km from earth's surface)
	High altitude (36,000 km above Geostationary e.v. INSAT)
Space borne	Low altitude (700-900 km.) above Sun synchronous e.g. Earth resources satellite e.g. (IRS, SPOT, LANDSAT, NOAA, ERS-1).
Sensor	Image forming
	Non-image forming

We are more concerned about the image forming sensors.
Image Forming Sensor

Active (e.g. microwave radar).
Passive (e.g. Photographic camera, Television camera, Optical scanners-normally in the atmosphere windows).

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These sensors are being used in Air-borne or Space-borne platforms to assist in mapping, inventorying or monitoring of earth resources. In case of Air borne survey we are getting direct photographs of the earth's surface with overlapping portions (minimum 30% forward overlap). Two photographs with such overlapped portions are called 'stereo-pairs'/'stereo-mates' and are used under stereoscope for getting 3D vision.

The aerial photographs (black & white) are available in different scale ranging from 1:60,000 to 1:10,000. The scale of A. P. depends on the flying height of the aircraft and the focal length of the camera used.

S=H/f, where S=scale of the photograph, H=flying height and f=focal length principle distance of the lenses.

In general, three types of aerial photography are used for various purposes:

a)	Vertical Photographs;	b)	Orthophotos and
c)	Oblique photographs;		High angle
			Low angle

In India, Survey of India is the controlling authority for airphotos.

The satellite data is available in digital form based on 'gray value variations' (DN values ranging from 0-255) and is supplied on CCT's (Computer Compatible Tape). The satellite imagery or hard copy are prepared in the laboratory using CCT data. In general, the scales of data products vary from 1:1 million to 1:50,000. The receiving station for satellite information is at Sadnagar, Hyderabad. The National Remote Sensing Agency (NRSA), Deptt. of Space, Govt. of India is the authority for satellite data.

Hard copy imagery are available in Panchromatic (B&W) mode as well as in False Colour Composite (FCC) mode. The CCT's are also used in the Digital image processing system. The imagery of different types of earth observing satellites available in India are IRS, LANDSAT, SPOT (both in B & WW and FCC), NOAA and CCT's.

Based on the tone, texture, pattern, shape/orientation etc. the different feature are identified in aero-space products by the interpreter.

In recent decades, with the advancement of the scientific and technological world, the aero-space data products especially the space-based earth observation system (i.e., satellites) provides more "operational" information due to its repetitive cycles which varies from 4 days in case of NOAA to 26 days in SPOT. The LANDSAT series of satellites have a repeat period ranging between 16 to 18 days whereas, in case of IRS it is 22 days. Presently, in Indian context we are having IRS-1A and IRS-1B with 11 days repeat period as a whole. The resolution of the satellites are also varying from microwave sensors on board SEASAT, of them only the AVHRRA (NOAA) is still in service where as the CZCS and SEASET ceased operation after eight years and 3.5 months respectively. All these satellites operate in visible and IR regions of EMR due to which they are not capable to supply desired information during heavy overcast or when atmospheric cloud cover is present i.e. they are not having all weather and all time capability. To overcome this problem a new generation of microwave satellites have been introduced in the 1990's. Since 1991 three such radar satellites viz. ERS-1 (European), JERS-1 (Japanese), ALMAZ (Russian) are on board.

In India, the ERS-1 data are available both in Photographic (B&W) and digital modes. It may be mentioned here that India has also conceptualised of developing a dedicated ocean satellite (viz. "OCEASNSAT") to meet the operational requirements of various oceanographic applications especially to increase the fishery potential of EEZ.

Basically, the information delivered by the operational earth observing satellites are used by two user communities viz.

i) Scientific users: whose role is to develop and maintain the knowledge base, which is scientific and technological foundation of entire system.

ii) Operational users or thematic users: who participate in the economy directly and whose output has direct economic value e.g. earth scientists, engineers, planners are customers who can utilise the thematic information extracted from the RS data.

Application of RS in Coastal & Marine Environments

In terms of actual applications of remote sensing in the fields under consideration there are three main elements for which it plays important role:

I. By providing baseline data on coastal geomorphology and habitats.

II. In studying the features related with the waters e.g. sediment plume (turgidity), interacting of wind and wave bottom topography, currents and sea surface temperature for locating potential fishing grounds in the offshore.

III. In detecting, mapping and measurements of pollutants in broad sense.

I. Coastal Geomorphology and Habitats:

In the present day scenario, in comparison to the Survey of India (SOI) toposheets, photo-mosaics and/or up-to-date geocoded satellite data products of similar scale (i.e. 1:50,000) provide more quantifiable information about the morphology, morpho-arrangement, morpho-genesis and constituent materials of the different landform units as well as the existing land cover/land use pattern at a glance to the users.

In coastal zone/estuarine areas the common landforms/mapping units are:

a) Marine cliffs and notch zones.
b) Marine wave cu platforms
c) Beaches, beach ridges/runnels, spits and tombolo bars,
d) Coastal dunes (active, inactive or dormant),
e) Vegetated/Non-vegetated tidal sandy/mud flats,
f) Marine terraces/flood plains,
g) Coral reefs/reef flats/reef caps/uplifted reefs,
h) Ramparts and cays,
i) Lagoons,
j) Fluvio-deltaic flats.

In response to the repetitive coverage, multi-seasonal or time-critical satellite imagery could be used to monitor the spatial variation of the aforesaid landform units/ mapping units, the fluctuation of the inundated areas of the tidal flats or the nature of the tidal creeks through seasons. Change detection in the mangrove forest boundary or deforested patches could also easily be identified in the RS data products, as the broad leaf, healthy mangroves have their very characteristic reflection pattern. It may be mentioned here that the range of scales for surveying and/or resolution of the RS data are also to be subject specific i.e. varies with the nature of the project undertaken, for instance, monitoring of the shore line changes requires more resolution than those needed in the study of general landform features. In view of that a 'zooming-in' or multiphase approach is suggested starting from use of geocoded imagery/aerial photographs (on scale 1:50,000) or air photos or digitally enhanced imagery (on scale 1:25,000/20,000/10,000) with subsequent field checks.

II. Study of Oceanic and Coastal Water (Physical) Properties:

The introduction of microwave satellite with the existing operational satellites, increased the potentiality to a great extent in studying the dynamics of coastal waves and currents, surface wind speed, sediment plumes, mapping of bottom topography/ coastal bathmetry (to c.20 mts depth) and identifying the potential zone of fishing grounds in the offshore based on the study of ocean colour, sea surface temperature (thermal gradients/fronts), upwelling zones (result of divergence and offshore currents) etc.

In the visible portion of the EM spectrum, radiation is able to penetrate water surface, to a certain extent, (unlike in the infrared or microwave region) and is reflected by suspended matter present in the water. Sequential imagery may be used for qualitative analysis of the suspended sediments in the areas of enhanced coastal or estuarine erosion.

Planktonic matter in surface water is readily detected by RS technology due to strong spectral signatures of chlorophyll-a, which is the pigment responsible for photosynthesis. Remotely sensed measurement of chlorophyll-a and hence phtyoplankton biomass is well established by various investigators.

Anomalous low salinity and temperature conditions are indicative of upwelling. During upwelling, subsurface waters of relatively low oxygen content ascends to the surface and the surface water may become markedly undersaturated. Upwelling zones are known to be highly productive regions due to the increased supply of nutrients brought into the euphotic zone from below. Upwelling in thermal front regions (Sharp boundaries between water masses by temperature gradient) have high potential as fishing grounds for pelagic species especially tuna. SST and ocean colour maps can thus guide fishing vessels to areas of high potential, thereby saving considerable time and fuel.

The imagery are found to be useful in providing information on bottom topography/bathymetry in highly specific conditions of sea state and currents. And hence are unlikely to provide an operational means for routine mapping of bottom topography or bathymetry mapping.

III. Study of Pollutants in the Marine Environment:

The main aim of the marine pollution studies is to measure the deterioration of marine environment over wide areas and time spans. It is necessary to set realistic limits on the fundamental issue of the international agreements on waste disposal in the sea.

The most common application of RS in marine pollution is the detection and mapping of oil slicks, which are regarded as one of the main sources of pelagic pollution.
Ideally, remote sensing system should be able to detect unknown oil discharges as difference in spectral radiance of ocean water and of a thin layer of crude oil.

The oil film has a noticeably higher radiance in the UV, visible and reflected IR bands. Intensity of reflectance in UV region is high comparable to visible and IR bands. In the visible and reflected IR regions the signature of an oil film is determined by indirect method e.g. the surface tension effect of an oil film, which dampens the small scale surface waves to create calm water or slicks. These slicks, produced by internet waves, typically have dark signatures in contrast to the bright signatures caused by sunlight (sunlight reflected from ripples on the sea surface) from the rougher, clean water.

Digital processing can enhance the appearance of oil slicks on satellite imagery.

Theoretically it is found that the oil films have cool radiant temperature relative to the water temperature and thermal IR detectors are sensitive to detect such differences.

As described earlier, oil on water changes the surface tension and hence under certain sea state and weather conditions can dramatically affect the surface roughness expressed by waves and ripples. Microwave Remote Sensing may be well suited for such change detection as the great benefit lies with the sensors to operate in all weather, day and night time.

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DIFFERENT EARTH OBSERVING SATELLITES

NASA (U.S.A.)	*Launched on*	*Resolution*
ERTS - 1	July 1972	79 Mts.
ERTS - 2 (Renamed as Landsat - 2)	Jan. 1975	79 Mts.
LANDSAT - 3	March 1978	79 Mts.
New Generation Satellite		
LANDSAT - 4	July 1982	79 Mts.
LANDSAT - 5	March 1984	79 Mts.
LANDSAT - 6 Failure		
SEASET (First Satellite designed for oceanography)	June 1978 Failed on Oct. 1978	
NIMBUS - 7	October 1978	825 Mts.
NOAA Satellite with AVHRR	Since 1970's	1.1 km.
French Satellites		
SPOT - 1	February 1986	PAN 10 Mts. MLA 20 Mts.
SPOT - 2	January 1990	PAN 10 Mts. MLA 20 Mts.
Indian Satellites		
Experimental Satellites		
BHASKARA - I	June 1979	
BHASKARA - II	November 1981	
Operational Satellites		
IRS - 1A	March 1988	LISS-I72.5 Mts.
IRS - 1B	August 1991	LISS-II 36.25 Mts.
IRS - P_2	October 1994 (By PSLV-D2)	LISS-II 36.25 Mts.
IRS - 1C	Tentative Launch in 1995 (5 days repeat cycle for PAN) (24 days repeat cycle for LISS-III)	PAN 10 Mts. LISS-III 23 Mts.

MICROWAVE (RADAR) SATELLITES

European Satellite		
ERS - 1	27th June, 1991	SAR 25 Mts.
Japanese Satellite		
JERS - 1	11th Feb., 1991	SAR 18 Mts.
Russian Satellite		
ALMAZ	31st March, 1991 SAR (Using a 9.6 cm microwave band)	
Future Microwave Satellite		
RADARSAT		
Future Ocean Satellite of India		
OCEANSAT	PAYLOADS Ocean Colour Monitor (OCM), Wind Scaterometer, Radar Altimeter, Thermal Infrared Monitor (Resolution - 1 km.) Passive Microwave Radiometer (PMR) - Channel (Resolution 9-20 km.) 2 Day Repeativity for OCM 6 Day Repeativity for Scatterometer Sun-Synchronous Orbit at 743 km.	

