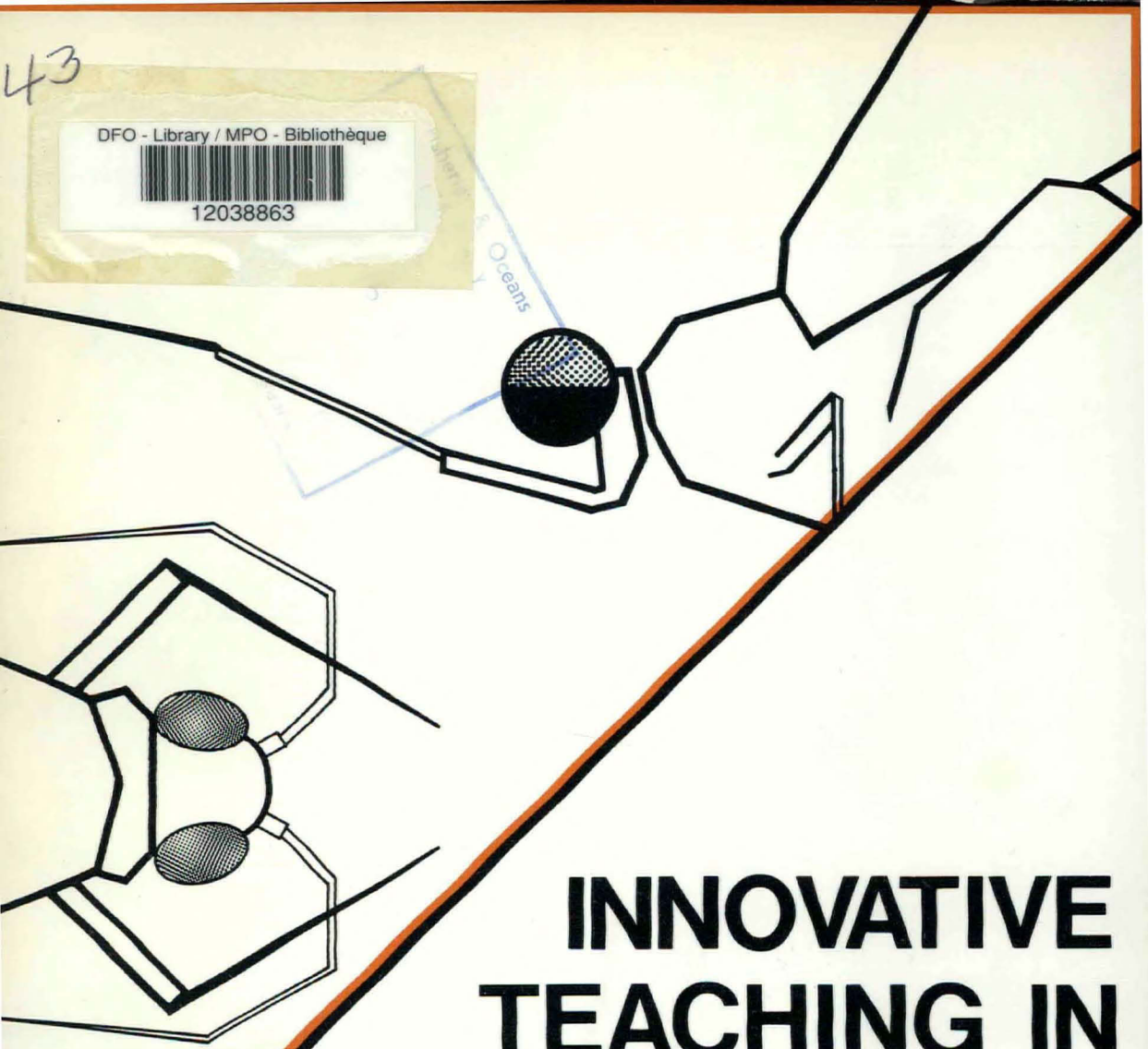

INNOVATIVE TEACHING IN AQUATIC ENTOMOLOGY

Edited by
V.H. RESH
D.M. ROSENBERG

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INNOVATIVE TEACHING IN AQUATIC ENTOMOLOGY

EDITED BY

VINCENT H. RESH
DIVISION OF ENTOMOLOGY AND PARASITOLOGY
UNIVERSITY OF CALIFORNIA
BERKELEY, CALIFORNIA 94720

AND

DAVID M. ROSENBERG
DEPARTMENT OF FISHERIES AND OCEANS
FRESHWATER INSTITUTE
WINNIPEG, MANITOBA R3T 2N6

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CONTENTS (concluded)

ABSTRACT

Resh, V. H., and D. M. Rosenberg (ed.). 1979. Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43: 118 p.

Various approaches used in teaching aquatic entomology are presented. An introductory paper (Morse) surveys methods used in North America for teaching taxonomy of aquatic insects. The subsequent papers can be grouped into: (1) primarily classroom/laboratory exercises, including data simulation in statistical evaluation (Green), nitrogen transformations in stream sediments (Kaushik, Robinson and Chatarpaul), stonefly drumming behavior (Szczytko and Stewart), a method for projecting images of live aquatic insects (Berg), and demonstrations of aquatic insect behavior (Anderson); (2) primarily field exercises, including the use of study streams in Michigan (White) and winter field field trips in Ontario (Mackay); and (3) combined classroom/laboratory and field exercises, including analyses of species associations (Hart) and species-area patterns of insects on stream rocks (McGinniss and Trush), development of self-instructional modules which demonstrate principles of aquatic entomology to undergraduate students (Gottfried and Resh) and graduate-level students (Stewart), and demonstrations of stream watershed community processes (Merritt, Cummins and Barnes).

RÉSUMÉ

Resh, V. H., and D. M. Rosenberg (ed.). 1979. Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43: 118 p.