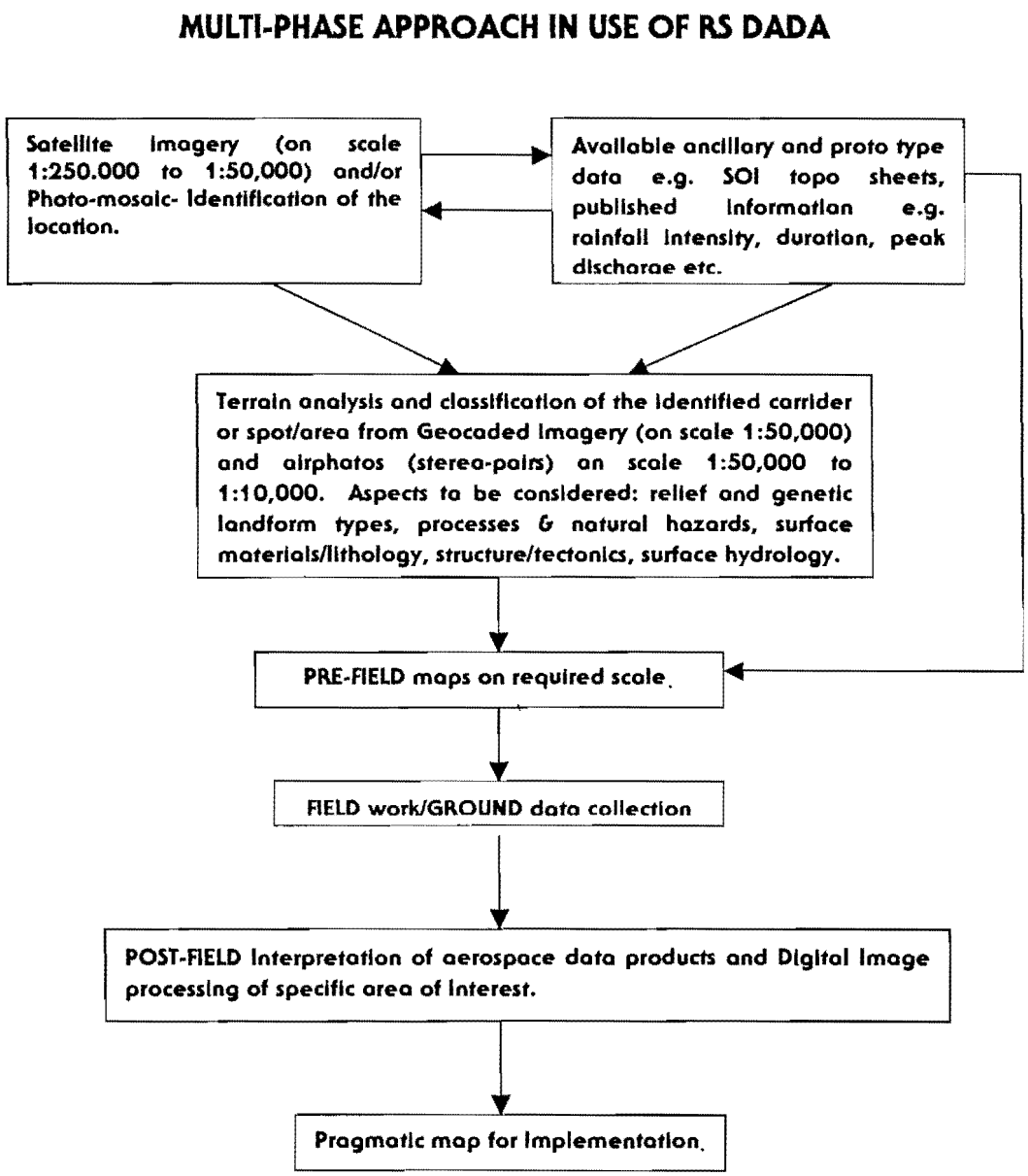

Fig. 20 - Multi- Phase Approaches in Use of RS data

FOREST COVER MAPPING USING SATELLITE IMAGERIES: A CASE STUDY

A. K. RAHA*

Introduction

Remote sensing provides a means for obtaining a synoptic view of the status of forests and condition on real time basis. So far, forest density mapping has been carried out on the basis of tonal variations within a given scene and it has attained operational status in India on 1:1 million and 1:250,000 scale using standard visual interpretation techniques.

The digital processing involve pattern recognition techniques which classify the image data into different forest types and their crown density classes on the basis of radiance information. The difference in the reflectance of different forest crown cover classes basically depend on spatial, spectral, radiometric and temporal resolutions but the differences in spectral radiance of each forest category becomes the basis for the identification and differentiation on satellite image. But quite often similar spectral reflectance by different earth features and dissimilar spectral reflectance by similar earth features create spectral confusion leading to misclassification/interpretation. These problems can be overcome by systematic ground truth information.

A case study was done for three districts namely Darjeeling, Jalpaiguri and Cooch Behar using IRS-1A LISS satellite data (Nov. 1988) in a VAX 11/780 environment with VIPS - 32 software support. With the objectives of:

1. To delineate the following three forest crown cover density classes in sal, mixed forests, subtropical broad-leaved forests and riverine forests:

(a)	Dense/Closed forest	:	density more than 40%
(b)	Open forest	:	density between 10% and 40%
(c)	Degraded forests	:	density less than 10%

2. To segregate all plantations irrespective of density.

3. To map landuse/landcover classes of tea gardens, water bodies, sandy areas, built-up areas and village orchards.

4. To incorporate various forest administrative boundaries as well as cultural features for effective ground control and better forest management strategies.

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Study Area

Darjeeling District

The Darjeeling district with a geographical area of 3,149 sq.km. (26°31'-27°13' N and 87° 59' - 88° 53 E) has three forest divisions namely, Darjeeling, Kalimpong and Kurseong. The shape of the districts is an irregular triangle. The district is bounded by Nepal on South-West, on south by West Dinajpur intercepted by the Mahananda river and on the east by Bangladesh and Jalpaiguri district.

Few places in India command range of variation in Forest types as found in such a small one as Darjeeling, showing trend in altitude differentiation, climatic variations, which have tremendous influence on forest types. The altitude varies from 100 m in the Terai valley to 3000 m on the Singalla roads, springing from the southern face of Kanchenjungha massif.

Climate: The district consists of two distinct tracts, the ridges and the deep valleys of the lower Himalayas and the altitudes vary between 300m to 3600m. On account of the hilly tracts, the rainfall varies considerably from place to place. Kurseong, on the southern slopes of the lower Himalayas gets an annual rainfall of 4050 mm while Kalimpong near the northern border of the district gets 2255 mm. The highest temperature during the summer month is around 33°C in May but at higher elevations the summer is pleasant.

Soils: The rocks are unaltered sedimentary, mostly confined to the hills on the south and different grades of metamorphic rocks over the rest of the area. The soils area generally composed of alluvial, a light sandy loam being the most common.

Rivers: The rivers of the district drain ultimately to the south, through the west to east ridge across it causes a series of Tista tributaries rising on its northern face to flow north-wards and others flow east or west before joining the main river, Brahmaputra. Some of the rivers are Lish, Gish, Chel, Mahanadi, Balaram, Mechi.

Data Used

In the present investigation, the following IRS-1A LISS II data sets pertaining to the period of November 1988 were used:

(a) P/R 19-49A1, A2, B1, B2 (D/P:25-11-88)

The survey of India Topographical maps (78 A, B) on 1:250,000 and 1:50,000 scale along with the aerial photographs on 1:50,000 scale, Working plant maps were used as collateral data.

Jalpaiguri District

The Jalpaiguri district with a geographical area of 6,227 sq.km. (26-27° 16' 0 N and 89° 53' - 88° 25 E) has four forest divisions, namely Jalpaiguri, Baikunthpur, Cooch Behar and Buxa. The district looks like an irregular rectangular and is bound in north by Bhutan and district Darjeeling, on the south the district of Cooch Behar and part of Bangladesh and on the east by Eastern Duars in Assam, which forms part of the right banks of Sankos river marking the boundary.

The vegetation is generally Tropical moist deciduous/semievergreen with sal as the dominant species with associates of mixed nature. The significant feature is mixing of riverine forests with naturally growing mixed type of forests and sal forests.

There are eight wild life sanctuaries in West Bengal, of which the Jaldapara wild life sanctuary with good collection of wild life, occupies the pride of place in the state of West Bengal.

Climate: The climate of the district is characterised by high humidity and heavy precipitation. Nov-Jan are the direst months, Jan-Feb is colder with slight mist; by the end of the March it begins to get warmer and is very hot in April.

Soil: As a whole the district is covered by alluvial deposits except in hilly northern fringes. The alluvium consists of coarse gravel near the hills, sandy clay and sandy loam on further south and some patches of black clay in the areas between Tista and the Jaldhaka.

Rivers: The principal rivers from west are (i) Mahananda (from western boundary) and (2) Tista. Between these two there are small rivers namely Saun, Karatoa, Chaol, Talma, Jamuna, Ponga, Karala.

Data Used

In the present study, the following IRS-1A LISS II data sets pertaining to the period of November 1988 were used:

(a) P/R 18-49A1, A2, B1, B2 (D/P: 24-11-88)

The Survey of India Topographical maps (78 B, F) on 1:250,000 and 1:50,000 scale along with the aerial photographs on 1:50,000 scale, Working Plan maps were used as collateral data.

Cooch Behar District

The district lies between 25°57'40"-26° 32'20" N and 89° 54'33" - 88°47'40" E with a total geographical areas of 3,387 sq.km. The district is bounded on the north by the Jalpaiguri district, east by Assam and south by Bangladesh.

The vegetation can broadly be differentiated into two types: (1) sporadic sal forests with mixed species/teak plantations and (2) Riverine forest.

Climate: The climate of the district is characterised by high humidity and heavy precipitation.

Soil: The soil of the district is of alluvial formation and has a large admixture of sand.

Rivers: The principal rivers beginning from the west are the Tista, Dharla, Jaldhaka, Torsa, Kalijani, Raidak or Sankosh and the Gadadhara. They flow from a north-easterly to a south-westerly direction.

Data Used

In the present investigation, the following IRS-1A LISS II data sets pertaining to the period of November 1988 were used:

(a) P/R 18-49 B1, B2 (D/P: 24-11-88)

The Survey of India Topographical maps (78 B, F) on 1:250,000 and 1:50,000 scale and other Forest Working Plan maps were used as collateral data.

Normal Method

The whole area under study is considered for classification at a time. If for some reason, there is spectral confusion which the interpreter feels might lead to misclassification or if forest vegetation has to be separated from orchards/social forestry area, then the classification has to be accomplished using the stratified approach.

Stratification Method

Stratification is a process of decision making to divide the area under study into two zones as forested area and non-forested area before attempting classification.

The Reserve Forest area mask is used to classify the reserve forest land in Darjeeling and Jalpaiguri districts (layer 1); similarly the area peripheral to the reserve forest is also classified (layer 2). Both these layers are composited.

The conclusion from the above study indicates that launching zone for stratified classification approach depends on general distribution of vegetation type within the scene.

The procedure for generating forest type and density maps through digital classification using maximum likelihood algorithm in VAX-11/78 (VIPS - 32 software) can be divided into four phases as given below.

Phase - 1: Preparation of Ground Truth Base Scene

1. After selecting the proper season, respective Computer Compatible Tapes (CCTs) were procured from National Remote Sensing Agency (NRSA), Hyderabad.

2. The rows of IRS-1A LISS II data were merged after finding out the overlapping area.

3. Transformation models for each scene was generated to rectify satellite data for geometric corrections.

4. The rectified scene was subjected to enhancement for improving the interpretation of image.

Contrast Enhancement: This technique expands the range of brightness values in an image so that the image can be efficiently displayed in a manner desired by the interpreter/analyst. It has also been noticed that the identification of different forest types/density classes needs thorough field knowledge as well as complete understanding of the corresponding image signatures. After keen observation of

certain known areas of different forest categories, one can decide the optimum stretching parameters to locate/identify various earth features correctly.