On présente différentes méthodes d'enseignement de l'entomologie aquatique. Un document d'introduction (Morse) examine les méthodes d'enseignement de la taxonomie des insectes aquatiques utilisées en Amérique du Nord. Les autres documents peuvent être classés ainsi qu'il suit: (1) essentiellement, des expériences en classe ou en laboratoire portant notamment sur les données simulées dans l'évaluation statistique (Green), les transformations de l'azote contenue dans les sédiments de cours d'eau (Kaushik, Robinson et Chatarpaul), le tambourinement de la plécoptère (Szczytko et Stewart), une méthode de la projection d'images d'insectes aquatiques vivants (Berg), et la démonstration du comportement des insectes aquatiques (Anderson); (2) principalement, des expériences en milieu naturel, notamment sur des cours d'eau choisis comme sites d'étude au Michigan (White) et des observations d'hiver en milieu naturel en Ontario (Mackay); (3) une combinaison d'expériences en classe ou en laboratoire et en milieu naturel portant notamment sur l'analyse des associations d'espèces (Hart) et les modèles région-espèces d'insectes sur les rochers d'un ruisseau (McGinniss et Trush), la mise au point de cours d'instruction programmés (appelés "modules") qui démontrent les principes de l'entomologie aquatique aux étudiants de premier cycle (Gottfried et Resh) et de deuxième cycle (Stewart), et l'illustration du développement de biocénoses dans le bassin versant d'un cours d'eau (Merritt, Cummins et Barnes).

FOREWORD

Most of the papers in this publication were presented at two symposia: *Innovative Teaching Approaches in Benthic Sciences*, held during the 26th Annual Meeting of the North American Benthological Society, May 10-12, 1978, Winnipeg, Canada, and *Innovative Teaching Techniques in Aquatic Entomology*, held during the Annual Meeting of the Entomological Society of America, November 26-30, 1978, Houston, Texas, and sponsored by the Teaching and Aquatic Insect Subsections of the E.S.A.