***Vegetation Index Map*:** Vegetation index is a convenient parameter for expressing the multi-spectral response of vegetation. The vegetation is the only land category known to strongly absorb in visible light (except green band) but to absorb little or no light in the near infra-red spectral region. Spectral reflectance of other materials such as soil or litter generally increases with wave length. Thus the difference of visible and near infra red reflectance represents photosynthetically active vegetation. Therefore, numerous vegetation canopy parameters such as leaf area index, above ground biomass and absorbed photosynthetically active radiation have been correlated with vegetation index. However, these functions are site and species specific, since vegetation index is a function of the entire geometry and optics of the vegetation canopy interface.

The Normalised Difference Vegetation Index (NDVI) is generated by: NDVI = (NIR-R) / (NIR + R). Since the normalisation process minimises the effect of illumination geometry as well as surface topography, it is generally superior to simple ratios.

***Tassled Cap Transformation (TCT)*:** Some workers transformed the original four channel data of IRS-1A LISS II to a new four dimensional space using the Gram-Schmidt sequential orthogonalisational technique. This transformation is called tassel cap because of its shape. The transformation identifies four new axes namely - the Soil brightness index (SBI); the Green Vegetation index (GVI); Yellow stuff index (YUI); Non-snow index (NSI). The first two indices contain most of the information (95.98 per cent). Nearly 98 per cent of the variance in bare soil spectra from several types of soils can be explained by the soil to the brightness index. Normally the soil signature will lie parallel to the brightness index axis and the greenness of the surface is indicated by the orthogonal deviation of the plots from the mean soil line. Farther the location of the plot from the soil line the greater is the vegetation on the ground covered by that pixel.

5. Representative photo-prints of NDVI and TCT along with mosaics of standard FCC outputs would be helpful in selecting ground truth points. The aerial photographs and Survey of India topographical maps can also be used for inaccessible areas and locating the sample areas.

The various steps involved in the phase - 1 are given in flow chart - 1.

Phase - 2: Training the Computer

Supervised classification technique involves "training" the computer to recognise a particular combination of digital number values (DNV) representing the reflectance in each of the different wavelength bands from a forest cover type/land use class of interest.

Classification Strategy

The ideal classification for forest management is one which satisfies the needs of the forest administrative planners with up-to-date information at minimum time, cost and also to improve the ability of the planner and appraise him of the condition, characteristics, the resource potential and the environmental constraints on the management of forests.

In the present study, each vegetation type (e.g. sal, teak) in a forest and other land use categories (e.g. agriculture, sandy areas, water bodies) are subdivided into 4 to 5 sub classes based on digital spectral response. The key to successful and accurate classification lies in defining a set of training areas which are truly representative of that particular cover class under study. The statistical information obtained from each training set is used to calculate the probabilities assigning the unknown pixel to its specific class with the help of Gaussian maximum likelihood classifier.

Spectral SeparabilityTests

Before subjecting to classification, it is pertinent to study and evaluate the spectral separability of the different classes formed by training sets. The following methods are employed to evaluate the spectral response pattern of classes for their spectral separability.

Confusion Matrix

The confusion matrix provides some indication of the spectral separability of different classes within the training set pixels.

Divergence Matrix

Statistical analysis for category spectral response pattern can be carried out for all pairs of classes and results can be presented in the form of matrix called divergence matrix. In general, the larger the divergence, the greater the "Statistical distance" between classes and higher is the probability of correct classification of different classes.

Ellipsoidal Graphical Representation

Based on probability contours displayed on the pericolour terminal between any two bands, it is possible to assess the spectral separability of different training sets.

Since homogenous training sets are expected to be classified accurately than less pure training sets, a decision can be taken for merging of these classes based on the ground realities or to correct the training sets if they are found mixing more with other training set classes.

Maximum Likelihood Classifier

The maximum likelihood classifier uses both the variance and covariance of the training set data to calculate the probability of known pixels occurring in all the classes. After evaluating the probability in each category of class, the unknown pixels would be assigned to the most likely class or labelled as 'reject' if the probability values are all below a particular threshold set by the analyst.

The probability of pixel X belonging to class C_i is given by

$$P(X/C_i) = \frac{e^{(-1/2)(X - M_i)(W_i)^{-1}(X - M_i)}}{(2\pi)^{n/2}(W_i)}$$

where n = No, of bands
M_i = Mean of class C_i
W_i = Variance - covariance matrix

Phase - 3: Preparation off Mask/Cultural Feature scene

Necessary mask files and overlay files were digitised using transformation models and these vector files were converted to roster files. Necessary correction, if required, can be done before extracting or overlaying.

Phase - 4: Preparation of Forest Cover Map showing Type/Density

1. Final district map showing different classes pertaining to forest density within each forest type was extracted using District/Forest Division Mask scene and then subjected to smoothening using mode filter. The overlay files generated in the phase-III were overlaid on to the image.

2. The confusion classes within the Reserve Forest were corrected based on the confusion matrix/divergence matrix/ellipsoidal display generated in the phase-II. After necessary corrections, the reserve forest scene is composited with the original scene.

3. The statistics for the entire district/forest division was generated to show aerial extent of each of the forest density classes under each forest type and other land use classes.

The forest composite map showing forest type/density maps for three districts of North Bengal was generated on 1:100,000 scale. In view of complexity in vegetation types of Darjeeling District, both forest density and type maps were generated separately on 1:100,000 scale.

Accuracy Estimation

The digital classification, especially for a large area has been proved to be useful in view of quick appraisal of status of forest cover, land use/land cover condition and also in depicting the aerial extent of various classes on real time basis. It is pertinent to assess their accuracy by contingency table method or confusion matrix to calculate individual overall accuracy of the classified output.

The qualitative approach is one such method through which the overall classification accuracy can be assessed. This is done by rationing the number of points found correctly on the classified image to that of total number of points checked in the field multiplied by hundred. This gives the total accuracy of the classified output in percentage.

The quantitative approach is one that estimates the amount of confusion among different training sets defined in the ground truth and the classes in the classified output by generating "confusing matrix" to check errors of omission and commission and finally the overall classification accuracy.

On random basis systematic sampling procedure has also been adopted for estimating the overall classification accuracy. For example, a sample grid (5 cm x 5 cm covering an area of 5 km x 5 km on 1:100,000 scale) drawn on a transparent paper was overlaid on photo-mosaic forest division/district and five samples are collected in the field covering each of the categories under study. The same area interpreted from aerial photograph on 1:50,000 scale has been aggregated for making them comparable with the photo-mosaic prepared by digital classification and an error matrix was prepared for each of the individual classes. Lastly overall classification accuracy is estimated based on the matrix.

More than 90 per cent accuracy in delineating various forest density classes under each of the forest type and about 85 per cent in case of land use/land cover categories were achieved in all the above mentioned approaches.

Analysis

The analysis is based on 1988 assessment using IRS-1A LISS II satellite data through digital classification techniques for North Bengal.

(i) Forest covers could be classified and grouped into density classes of dense forest (density>0.4), open forest (density between 0.1 and 0.4) and degraded forests (<0.1).

(ii) Species-wise classification could be made for monoculture plantation of sal and teak species and dominant mixed forests.

(iii) Natural forest could be classified into sal, mixed, riverine, subtropical broad-leaf, coniferous and rhododendron forests.

(iv) The present study on the basis of satellite imageries shows an increase in total forest cover and dense forest cover in the State when compared with the data of Forest Survey of India, Dehradun. They carried out Forest cover mapping of the entire West Bengal through standard visual interpretation techniques using False Colour Composite of Lands at TM with a resolution of 30 m on 1:250,000 scale. In view of interpretability limitations in visual techniques, the forest areas less than 25 hectare could not be detected. However, IRS-1a LISS II data with spatial resolution on 36 m, which has been used for the present study could possibly delineate even smaller than 0.4 ha due to digital approach.

(v) The Statistical data obtained from the present study shows that 25.44 per cent of the total geographical area (12,763 sq.km.) of North Bengal is under actual forest cover, as compared to the 23.91 per cent recorded by State Forest Deptt., West Bengal. If the tree cover in the form of village orchards are also taken into consideration while computing the total vegetation cover in North Bengal, it is seen that nearly 27.79 per cent of the total land area is under actual vegetative cover in North Bengal.

The total forest cover of Darjeeling district alone as recorded by the present study, State Forest Department, West Bengal, and Forest Survey of India, Dehradun was 1609.66, 1204 and 1435 sq. kms. respectively. While that total forest cover of Jalpaiguri district similarly was 1601.73, 1790 and 1537 sq.kms. respectively and finally the forest cover of Cooch Behar in the same way was 34.98, 57 and 33 sq. kms. respectively.

Earlier the recorded forest land of West Bengal has been estimated by taking into consideration the total forest ecosystem (inclusive of river and wetland ecosystem) existing within the notified forest land. It is therefore, in the present study the same area has been included within the forest cover while computing the total forest cover. In view of the understanding the concept of total ecosystem while assessing forest cover, the total percentage of forest cover as per the State Forest Department was 23.91 per cent and by the present study is 25.44 per cent. however for the sake of comparison with the Forest Survey of India, Dehradun, the total forest cover within the North Bengal would be 22.23 per cent after excluding the existing wetland and river ecosystem within the notified forest land and by Forest Survey of India was 23.54 per cent.

The present study highlights the important role played by remote sensing in forest cover mapping and for an overall understanding of the performance of different species under different environmental

conditions. This would be very important in order to optimise limited available inputs for accelerating growth and higher productivity of the species, as well as for better utilisation of the available land resources. no doubt, the technology is the quickest, the cheapest (Rs. 116/- per hectare as compared to conventional surveys of Rs. 200/- per hectare) and the best means to obtain reliable, up-to-date information on forest resources.

The present study also highlights the capability of remote sensing techniques for mapping broad forest cover density classes showing their type, extent and location. However, it is rather difficult to demarcate species unless they occur in gregarious form such as sal species. The floristic variation in which dominance of a single species is not prominent, can be classified under "mixed forests".

COMPUTERISATION OF INVENTORY

A. K. SANYAL*

National Zoological Collection (NZC)

The NZC of the Zoological Survey of India contains both the 'Type' and General Identified materials from India as well as neighbouring countries Bangladesh, Bhutan, Maldives, Myanmar, Nepal, Pakistan and Sri Lanka. The 'Type' specimens are registered in the 'Register for Type Collections' and the 'Non-Type' ones are registered in a separate Register named as 'Register for Identified Collections'. Besides these registered specimens, there are a huge number of identified but unregistered collection with are in a continuous process of registration. In this way every year 10-20 thousand specimens are included in the list of registered collection in NZC.

A total number of about 17,000 'Type' and nearly 3 lakhs 'Non-Type' specimens are contained in the Registers and these are readily available to any researcher on animal resources of India. But the problem lies with handling of huge number of entries, particularly the quick searching of required information pose great difficulties. The computerisation work has therefore been taken up, which is done with IMPACT 8930 Mainframe and UNIFY Data Base Management System.

Computerisation of NZC

In the first phase computerisation of all data pertaining to 'Type' and 'identified' Collection in ZSI has been taken up. To make these data most usable and to analyse them easily for better understanding of species diversity, two basic databases viz., 'Type Data Base' and 'Identified Species Database' have been developed.

The data are fed in the computer system through the following 'Data Entry Format':

Data Entry Format

Institution Code	:	Section Code	:	Type Code
Registration No.	:	Type Name	:	
Species Name	:	Preservation Name		
Phylum Name	:	Condition Name	:	
Order Name	:	Host-Type Name	:	
Class Name	:			
Family Name	:	Habitat	:	
Valid Name	:	Host/Parasite/Hyper-Parasite		
		Site of Infection	:	

Geographical Information

Continent/Country	Count Information
State	Lot/Several/Exact
District/Maj. Isl.	Male -
Specific Locality	Female -

* Zoological Survey of India, Calcutta.

Ocean/Sea/Gulfetc. Other -
Altitude/Depth Total -
Longitude/Latitude *Collection data*
Collection date -
Entry Date-
Collector's Name Determined By: Collection Source:

All 'Types' in the NZC include 1162 Types as follows: from Bangladesh (14), Bhutan (72), Maldives (10), Myanmar (767), Nepal (114), Pakistan (108) and Sri Lanka (77). More than 6000 entries pertaining to 'Identified Collection' have already been computerised and are stored in the discs.

Further to retrieve these entries two query software have been developed:

a) Type Data Query Software:

Type Code	:	Category:		Book No.			
Specimen Name	:	Valid Name	:	Host/Habitat	:		
Region	:	Phylum :		Order	:	Class:	Family:
Registration No.	:	Sex	:	No.of Exs.	:		
Preservation Code:		Preservation Type:					
Condition Code	:	Condition Type	:				
Shelf/Location	:	Entry Date	:	Collection Date	:		
Collector's Name	:	Collection Place	:				
Reference	:	Year : Volume	:	Page	:		
Jour Name	:						

b) Identified Collection Data Query Software

Institute :		Section :	:	Registration Number:		
Collection data	:	Date of Entry	:	Continent/Country	:	
State	:	District/Island	:	Locality	:	
Preservation Name:		Preservation Code:				
Condition Name	:	Condition Code	:			
Type Name	:	Type Code	:			
Host-Parasite	:	Host Parasite Code	:			
Collector	:	Name of specimen	:	Determined By	:	
Phylum	:	Class :		Order	:	Family

Environmental Information System (ENVIS) Centre on Animal Ecology

The Zoological Survey of India is one of the 18 ENVIS Centres in India. The ENVIS Centre in ZSI was established in 1992 with the immediate objectives (i) to provide national environmental information on animal ecology relevant to present needs and capable of development to meet the future needs to users, organisations, processors, and disseminators of information, (ii) to build up storage, retrieval and dissemination capabilities with the ultimate objective of disseminating information on animal ecology speedily to the users, (iii) to promote exchange of information amongst developing countries, (iv) establishment of data bank on animal ecology, (v) responding to user queries, etc.

The Centre is presently engaged in computerisation of all available information on habitat, associate species, status, distribution, population, disease caused, general behaviour, conservation strategies adopted, management control, environmental stress, breeding behaviour and breeding techniques related to the endangered animals of India. The following database know as `Species Database' has been developed to maintain the system:

Species Data Base

1. Habitat
- Type (Terrestrial, Aquatic, etc.)
- Altitude Limit
- Vegetation
- Non-Biotic Factor

2. Associate Species

3. Population (No. of Exs.)
- Wild
- Captive
- Others

4. Status
- Legal (Schedule)
- Taxonomy (Phylum, Class, etc.)

5. *Distribution*
- Geographical (Countries, etc.)
- Ecological

6. *Disease*

7. *General Behaviour*

8. *Conservation*
- Species
- Habitat

9. *Management Control*
- Habitat
- Captive Breeding
- Pest Management
- Food & Feeding
- Morphology

10. *Environmental Stress*
- General
- Human Interference
- Pollution

11. *Breeding & Breeding Techniques*

Similarly the database for published references related to each item mentioned in the species Database has also been prepared in the name of `Paper Reference Database'.

Paper Reference Database

Name of species :
Paper Sl. No. :
Journal Name :
Title : Author : Vol. No.: Page No. : Publ. yr. :

UNESCO Card Catalogue

S. K. CHANDA*

The general information programme was established in the year 1976 to provide a focus for UNESCO's activities in the fields of specialised information system, documentation, libraries and archives. An inter-governmental council of 30 members elected by the UNESCO's general conference is responsible for guiding the conception and planning of the programme. UNESCO's inter-governmental informatics (IIP) programme was set up to reinforce the promotion of internal co-operation and to increase resources for collaborative efforts in informatics. Since 1985, IIP has launched numerous projects in informatics providing multilateral and bilateral support for national and regional efforts.

An International Conference of UNESCO was held in India at New Delhi during the year 1962-63. Most of the representatives of the SE Asian countries were present. It was decided in the Conference that a centralised Card Index Centre should be set up in any one of the Southeast Asian Countries. The centre will be the custodian for the cataloguing of the references for all the Type materials (from Protozoa to Mammalia) described from SE Asian countries. Accordingly, a decision was taken that the Zoological Survey of India is the competent organisation for this work. It was accepted by the then Director, Zoological Survey of India.

The work started during 1963 by a number of scientific persons, collecting references from various Indian and International journals, abstracts etc., consulting original literature as well as by personal contact with the authors. Efforts were also made to collect the original literature from the respective authors to make correct entry as far as possible. The format of the Reference card where the entries are made is presented here:

Genus :				Phylum :				
Species :				Class :				
				Order :				
Reference:				Family :				
Sl. No.	Regd. No.	Repository	No. of exs.	Loc	Date/ Host	Collector	Type Cat.	Rem.

Following symbols are used for the Type repositories:

1. IARI: Indian Agricultural Research Institute, New Delhi (India).

* Zoological Survey of India, Calcutta.

2. IVRI: Indian Veterinary Research Institute, Izatnagar (India).

3. BNHS: Bombay Natural History Society, Bombay (India).

4. CMFRI: Central Marine Fisheries Research Institute, Cochin, (India).

5. FRI: Forest Research Institute, Dehradun (India).

6. MGM: Madras Government Museum, Madras (India).

7. MII: Malaria Institute of India, New Delhi (India).

8. NMC: National Museum, Colombo (Sri Lanka).

9. ORLC: Oceanography Research Laboratory, Kerala University.

10. Z.S.I.: Zoological Survey of India, Calcutta (India).

11. Z.S.P.: zoological Survey of Pakistan, Karachi (Pakistan).

General Abbreviations

Coll. : Collector
Colln. : Collection

Ex; Exs. : Example, examples.
Ref. : Reference.
Regd. No. : Registration
Sp. spp. : Species (singular and plural).
Subsp., subspp. : Subspecies (singular and plural).

In due course this work will be very useful to the scientists working in the South Asian countries, on various faunal aspects (from Protozoa to Mammalia). It will also provide some knowledge on the biodiversity of the faunal components of South Asian countries. Moreover, it will be easy to know at a glance the number of species described from a particular country.

PART VI

METHODS FOR ESTIMATIONS

METHODS FOR ESTIMATING SPECIES RICHNESS AND SPECIES DIVERSITY

J. R. B. ALFRED*

The simplest method for target areas for conservation action is to identify countries with the highest number of species (greatest species richness), e.g. the concept of **Megadiversity Countries** based on species of vertebrates, swallowtail butterflies and higher plants. The 12 countries are Mexico, Colombia, Ecuador Peru, Brazil, Zaire, Madagascar, China, India, Malaysia, Indonesia and Australia.

These hold upto 70% of the world's species diversity. It involves species inventory within a geopolitical boundary and conservation at the country level. However, if fails in uniqueness of fauna and flora and overlap in species composition between different regions situated close to one another geographically, e.g. 271 species of mammals from Ecuador and 344 from adjacent Peru have 208 common.

No consideration for threatened or otherwise of special concern. An alternate approach is to identify areas with the greatest numbers of endemic or restricted range species based on which Hot Spots are categorised. The next method is the Critical Fauna Analysis where entire sets of taxa within the group are considered called complement. The single most important site is that at which the greatest proportion of complement is represented.

Those not included are called residual complement, e.g. five countries with highest numbers of endemic swallowtail species enacted conservation plans, and then 54% of the world's total number of swallowtail species would be conserved. If the next 5 countries were included the total protected species would rise to 68%. Similarly additions of 15, 20, 25, 30, 35, 40 and 45 countries would protect respectively 77, 90, 93, 95, 96, 97 and 99% of the world's swallowtails.

Species/Area relationships: The number of species in an area increases with the size of the area and this increase follows a predictable pattern known as the Arrhenius Relationship.

$$\text{Log } S = C + Z \text{ Log } A$$

Where S = No. of Species, A = Area and C & Z are constants.

In comparative studies of species no., sample size and area must be carefully controlled to eliminate sampling biases.

More species occur in tropical communities than in temperate and arctic communities, e.g. a small region at 60° latitude might have 10 species of ants, at 40° there may be 50-100 species and within 20° of the equator 100-200 species. Within latitudinal limits (belts) the no. of species varies widely among habitats according to productivity and degree of structural heterogeneity. No. of species increases with productivity of the habitat. The marsh with high productivity has a uniform habitat with very little ecological specialisation, while deserts with low productivity have a complex vegetation structure. No. of species in a habitat is strongly related to structure of the vegetation in the habitat, say, height of trees.

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* Zoological Survey of India, Calcutta.

How species are added to a community: Habitat selection increases in areas with many species. Surveys have shown that with decreasing nos. of species, each species is found in a greater no. of habitats and in greater abundance.

***Increased species diversity in the Tropical compared to Temperate and Arctic Regions*:**

Non equilibrium Hypothesis: The tropics are older and more stable.

Equilibrium Hypothesis

1. Speciation rates higher in tropics

   A: Tropical populations are more sedentary, facilitating geographical isolation.

   B: Evolution proceeds faster due to
   - i) Larger no. of generations/year
   - ii) Greater productivity.

2. Extinction rates lower in tropics

   A: Competition less stringent due to
   - i) Presence of more resources
   - ii) Increased spatial heterogeneity
   - iii) Increased control over competing populations exercised by predators.

Diversity at different scales: The differences between habitats are referred to as beta (ß) diversity, while diversity within a site or habitat is alpha (α) diversity. Thus, an area with a wide range of dissimilar habitats will have high-diversity, even if each of its constituents habitats may have low-diversity. Differences in the site diversity over large areas, such as continents, are sometimes, referred to as *gamma (γ) diversity*.

Margalef's Index

The simplest measure of species diversity is the number of species or the species richness, which was calculated after Margalef (1968) as modified by Brower & Zar (1977):

$D_a = (s-1)/\log N$

where, D_a = margalef's index
s = number of species and
N = total number of individuals.

Shannon-Wiener Diversity Index

This measure of species diversity, based on information theory or related to the concept of "uncertainty", was calculated after Shannon & Wiener (1949):

$H' = \Sigma s\ P_i \log P_i / i = 1$

where, H' = measure of Shannon-Wiener diversity
s = number of species and
P_i = proportion of the total number of individual occurring in species i.

However, in the present context, H' was calculated, using its algebraic manipulation as the equivalent equation after Brower & Zar (1977):

$$H' = (N \log N - \Sigma n_i \log n_i)/N$$

Where, n^i = total number of individuals occurring in species i or abundance of species i, and
N = total number of individuals of all the species.

H_{max}'

The maximum possible diversity of H' or H_{max}' was calculated using the following formula:

$$H_{max} = \log s$$

Where, s = number of species or category.