In organizing these symposia, several patterns appeared that were not anticipated. First, although we issued invitations to present papers at these symposia we expected some refusals and had planned to complete the program with contributed papers. However, almost all of the invited authors accepted immediately! Thus, we had to omit many useful and important contributions on this topic, which, hopefully, will be presented in future symposia. Second, after announcements of the symposia appeared, numerous requests for advance copies of the proceedings were received. Those requesting advance copies often indicated that a "proven set" of activities would be useful in helping to teach courses in subject areas (e.g. aquatic entomology, stream ecology, benthic ecology, water pollution biology) in which they themselves had not taken courses. Third, to some, use of the word *innovative* represented faddism in education. Others expressed the attitude that "the organisms are interesting enough and gimmicks will only detract from them". However, we think that the following papers are neither fads nor gimmicks but, rather, they represent concepts and ideas that enhance the teaching of aquatic entomology.

Interest and enrollment in aquatic entomology courses have grown tremendously, as can be seen in John Morse's presentation. We expect that this trend will continue and we hope that additional symposia emphasizing teaching techniques will be held.

We thank the Freshwater Institute, Department of Fisheries and Oceans, Winnipeg, Canada, and the North American Benthological Society for financial assistance in the publication of these proceedings. Dr. Kenneth Stewart, North Texas State University, Denton, provided major assistance in the organization of the symposia while President of N.A.B.S. and Chairman of the Aquatic Insect Subsection of the E.S.A. We thank Dr. Dennis Lehmkuhl, University of Saskatchewan, Saskatoon, Dr. William Peters, Florida A & M University, Tallahassee, and Mr. Jerome Jones, University of Maryland, College Park, for presenting papers at the symposia but which are not included in the proceedings. We also thank the many reviewers for assisting in the preparation of this volume. Mr. Eric Marshall, Ms. Mary Layton, and Ms. Jan Steeves, Freshwater Institute, provided editorial advice and assistance and Mr. Laurie Taite, Freshwater Institute, helped with graphics and layout. Ms. Carol Larson and Ms. Christine Swendrowski typed the many drafts of the manuscript.

V.H.R.

D.M.R.

Fall, 1979

TECHNIQUES USED IN TEACHING AQUATIC INSECT TAXONOMY IN NORTH AMERICAN COLLEGES AND UNIVERSITIES[1]

John C. Morse

Department of Entomology & Economic Zoology, Clemson University
Clemson, South Carolina 29631

ABSTRACT

Instruction in the identification of aquatic insects is a necessary aspect of courses in aquatic entomology. To discover how much emphasis this aspect receives and how taxonomic training is accomplished in such courses in North American colleges and universities, a questionnaire was sent to selected instructors. By this means, a wide variety of useful didactic techniques was revealed, many of which are a credit to the resourcefulness and sensitivity of these aquatic entomology teachers. An ancillary benefit of the survey was the compilation of a roster of instructors for courses in aquatic entomology in North America and a general survey of those courses. The latter survey demonstrated that at least 79 instructors teach aquatic insect courses in 71 colleges and universities in North America to an average of more than 820 students annually.

INTRODUCTION

In view of the fact that training in the identification of aquatic insects is an essential feature of courses in aquatic entomology (either directly within the scope of such courses or as a prerequisite for them), a discussion of how this taxonomy is taught in North American colleges and universities is especially germane to this symposium. The purpose of this paper is to review these didactic techniques briefly, highlighting selected practices that have proven effective or that are particularly novel and have potential for more general use.

METHODS

To conduct this review, it was first necessary to distribute a questionnaire for completion by North American instructors of courses in aquatic entomology. For purposes of this discussion, a course is considered an "aquatic entomology course" or an "aquatic insect course" if at least 50% of the subject matter concerns aquatic Insecta.

Initially, 50 questionnaires were sent in August, 1978, to various known or reported instructors of such courses. The first page of this four-page questionnaire inquired about general features of the course. The other three pages asked for specific information on teaching aquatic insect systematics.