Evenness

The evenness (Pielou, 1966) of the individual's distribution among the species, designated by the quantity J' (also sometimes referred to as homogeneity or relative diversity), was calculated using the following formula:

$$J' = H'/H_{max}'$$

Redundance Value

The redundance value, designated by the quantity R, was calculated using the formula:

$$R = 1 - J'$$

Community Similarity

The similarities/dissimilarities of the collembolan and oribatid communities in the forest and jhum systems were worked out using the following indices:

Sorensen's Coefficient of Similarity (CCs)

The coefficient of community similarity of Sorensen (1948), also known as Quotient of Similarity (Q/S) was calculated after Brower & Zar (1977):

$$CC_s = 2c/s_1+s_2$$

Where, c = number of species common to both communities
s_1 = number of species in community 1 (forest)
s_2 = number of species in community 2 (jhum)

Morisita's Index

This measure of community similarity (Morisita, 1959) is based on Simpson's index of dominance (Simpson, 1949), the probability that two randomly selected individuals from a community will be of the same species.

$$I = \Sigma x_i (x_i-1) / N_i (N_i - 1)$$

Is the Simpson dominance index for community 1, where X_i is the number of individuals in species i in community 1 (forest), and N_i is the total number of individuals in community 1 i.e., forest ($N_i = x_i$); likewise:

$$\lambda = \Sigma y_i(y_i - 1) N_2 (N_2 - 1)$$

where, for community 2, is Simpson's dominance index, y_i is the abundance of species i, and $N_2 = y_i$ the total number of individuals in community 2 (jhum).

The Morisita index of community similarity (also called Morisita's index of overlap; Horn, 1966) is:

$$I_M = 2\Sigma x_i y_i / (\lambda_1 + \lambda_2) N_1 N_2$$

Average Faunal Resemblance

The average faunal resemblance between the forest and jhum communities were calculated using the formula:

$$\text{Average Faunal resemblance} = c(s_1 + s_2) / 2s_1s_2 \times 100$$

Where, c = number of species common to both communities
s_1 = total number of species in community 1 (forest)
s_2 = total number of species is community 2 (jhum)

The structures of the environment can also analysed. The lotic habitat can be measured using the criteria of depth, substrate and current. For convenience 5 to 20 cm. deep corresponded to the shallow margins of the stream/river (riffles); 20 to 50 cm, to shallow pools; 50 to 70 cm., to moderate pools; 75 to 100 cm., to deep pools; 100-150 cm., to deeper pools and finally 150-200 cm., to the deepest areas. The substrates or bottom types can be categorised based on their physical and biotic structures, of substrates increasing in size from silt to large boulders, and one category can be the litter and vegetation along with twigs, leaves and branches. Another category could include structures such as large tree trunks or bed rock slabs, tins, glass and cloth pieces and large sheets of rocks. The current can be estimated by the movement of water about a measuring pole and assigned to categories related to the respective corresponding velocities calibrated with a float and stop watch.

The habitat structures can be analysed by the incorporation of point samples with regularity at each study site and station of the system. 10 to 20 cm. beginning from left bank, points at 5 meters across river and 1 meter across for stream can be taken. Very narrow streams which had only 1 m. as their maximum width, 0.33 m. intervals can be used. Such points should be repeated as sets moving up stream. The initial sampling would require 60 to 100 points for an adequate measurement of the habitat. The data for habitat should be also analysed for the usual diversity indices.

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TABLE - 1

Dimension		Current number and description						
Depth	Range	1	2	3	4	5	6	7
		0-5	5-20	20-40	40-60	60-80	80-100	100
	Description	Very Shallow	Shallow	Moderately Shallow	Moderate	Moderately deep	Deep	Very deep
Current	Flow velocity (m/s)	0-=0.05	0.05-0.2	0.2-0.4	0.4-0.7	0.7-1.0	1.0	
	Description	Very slow	slow	Moderate	Fast	Very fast	Torrent	
*Substrate	Diameter (mm)	0.05	0.05-2	2-10	10-20	20-40	40	
	Description	Silt	Sand	Gravel	Pebble	Stone	Boulder	

* Additional substrate categories include (7) Litter/vegetation (biotic) and (8) Miscellaneous (Abiotic) Tins, Cloth pieces, Glass pieces, Logs, Wood pieces and large sheets of rock.

CENSUS OF WILD ANIMALS

P. SANYAL*

In order to start with any basic ecological study the determination of faunal status is a primary necessity. Irrespective of nature of terrain, nature of vegetation, the assessment of population dynamics becomes an essential component of such basic study. This involves carrying out census of animals either in terms of absolute numbers or simply finding out the trends of animal disposition. Census helps in finding out the

- State of health of the wilderness,
- Determining the geographical distribution/abundance in poorly known areas
- Determining the distribution/abundance by habitat and basic population monitoring,
- Determination of population size,
- Determination of animal density response to management inputs, and
- Determination of the state of Biodiversity.

Materials and Methods

The common methods of animal census are either through (i) direct count or from (ii) study of animal marks like `pug marks', `spoor marks', dung counts, call counts, kill counts etc.

Best time of census: It depends on the circumstances in each case; after rains is usually the time for the ungulates. For carnivores, end of winter is better. For tigers the month February is good for high rainfall areas and April for rest of India.

Total and sample counts: Whilst it may be possible for large ungulates, e.g. Tiger/Leopad to get a *total count;* for others *sample estimates* are in vogue. In general, larger area and smaller animals lead to sample counts.

How to calculate the sample size: While finding out animal populations on the basis of sample estimates it is mandatory to calculate the appropriate sample size for the predetermined (depending on the availability of time, economic and human resources) sampling intensities (or). In order to do this, a quick preliminary survey may be carried out over small representative area, then find out the standard deviation (S). Desired confidence limit is set i.e. estimate mean value " confidence limit (L). for 95% confidence limit this range is set by the standard error times a factor Z = 1.96 or say 2, for 99% confidence limit `Z' is taken as 3 and the factor `r' indicates ratio of size of sampling unit to the ratio of sampling area. This gives an equation for the desired number of sampling units(n).

$$n = z^2 \times S^2 / (r \times L)^2$$

* Department of Environment, Govt. of West Bengal, Calcutta.

Equipment: Census equipment vary with the methods and objectives of the census. In case of

A. Ungulates

- Water hole count method is used more in arid areas. This needs a machine or watch tower, a binocular, Hand-compass to ascertain the directions of entry and exit and a Note book and Dot pen to be used by the watcher.

- 'Block counts' are usually done as a *total count*. Binoculars are provided to experienced staff to reach an informed guess. Increasingly however search is made on a particular day with hand held compass and the process is repeated. In case of larger blocks (5 to 10km^2) more than a day is taken for survey. As usual Note book, Dot pen, Block map, 2m tape is provided to counting staff.

- 'Transact count' is carried out due to paucity of funds or time. This can be done from vehicle, using binoculars or on foot by using the 'Range Finder' taking recourse to different estimators (Burnham, 1980).

B. Carnivores

For large carnivores 'pug mark' census is universally applied. This needs Tiger Tracer, Plaster of paris, a small wooden or galvanised sheet frame for taking plaster casts, a water bottle, a mixing mug for plaster cast, tracing paper, compartment map, Note book, dot pen, coloured free-flowing pen set for tracing, small packet of 'French chalk' to make the pug-mark notches prominent, a 2m tape and a hand held-compass.

More recently 'camera trap' method has been applied which needs a special type of camera (Karanth, 1994). Trail Master brand camera is used in India (Manufactured by Goodson Associates, lenexa KS 66215, U.S.A.)

Smaller animals are usually sampled over smaller area.

C. Rare/Target animals

For specific target animals like primates, Crocodiles, Turtles, Birds, rare species, normally a vehicle or vessel (in water is used. In these cases generally the study of population trend is more important than actual population count.

Results and Discussion

Census of Ungulates

A. Population Trends

In case of rare species or for routine monitoring population trend is the most useful tool for the wildlife managers. This can be carried out on a (i) Block-wise basis, (ii) from Road-side sighting index monitoring, (iii) from Dung surveys, and (iv) from water hole counts.

! Block trend probability of a survey block of an animal is p = n/s where in = No. of blocks with animals, s = No. of sampled block

variance of p or V(p) = p(1-p)/(s-1)

for example, if 100 blocks have been surveyed and 60 have presence of rare species, then p = 60/100 = 0.6 : v(p) = 0.6 x 0.4)/99 = 0.0024.

This means real probability lies between 0.6 " 0.0024. After subsequent census it can be examined if the value of 'p' differs significantly from the earlier value in order to predict animal trend.

! Road side index monitoring can be done on the basis of frequency of the sightings of the animals and thus the population trend can be calculated.

! In case of Dung Survey method, done along permanent transacts the additions of dung over older ones gives a population trend. As the identification of old and new ones pose problem it is better to check the coefficient of Variation (CV). If CV is <10% the population trend can be ascertained.

CV = (s/X) x 100

Where s = standard deviation and X - mean. Commonly used transact dimension is 25 m x 2m.

! Water hole method: The driest period is the best. Observation from 'Machan' starts in the evening and continues for 24 hours. Count time is one hour after arrival at Machan.. Three repeated counts can give rise to CV value to check census reliability. While carrying out this method care should be taken for two assumptions:

- There exists a linear relation between No. of animals seen drinking per time and actual No. present in the area.
- Field staff are able to estimate an unbiased record of animals visiting the water hole.

B. Actual Population

For the estimation of the actual population following analysis is needed:

! In case of Dung Samples for Cheetal, Hog Deer, Barking Deer, basic information required is (i) an estimate of dung density per time period (ii) information on estimated defecation rate.

Grid of sampling plots are selected on map and sampled with two random start.

Cheetal density = (Observed Dung density for specific time) / (Estimated defecation rate for same time period for single animal).

The standard error of difference of Means (SEd) will suggest probability of accuracy.

$SEd = (\sigma 1^2/n_1) + (\sigma 2^2/n_2)$
where, $\sigma 1$ = Standard deviation of the data of 1st random start
n_1 = No. of animals of the data of 1st random start
$\sigma 2$ = Standard Deviation of the data of 2nd random start
n_1 = No. of animals of the data of 1st random start.

If, the Difference of means of the data of those two random starts<(2xSEd). Confidence limit of accuracy will be > 95%.

! Block counting: Total counting is done in case of smaller areas. For sample counts, normally 10% sampling is done with Line abreast drive count method. One essential feature is that there should be clearly defined straight line along the axis of counting block, will proceed along lines not greater than 3 km drive. Prior stratification should be used where it is needed. Analysis of variance also should be done in similar manner as described in the last paragraph.

! In case of Line Drive Census, each person or a group of two people in difficult terrain should be separated from the next group by the largest distance possible without loosing sight or sound contact. this may be 20 m in closed, 50 m in deciduous and 100 m in open woodlands. Counting group marches along a line. One sighting animals or group of animals following are recorded: (i) species, (ii) number, (iii) compartment No., (iv) time of observation, (v) direction of movement. Data analysis may be done as follows:

A data comes from 23 separate block or line sambar

Block/Line	: 1	2	3	23	Mean	St. Dev.
Density	: 11.2	6.9	4.8	6.4	10.4	4.03

Std. Error SE - 4.03/(23) = 0.84

Confidence Limits (at 5% Level) SE x 't' value = SE x 2.07 = " 1.74

The mean density and confidence limits for the Protected Area is thus 10.4 " 1.74 per km^2.

In southern Bengal the Elephant census was done accordingly during 1993. Three main group were identified over 4590.9 km^2 as below:

Arabari	:	22 Nos.
Mirga	:	19 Nos.
Dhadka	:	14 Nos.

The sex ratio is 1:1.4 and the calf ratio is 1:6. Maljuria (Male pair or three) groups were located as below:

Purpulia: 3 elephants, W. Midnapore: 2 elephants, E.Midnapore: 2 elephants, Ramrama (Also a part of W. Midnapore) : 2 elephants.

3 Nos. of solitary elephants were located in Jamboni, Bagdoba and Basudevpur (Bankura). Tusker: Makna ratio of unattached male were found to be 2:1. Thus total 67 elephants were encountered during 1993 census (Project Elephant, West Bengal, 1995).