[1]Paper presented in the symposium on *Innovative Teaching Techniques in Aquatic Entomology* at the Annual Meeting of the Entomological Society of America, Houston, Texas, November 26 - 30, 1978.

Table 1. Roster of Instructors for Courses in Aquatic Entomology in North America (compiled by J. C. Morse and V. H. Resh, 1978)

N H Anderson	Dept of Entomology	Oregon State Univ	Corvallis, OR 97331
R D Anderson	Dept of Life Sciences	S Utah State Col	Cedar City, UT 84720
S L Arnold	Dept of Biological Sci	State Univ Col	Brockport, NY 14420
R W Baumann	Dept of Zoology	Brigham Young Univ	Provo, UT 84602
W M Beck	Univ P O Box 111	Florida A & M Univ	Tallahassee, FL 32307
C O Berg	Dept of Entomology	Cornell Univ	Ithaca, NY 14853
L Berner (at Univ of Minn)	Dept of Zoology	Univ of Florida	Gainesville, FL 32611
H A Borchers	Dept of Biology	Bemidji State Col	Bemidji, MN 56601
J Brezner & H Miller	Dept of Envir & For Biol	SUNY Col of Envir Sci & For	Syracuse, NY 13210
W U Brigham & J D Unzicker	(at Univ of Illinois)	Illinois Natural Hist Surv	Urbana, IL 61801
N W Britt	Dept of Entomology	Ohio State Univ	Columbus, OH 43210
D F M Brown	Dept of Biology	E Michigan Univ	Ypsilanti, MI 48197
M A Brusven	Dept of Entomology	Univ of Idaho	Moscow, ID 83843
G B Castle	Dept of Zoology	Arizona State Univ	Tempe, AZ 85281
H F Clifford	Dept of Zoology	Univ of Alberta	Edmonton, Alta. T6G 2E1
W P Coffman	Dept of Biological Sci	Univ of Pittsburg	Pittsburg, PA 15260
E F Cook	Dept of Ent, Fish & Wldlf	Univ of Minnesota	St. Paul, MN 55108
C V Covell, Jr	Dept of Biology	Univ of Louisville	Louisville, KY 40208
B C Cowell	Dept of Biology	Univ of S Florida	Tampa, FL 33620
M E Dakin, Jr	Dept of Biology	Univ of SW Louisiana	Lafayette, LA 70504
W A Drew	Dept of Entomology	Oklahoma State Univ	Stillwater, OK 74074
G F Edmunds, Jr	Dept of Biology	Univ of Utah	Salt Lake City, UT 84112
D A Etnier	Dept of Zoology	Univ of Tennessee	Knoxville, TN 37916
B A Foote	Dept of Biological Sci	Kent State Univ	Kent, OH 44242
W D Fronk	Dept of Zoology & Entomol	Colorado State Univ	Ft. Collins, CO 80591
R C Funk	Dept of Zoology	E Illinois Univ	Charleston, IL 61920
K E Gibbs	Dept of Entomology	Univ of Maine	Orono, ME 04473
A A Grigarick	Dept of Entomology	Univ of California	Davis, CA 95616
R Gundersen	Dept of Biological Sci	St. Cloud State Univ	St. Cloud, MN 56301
R D Hall	Dept of Entomology	Univ of Missouri	Columbia, MO 65201
W J Hanson	Dept of Biology	Utah State Univ	Logan, UT 84322
W N Harman	Dept of Biology	SUNY	Oneonta, NY 13825
R A Hellenthal & G B Craig	Dept of Biology	Univ of Notre Dame	Notre Dame, IN 46556
W L Hilsenhoff	Dept of Entomology	Univ of Wisconsin	Madison, WI 53706
F G Howell	Dept of Biology	Univ of S Mississippi	Hattiesburg, MS 39401
G Z Jacobi	Col of Natural Sciences	Univ of Wisconsin	Stevens Point, WI 54481
K C Kim	Dept of Entomology	Pennsylvania State Univ	University Park, PA 16802