- **Transacts**: Transact based animal counting can be either vehicle based or can be done on foot. Some times permanent transacts are also laid out. Although under this method measurement of length is easy but width poses problem.

Density (D) = Number of animals seen) Transact area.

A Range finder is used to get the perpendicular distance of a centre of animal group.

If the angle (a) is noted and the distance is either taped or guessed as `d', the perpendicular distance = d x Sin(a). Mean perpendicular sighting distance (r) gives the estimate of Strip width or Lateral visibility.

Density D = n / (L x 2 x r) where, L = length of transact and n = number of clusters detected.

Census of Carnivores

For the Tiger and Leopard census the best time of `pug-mark' census is February in North India and April for the rest of areas.

There are 4 distinct phases of work (Panwar, 1979)

- Organisation
- Pre-census training of personnel for 7 days
- Census operation in 2 regimes viz.

Regime I: -5 days census. The first two days are reconnoitry and next three days are intensive census with `Tiger tracer' and Plaster Cast collecting equipment.

Regime II: - 2 days repeat census.

- Data collection, analysis, finalisation.

The pug mark identification is now a days carried out by measuring 18 parameters of pugs and feeding them to "Cluster Analysis Programme" in a Systat software (Das and Sanyal, 1993). Following measurements may be taken as a set of independent stable parameters for identification of pug marks.

1. Pug length (1)
2. Pug width (1)
3. Distance of each toe centre from the pad centre (4)
4. Distance of successive toe centres (3)

5. Area of pad (1)

6. Length of major and minor axes of pad (2)

7. Ratio of major axis of each toe to minor axis (4)

8. Ratio of area of pad to the area of rectangle containing the pug (1)

9. Ratio of the area of pad to the pentagon of the centres of pad and toes (1).

More recently for smaller tiger reserves a cross checking by Camera -trap method and also by "Prey-base Model", are applied (Karanth, 1994).

Camera-trap method is however, not applicable for most of the reserves. The high value cameras can be stolen away.

The estimation of prey being based on sample survey only, can lead to more erroneous population figures for large carnivores through model prediction. Nevertheless a check from all three angles viz. 'pug-mark' counts. Camera - trap and 'prey-base' census, can indeed help reaching the correct census of top predators, the indicators of the health of wilderness.

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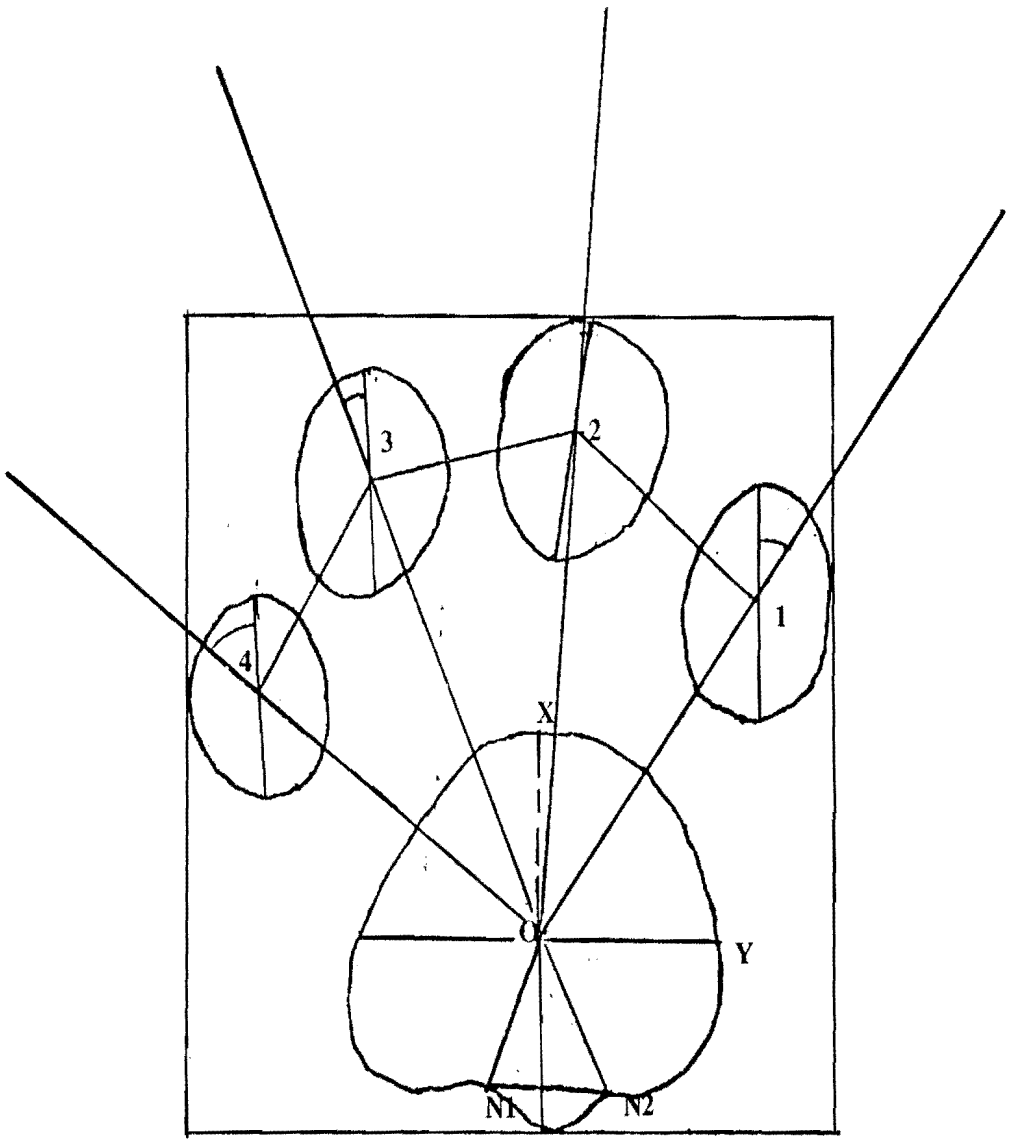

Fig.21 - Measurements of a Pug Mark of Tiger

METHOD OF ESTIMATING INSECT COMPONENTS FROM HABITAT AND NEST

P. K. MAITI*

Introduction

Both larvae and adults of many groups of insects in general, and social and sub-social insects in particular, construct extensive nests, tunnels, runways, galleries, cocoons, pupal cell, etc. in their habitats. Such nests may be prepared in the soil and wood; hanging from trees; in the cracks and crevices in wood, soil etc.

For example, sub-social insects, such as, Scarabaeid and Scolytid beetles excavate their nest in soil and wood respectively. In social insects, such nest building is a very common feature. Termites build extensive mound, and carton nests in and above the ground, apart from excavating extensive tunnels in wood in its many forms. Further, the nasute termites prepare arboreal hanging nest as does some ants, honey bees, wasps, hornets, etc.

Sampling Techniques and Estimation

(a) *Sampling*: The main object of taking sample of any community/ population from a habitat is to get relevant representatives of the large community or population.

(b) *Sampling unit*, its selection and size:

i) Sample unit should be of such nature that the unit in the entire sphere of sampling reflects and equal chance of selection.

ii) The sampling unit may not be changed in the growth or any change of general habitat of the area.

iii) The proportion of population using sampling unit as a habit must remain constant.

iv) Sample unit must be reasonably small so that enough units can be examined on a given plot and a given data to provide an adequate estimate of variance.

v) The sampling unit must be easily delineated in the field.

vi) The sampling unit may be such a size as to provide a reasonable balance between variance and cost.

* Zoological Survey of India, Calcutta.

(c) *Number of samples:* Within an ecologically homogeneous habitat, the number of sample required (N) be estimated approximately as

$N = (IS/DX)^2$
where S = Standard error
D = Required level of accuracy expressed as a decimal (normally 0.1)
T = a quantity depending on the number of samples.

Placement of traps: For effective sampling of insects different types of traps are to be used to cover all possible habitats above the soil in a quadrate within the study area.

i) *Light Trap*: It should be placed in the centre of plot and at a height of 4-5 feet from the ground.

ii) *Pitfall Trap*: Usually 3-5 traps to be kept in different corners of the plots.

iii) *Scented Traps*: Usually 2-3 traps may be hanged at the height of one metre from the ground level in each plot.

iv) *Net Sweeps:* The nets are used in systematic sweeping of the ground level vegetation.

v) Lastly, the all out search be made where manpower is sufficient in number.

Sampling Soil and Litter - Inhabiting Insects

The sampling of large, relatively static aggregations of insects in the nest built mostly by the social insects presents quite different problems which will be discussed here with particular reference to termite mounds. Such mounds are quite frequently found within our limits in the tropics. There are two aspects to this problem, estimating the density of mounds per unit area and estimating the number of termites per mound.

Estimating the Density of Mounds

Quadrant or Direct Counting

In grassland or sparsely wooded country it is possible to mark out the quadrants and counts the mounds in each quadrant. Marking out large quadrants is a tedious job and is virtually impossible in area with a high density of trees and shrubs. More often, mounds may be counted in the whole of a particular area. The most suitable size of quadrant is lobe. Selected by the density of mounds the higher the density the smaller the size of quadrant that will give a reliable estimate.

Measurement of Distance Between `nearest neighbours'

If the mounds are distributed at random their density may be estimated from the following expression: $m = 1/r^2$ where m = density per unit area, r = mean distance between nearest neighbours. However, in few cases where this method has been adopted for studying out nests & termite mounds, the distribution of these nests is found to be non-random.

Estimating the Population in the Mounds

The methods used fall into two groups; those where the whole mound is sampled and the total population, or an estimated proportion, is counted; and those where a sample taken from the mound and the termites counted in the sample. In the first method total population may be counted from the material extracted from the mound and floated on water for convenience.

On the other hand, core sample (volume 200 cm^3) by swinging a weighted club with a sharpened tubular side arm into the base of the mound is taken. By this method samples were obtained rapidly before the termites had time to respond to the disturbance.

Sampling from Wood Nests (bark-beetles)

The species of beetles (*Scolytus*) feed in the sapwood between bark and wood of living trees. To note the size and composition of these beetles in association with other bark fauna, the sampling can be done on dead wood, by cutting the bark in size with the help of a penknife, axe or chisel, chalk and measuring tape.

Five separate areas, 20x20 cm quadrants, may be cut on felled logs and their bark is removed to reveal the galleries either on the surface of the wood or inner surface of the bark. For each insect species, the counting and recording the individual available after sampling may be done. On the other hand, counting of individual mother galleries as well as of the larval galleries from each quadrant being engraved on the surface of wood or underside the bark, may be done to calculate the possible population of each species in different quadrants. This sampling technique will be repeated in different logs of trees to collect maximum number of individuals per species. A simple calculation will provide the diversity index of the bark beetle communities in some specific host in a habitat.

Sampling Insect from Soil Nest (Mound)

Sampling from insect nest is difficult, yet some devices have been proposed which can be mentioned below :

i) If the nest, being sizeable and represented by small population may be totally explored to have the total count of individuals representing the nest. This may be done in case of scarabaeid beetles and in some social wasps.

ii) In bigger nests, the sampling is done with the help of a shovel penetrating at different surface of the nest to get the representative population, as in the case of termite mound, ant nests, etc.

iii) At times, net sweep is done on surface of the nest to get the population estimates.

Indirect Clue for Recognition of Insect Community

Interestingly enough, at the time of survey if the insects are not available for sampling, some indirect clue may be found in the habitat. The impression of tunnels, galleries, casts, exuvae, pupal chambers, entrance and exit holes, etc. of different insects serve as materials for biodiversity study. The characteristics faecal pellets, grass granules of wood-boring insects also are useful for the purpose.

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METHODS OF ESTIMATING ECOLOGICALLY RICH AREAS, ENDEMIC AREAS, HOTSPOTS AND PROTECTED AREAS

J. R. B. ALFRED**

Develop and distribute a biological classification system for use in global analysis of protected area coverage.

Also include:

a) Areas with rare species and communities
b) Undistributed areas requiring protection
c) Areas to ensure an adequate geographic distribution across all significant Biogeographic subdivisions.

Classification may be at four scales:

a) ***The BiogeographicZones:*** Which are the major species groupings. These zones indicate a distinctive set of physical and historical conditions. The Himalayas and Gangetic Plains are examples of two adjacent but obviously extremely different zones.

b) ***The Biotic Provinces:*** a further level of detail within these zones. Provinces contain some distinctive species elements as for example the differences between the Northwest and west Himalayas. This division is taken to be the Sutlej river.

c) ***The Biogeographic Regions:*** these subdivisions are typically distinct geographic areas (landforms etc.) within a large province. They may or may not contain distinctive species elements. An example of regional subdivision is the Kinnaur, Garwhal and Kumaon distinction in the Western Himalayas province.

d) ***The Biomes:*** the major ecosystem groupings found within each province and region. Examples would be alpine, subalpine, temperate conifer forests etc., within the Western Himalayas.

Assigning Conservation Values

Conservation values can be assigned to an element such as a taxon or a locality with reference to an attribute e.g. the extent of geographical distribution of a given taxon or the number of taxa in a given locality (Daniel's et al. 1991).

The different states of such attributes are then assigned values based on a criterion.

The more restricted the geographical distribution of a taxon, the greater the value of the taxon, or the larger the number of taxa present in a locality the more valuable the locality. The actual values may be

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either be ranks along a scale or a specific number. Conservation values can be one of the inputs for identifying a set of protected localities/hotspots/endemic areas. The attributes of a taxa can be the extent of geographical distribution, their habitat preference, their taxonomic position and their degree of endangerment. The criteria translating these attributes into values are based on the assumption that the rarer or more restricted a taxon the more valuable it is, and the more taxonomically unique or endangered a taxon the more valuable it is. We may have four attributes relating to the geographical range, and one each for habitat preference, taxonomic distinctions and degree of endangerment.

Geographical Range

G1 - Over the entire world (division into 6 zoogeographical regions).

G2 - Over the oriental region (division into 9 sub-regions).

G3 - Over the Indian subregion (division into 8 provinces)

G4 - Over the particular province (division into 4 sections)

Conservation value for a taxon by geographical range can be:

$$G = (N - a)/(N - 1)$$

Where N is the No. of subdivisions at a given geographical level, and a is the no. of subdivision from which the taxon is known.

The four levels of geographical distribution are to be treated as separate dimensions in view of the distinctive patterns of distribution along each level. For instance a taxon restricted to a small area may be wide spread within the province. Similarly a taxon found only in a province in India may be wide spread in South Asia.

Conservation value of each taxon by habitat preference can be computed as

$$H = (N-a)/(N-1)$$

Where a is the number of habitats in which the taxon is known to occur and N is a fixed number (The total no of habitats in that area).

Taxa with more restricted habitat preferences are assigned higher values. Conservation value of taxon reflecting its taxonomic distinctness can be calculated as -

$$T = 1/(axb)$$

Where a is the number of species known in the genus to which the taxon belongs and b is the number of races under the species to which it belongs. Races or subspecies are the lowest distinct taxonomic units below the level of species and hence indicate totally of the genes contained in any species. The best will be to measure the taxonomic distinctness on the basis of detailed information contained in the phylogenetic tree.

Conservation Value by Degree of Endangerment for Each Taxon can be -

$E = p$,

where p = is the proportion of endangered taxa in the family to which the taxon belongs.

For example Four types can be distinguished -

1) Taxa endemic to islands
2) Taxa narrowly confined to scarce habitats
3) Taxa particularly sought after by human consumers
0 Taxa that depend on easily disturbed foodchains.

The next step is the occurrence of taxa in given localities, habitats or zones. The criterion for assigning conservation values to such geographical elements could be species richness (no. of that taxa) or a measure of diversity (e.g. Shannon-Wiener index), the total conservation value summed for all taxa, occurring in a given geographical element or the near conservation value of a taxa in each geographical element.

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METHODS OF PREPARING QUESTIONNAIRE FOR COLLECTION OF DATA ON ECOLOGY

J. R. B. ALFRED*

This will be based on a landscape ecological approach. The landscape concept in ecology combines modern ecological principles with a geographical foundation. A landscape may be understood as a heterogeneous land and water area composed of a cluster of interacting ecosystems, which are repeated throughout. For instance, at one point in a typical low elevation landscape, we may come across a stretch of rice fields, a patch of evergreen forest, a grassy bank, an arecanut plantation, a pond, a road etc. At another point in the same landscape could be similar elements with some variations like the occurrence of two patches of evergreen forest as well as some new elements like a rubber plantation and a stream. All such easily distinguished components or units of the landscape are known as **landscape elements** (LSE). For convenience, focus attention on individual elements covering an area of $250m^2$ or more.

The landscape ecology urges us to view the various elements of the landscape (LSE) as closely in energy and material relationships and not as separate ecosystems. Such a dynamic landscape composed of a mosaic of LSE types, should form the focus of the study. A rich biodiversity is distributed among the various elements of this landscape. Man has been living as a part of the landscape and playing a key role in its transformation which could be a gradual process or a rapid one bringing out dramatic changes, often of destructive nature.

A compact landscape with a number of LSE types, and having an approximate total area of $25m^2$ may be chosen for the study. This task is easy if boundaries are precisely delineated of the chosen landscape on the copy of a topomap of the scale 1:50,000. Here 1 cm. is equal to 500 m or 0.5 km. A square of 10x10 cm. covers $25km^2$. There is no necessity for the chosen landscape to be of square shape. It can be of any other convenient shape but approximately covering 25 km^2 area.

The selected portion of the map depicting the area under study may be enlarged about 10 times. Such an enlarged map, preferably cloth bound, may be carried to the field for mapping the various LSE. LSE of less than $2500m^2$ in area need not be included in the landscape mapping and need not be sampled also. However special elements of conservation value, of even smaller extent like a small pond sheltering a rare species of fish or tulle o sacred groves which are relic patches of vegetation on a landscape may be given special consideration and treated as distinct landscape elements. An example of LSE types by the classification given below:

1. Aquatic-freshwater LSE types

Flowing

Perennial
Stream/river along steep slope (30°)
Stream/river along medium slope (15° - 30°)
Stream/river on low slope to plain (15°)
Sewage water canal

* Zoological Survey of India, Calcutta.

Seasonal

Stream/river along steep slope (30°)
Stream/river along medium slope (15°-30°)
Stream/river on low slope to plain (15°)
Irrigation canals.

2. **Terrestrial natural LSE types**

Forest

Evergreen

Size of the LSE>40ha
-do-20-40ha
-do-10-20ha
-do-5-10ha
-do-<1ha

Disturbed evergreen

Size of the LSE>40ha
-do-20-40ha
-do-10-20ha
-do-5-10ha
-do-1-5ha
-do-<1ha

Non-forest

Scrub jungle
Savanna
Grassland
Landscapes

3. **Terrestrial man-made LSE types**

Human habitation
(To include homestead gardens, hedges, walls, compost pits etc.)
Quarries, mines, mine dumps

Roads, cart tracks, foot paths

Cultivation-annual/biennial/seasonal crops

Perennial plantation/orchard crops

Forest tree/other timber plantations.

There would occur many microhabitats, with their characteristic biota within the various LSE. These are like decaying logs, rolled leaf, hollows and holes of trees, cave, leaf litter, etc. These microhabitats will not be considered as distinct LSE. However, if the taxa under investigation occur in any one of these, that microhabitat should be noted in the data sheet.

Stratified random sampling is recommended. Each LSE type forms a stratum for sampling. Under each LSE one or more LSE may be randomly selected for sampling. All of the LSE serial numbers under one type may be written on bits of paper, shuffled, and the required number of bits may be picked up. These would carry the serial numbers of the randomly selected LSE to be sampled.

Transact with Quadrant Method

(a) *Transact layout*: The alignment of the transact should be along a straight line parallel to the longer axis of the element being sampled. In case of winding or curved landscape elements like rivers or roadside the transact need not be along a straight line. If there is any practical difficulty in making a straight line transact such an inaccessibility of the terrain, a zigzag or winding transact may be laid. However, the reason for the deviation may be stated in the transact Information Sheet. Edges of the LSE should be avoided while laying the transact. Please remember to mark the layout of the transact and quadrates with paint or other markings so that field studies could be repeated later along the same transact.

(b) *Length of the transact*: Length of the transact shall be 600 m. In case an LSE cannot accommodate the 600 m transact there are three options before us. 1. Make two or more parallel transact avoiding overlapping. 2. If that be not possible move on to another LSE of the same type. 3. If there is no second LSE of the same type more quadrates may be accommodated by reducing interquadrate distance to a minimum of 20m instead of the recommended 40m. The reason for such deviation may be written on the data sheet.

(c) *Number of quadrats along a transect*: A transect of 600m normally should accommodate 12 quadrats of 10m x 10m with an inter-quadrant distance of 40m. But in special cases like a small but rare LSE the quadrant number could be lower. The inter-quadrant distance also could be lesser than 40m but not lesser than 20m. Parallel transects, at least 20m apart from each other to avoid overlapping, could also accommodate more quadrats. In some instance, as in monoculture areas with poor diversity, a minimum of 6 quadrats may be studied. However transect length, as far as possible, should be 600m.

(d) *Alignment of quadrats along the transect*: Quadrats are to be laid on either side of the transect alternatively. In exceptional cases like a linear landscape element likes roadside or streambank or in case of a steep and unworkable slope or occurrence of an altogether different landscape element on one side the quadrats can be made on the same side of the transect. The reason for such a deviation may be stated in the data sheet.

Selection of Random Quadrats

It has been stated that a transect should have 12 quadrats of 10m x 10m with an inter-quadrant distance of normally 40 meters. In case of a large LSE the variations in biodiversity could be better sampled by laying two transects of 600m within the same LSE and choosing randomly 6 quadrats in each of them.

Number of Transects/Quadrats

There is no hard and fast rule as to how many transects/quadrats need to be studied in a landscape. Our first aim is to sample as many representative landscape elements by transect cum quadrant method as possible. The more diverse (species rich) elements should be studied more intensively than species poor elements. For instance the evergreen forests are more diverse in plant species than a

monocular teak plantation. More transects in such LSE types like evergreen forests are advisable attempts as many transects so as to collect data from minimum of 200 quadrats from the entire study area. Aquatic habitats, however, should be sampled separately by appropriate methods.

Recording of Data

(a) *Transect information sheet*: Before laying the transect within a randomly chosen LSE details regarding the particular LSE should be entered in the Transect information sheet.

(b) *Tree data sheet*: The data on trees within a quadrant is to be entered in the Tree data sheet, separately for each quadrant.

(c) *Other plants data sheet*: Details regarding all the other plants of the quadrant may be entered in this sheet. There is no need to make sub-quadrant may be entered in this sheet. There is no need to make sub-quadrats for enumerating the ground layer plants. The occurrence of all such plant species may simply be noted down. However an optional data sheet for those who are interested in population dynamics of the ground layer plants may also be prepared.

Where transects and quadrats or transects along are used for plant studies the same transects/quadrats also may be surveyed for animals on separate occasions. Special sampling techniques, for study of organisms in aquatic habitats is done separately.

Studies on amphibians and reptiles may be conducted inside the same quadrats as for vegetation (apart form the opportunistic observations within different LSE). The quadrats will have to be thoroughly searched for amphibians and reptiles once in a month, by turning stones, leaf litter, under bark, logs, tree holes etc.

BIODIVERSITY INVENTORY

Transect Information Sheet

1. Name of investigator/investigators:
2. Study area :
3. Locality :
4. Village :
5. Taluk :
6. District :
7. Altitude range of transect : From.....m to ....m
8. LSE Type : Code No. LSE No.
   (Details as shown in the map)
9. Local name of landscape element :
10. Approximate area of LSE in ha :
11. Location : Hilltop/slope/foothill/spur/valley
12. Soil colour :
13. Rock outcrops : Low or nil/moderate/high
14. Distance to the nearest waterbody, if any:
15. Type of nearest water body, if any :
16. Occurrence of shifting cultivation :
17. Details of logging :

18. Lopping of small branches and ground vegetation :
19. Firewood collection :
20. Litter collection :
21. Details of any other materials extracted from the LSE
    By local villagers :
    By others (specify) :
22. Fire incidence in or around the LSE :
23. Grazing :
24. Distance to the nearest human habitation from the LSE :
25. Brief history and description of the LSE under study :
26. Surrounding landscape elements :

Abbreviations for the sampling sheets

1. Habit -Ts : tree sampling; S : shrub; L: Liana;
   Cl : cluster; Us : undershrub; H: herb

2. Phenology - LL: leafless; Fl: flower; Fy: young fruit; FM : mature fruit.

3. Tree height may be estimated to the nearest mater

4. GBH - measure with at 1.3m above the ground level for all trees of 30 cm or more girth; if large buttresses present the girth may be measured just above the buttresses.

5. If the tree forks from below the girth of each branch may be recorded at 1.3m above ground separately.

6. Canopy cover - O = no canopy cover; 1 = adjacent canopies meet; 2 = crowns overlap still allowing light; 3 = sky not visible.

ANIMAL SAMPLING SHEET
(General)

Date	:	Quadrant size	:
Page no.	:	Quadrant interval	:
Transect no.	:		
Quadrant no.	:	Taxa	:
Name of LSE	:	No. of LSE on map	:

S. No.	Species Name	Local name No.	Microhabitat	Remarks
1.				
2.				
3.				
4.				
5.				

*The number of individuals need not be counted in case of small colonial organisms like termites, ants etc.

DATA SHEET FOR SAMPLING AMPHIBIANS

Date :
Transect No. :
Quadrant no. :
Time of observation :
Name of LSE :
No. of LSE on map :
From ..... to.....

S. no.	Name species	L. name	No. Stage	Size class	Microhabitat	Remarks
1.						
2.						
3.						
4.						
5.						

*Tadpole, limb development, adult etc.

Note: If identification is difficult in the field give codes to the species. If possible measure the length & breadth of the animal. Otherwise mention its size (e.g.. small, medium, large etc.)

DATA SHEET FOR BUTTERFLIES

Date :
Transect no. :
Name of LSE :
No. of LSE on the map :

S. No.	Time/steps	Species	Aprox no.	Sighting Distance above Distance ground
1.				
2.				
3.				
4.				
5.				

DATA SHEET FOR SAMPLING REPTILES

Date : Name of the LSE :
Transect no. : No. of LSE on map :
Quadrant no. :
Time of observation : From ..... To .....

S. No.	Species name	Local name	No.	Length	breadth	Microhab	Remarks
1.							
2.							
3.							
4.							
5.							

OPPORTUNISTIC SAMPLING

S. no.	Date	Timings	Species	Sighting	LSE	Location type/no.	Remarks
1.							
2.							
3.							
4.							
5.							

Note: This table is common to all groups of animals. The remarks will include microhabitat or animal activity or any other details. Data can be sorted into taxonomic groups later.

Data entry of all the sampling sheets may be made in the rough note book in the field, using same format but employing suitable nomenclature abbreviations. Final and standardised data sheets may be prepared later.

BIRDS/BUTTERFLY SAMPLING SHEET

	Essential Records				Optional Data			
S. No.	Step no./ Distance time	Name of species	Number of individuals	S = Seen H = Hard O = Overheard	Above ground distance	Activity	Sex/s and age/s	Other
1.								
2.								
3.								
4.								
5.								

1. Covering 600m in 2 hours (8.00 to 10.00 a.m. for birds and suitable time in the afternoon for butterflies) necessitates walking 5m every 1 minute. This is equivalent to 7 average steps or 2 length of one quadrant or 1/8 interquadrate distance. The average distance covered per pace may vary from 0.5m on ascends to 0.9m on descends, and accordingly modifications should be made keeping distance and time constant.

2. Species names may be suitably abbreviated for field recordings.

3. Number of individuals and details of sex, age etc. may be approximate and the latter can be abbreviated.

4. For birds heard; speculation may be made about number of individuals on the basis of observed degree of flocking or nature of calls heard, only if sufficient experience or expertise is available.

5. Sighting distance and distance above ground may be approximated as 5, 10, 20, 50, 50+m.

6. Activity may be recorded as Fr-foaging, Fl-flying, St-sitting, Sg-singing etc.

7. Birds in the semicircle ahead of the investigator are to be recorded without repetitions.

For birds as well as butterflies, 4 samples each of 2 hours need to be taken from only the major LSE types (in their respective larger element/s). The butterflies may be sampled during suitable time in the afternoon such as 10.00 a.m, at 12.00 noon. Thus, if there are 6 LSE types each with at least one fairly largely patch in a locality; advisable number of samples would be (6 types x 4 samples each) 24 samples. This requires 24 days spread over 8-14 dry months. All the 4 samples per type may be taken along same plant transact

METHODS OF PREPARING FAUNAL INVENTORISATION

J. R. B. ALFRED*

Species Diversity:

Measuring biological diversity
Species richness and its country and regional distribution.

Micro-organisms:

Assessment of diversity
Regions and habitats of maximum diversity
Role in biodiversity and biosphere functions
Contribution to sustainable development
Ex situ conservation.

Fauna:

Nematode Diversity
Deep sea invertebrates
Soil microfana
Fishes-freshwater, subterranean, and coral reef
Higher vertebrates

Centres of Species Diversity:

Measuring and determining areas of conservation priority
Areas of endemism
Areas of rich diversity

Threatened Species:

Regionally
Nationally/Geopolitically

Habitats and Ecosystems:

Mapping of Ecosystems
Tropical Forests
Grass Lands Values threat conservation management
Wetlands
Coral Reefs
Mangroves

Uses and values of Bioresources:

Animal use-terrestrial
fisheries
domestic
Current uses
Community uses

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* Zoological Survey of India, Calcutta.

Eco-tourism
Crop Yields
Production of pharmaceuticals

National Legislation:

Protection of Wildlife
Protected areas

International Instruments:

Multilateral Treaties
International obligations: Protected areas
Regional seas programme

Species Inventory:

Current status
No. of described species
Deficiencies of existing database
Prediction from existing partial inventory
Taxonomic Specialists
Location of voucher specimens
Access to Literature

Sampling the hyperdiverse but poorly known
Prospects for improved Species Richness
Intellectual Property Rights
Regulated Trading

PREPARATION OF THE FAUNAL INVENTORY WITH SUGGESTED STEPS FOR RECORDING DATA ON SPECIES

R. K. VARSHNEY*

Introduction

Faunal inventories are already in existence for some countries and regions. However, a thorough attempt for the documentation of species in the countries of South Asia shall be most welcome. The 12-volume 'Fauna of West Bengal', published by the Zoological Survey of India (1992 onwards), can be treated as a model for such inventories animal group-wise.

Although printed cards and registers are suggested here, as in practice in many museums and research institutions, the current trend is to store data on floppies and use this computerisation data for various type of lists. Such modernisation is indeed useful.

Some steps and sample sheets are suggested as follows:

Data Recording

Country -
Region/Ecosystem -
Name of Animal Group explored -
- Phylum -
- Class -
- Order -
- Family -

Frequency of data collection -
Mode of observation -
- Capture -
- Visual sighting -
- Hearsay report -
- Published -

Locality data -
- Latitude -
- Longitude -
- Altitude -
- Province -
- District -
- Topography -
- Major vegetation -

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Status
- Size of sample -
- Relative abundance -
- Very common/Common/Occasional/Rare -

Seasonal data -
(Record fluctuations)

Other observations -

Field Record

1. Frequency of data collection -
2. Sl. No. of entry and name of locality -
3. Data and time of entry -
4. Seasonal data -
5. Observations
   - Animal Group 1 -
   - 2 -
   - 3 -
6. Inter-relation with any other animal/plant -
7. Population status at the site -
8. Species identified in field No. of exs.
   1.
   2.
   3.
   4.
   5.
9. Unidentified lots/exs.taken-
10. Other data, if any -

.....
Signature of the Enumerator

(Indicate here when this data was
incorporated on cards/register etc.)

Laboratory Work

1. Label and put suitable preservation in all lots.

2. Note the name and address of a specialist for the relevant animal group/family.
   Write to him asking his permission to send the material for identification.
   Despatch the material to specialist by Registered mail
   (Always ensure that extra sufficient paddings have been given around the specimen containers).
   Keep a duplicate list of specimens in own file, recording all particulars of the specimens sent.
   On identification, note data on cards and preserve specimens.

3. Consult literature.
   Note reference which give description and/or figure of species.
   Note regional lists, if any.
   Note the broad range of distribution of each species.
   Note the interrelationships with other animals/plants, if any.
   Note natural habitat and preferred niches for each species.
   Note population status and seasonal fluctuations etc.

4. Complete the Species Card entries
   Prepare a final Systematic list.
   Add synonyms, if known.

5. Arrange for publication of the faunal inventory.

Sample Species Card

Genus :
Species (and author) :
Common name :

Phylum :
Class :
Order :
Family :

Description & figure In (Reference)

S. No.	Reg. No.	Locality	Collector/Donor	Date of Collection	No. of exs. (and sex)	D/W
1.						
2.						
3.						
4.						
5.						

List of Endangered Species

S. No.	Name of species	Country/Region where endangered	Last record	IUCN category	CITES Appendix No.	I.W.L. (P) Act Schedule No.
1.						
2.						
3.						
4.						
5.						

Proforma for a Systematic List for Faunal Inventory

Phylum
Class
Order
Family

Genus

1. Species
   Original Reference -
   Recent Reference -
   Common Name, if any -
   Diversity Index -
   Material examined (no. of exs./sex)-
   Hosts(s) (if relevant)-
   Natural habitat -
   Population status -
   Distribution

   i) Within Country (Province/Distt./Locality) -
   ii) Outside Country

2.
3.
4.
5.

Genus -

1. Species -
2.

Family -

Genus

1. Species
2.
3.
4.

Selected References

Zoological survey of India Publications:

1980. Proceedings of the Workshop on techniques in parasitology: 148 pp.

1982. Handbook of Insect Collection, preservation and study: 65 pp.

1986. Manual: Collection, preservation and identification of insects and mites of economic importance: 307 pp.

1988. Management of Mammal collection in tropical environment: 654 pp.

1990. Taxonomy in environment and biology: 330 pp.

1990. Handbook of collection and preservation of animals: 236 pp.

1991. Animal resources of India: protozoa to Mammalia: State of the art: 694 pp.

1987. Onwards. State Fauna Series. Fauna of Orissa (4 vols.); Fauna of Lakshadweep (one vol.); Fauna of West Bengal 912 vols.); Fauna of Andhra Pradesh (one vol.); Fauna of Meghalya 910 vols.); Fauna of Delhi (one vol.).