P K Lago	Dept of Biology	Univ of Mississippi	University, MS 38677
R W Lake	Dept of Entomol & Appl Ec	Univ of Delaware	Newark, DE 19711
D M Lehmkuhl	Dept of Biology	Univ of Saskatchewan	Saskatoon, Sask. S7N 0W0
R J Mackay & G B Wiggins	Dept of Zoology	Univ of Toronto	Toronto, Ont. M5S 1A1
G R Marzolf	Div of Biology	Kansas State Univ	Manhattan, KS 66506
W P McCafferty	Dept of Entomology	Purdue Univ	W Lafayette, IN 47907
R W Merritt & K W Cummins	Dept of Entomology	Michigan State Univ	E Lansing, MI 48824
G W Minshall	Dept of Biology	Idaho State Univ	Pocatello, ID 83209
J C Morse	Dept of Entomol & Ec Zool	Clemson Univ	Clemson, SC 29631
S Mozley	Dept of Zoology	North Carolina State Univ	Raleigh, NC 27614
M S Mulla	Dept of Entomology	Univ of California	Riverside, CA 92521
C M Murvosh	Dept of Biological Sci	Univ of Nevada	Las Vegas, NV 89154
C Olson & R L Smith	Dept of Entomology	Univ of Arizona	Tucson, AZ 85721
K P Pruess	Dept of Entomology	Univ of Nebraska	Lincoln, NE 68583
S Radinovsky	Dept of Biology	Millersville State Col	Millersville, PA 17551
V H Resh	Div of Entomol & Parasit	Univ of California	Berkeley, CA 94720
S S Roback (at Univ of PA)	Dept of Entomology	Acad of Natural Sciences	Philadelphia, PA 19406
J F Scheiring	Dept of Biology	Univ of Alabama	University, AL 35486
J H Shaddy	Dept of Biology	NE Missouri State Univ	Kirksville, MO 63501
A L Sheldon	Dept of Zoology	Univ of Montana	Missoula, MT 59801
J A Slater	Biological Sci Group	Univ of Connecticut	Storrs, CT 06268
E L Sleeper	Dept of Biology	California State Univ	Long Beach, CA 90840
S D Smith	Dept of Biological Sci	Central Washington State U	Ellensburg, WA 98926
S M Smith	Dept of Biology	Univ of Waterloo	Waterloo, Ont. N2L 3G1
P J Spangler	Dept of Entomology	Univ of Maryland	College Park, MD 20742
K W Stewart	Dept of Biological Sci	N Texas State Univ	Denton, TX 76203
D C Tarter	Dept of Biological Sci	Marshall Univ	Huntington, WV 25701
B G Torke	Dept of Biology	Ball State Univ	Muncie, IN 47306
J R Voshell, Jr	Dept of Entomology	VPI & SU	Blacksburg, VA 24061
J B Wallace	Dept of Entomology	Univ of Georgia	Athens, GA 30602
M J Westfall	Dept of Zoology	Univ of Florida	Gainesville, FL 32611
D S White	Sch of Natural Resources	Univ of Michigan	Ann Arbor, MI 48105
P V Winger (at Tenn Tech U)	Sch of Forest Resources	Univ of Georgia	Athens, GA 30602
W C Young & H H Hannan	Dept of Biology	SW Texas State Univ	San Marcos, TX 78666
J R Zimmerman	Dept of Biology	New Mexico State Univ	Las Cruces, NM 88003

TEACHING AQUATIC INSECT TAXONOMY IN NORTH AMERICA

When the results of this survey were presented at the Entomological Society of America meeting in Houston in November, 1978, the names of many other instructors of aquatic insect courses were added by symposium participants to the original list. Accordingly, a modified, one-page, general questionnaire was sent in January, 1979, to 26 additional instructors in an attempt to make the general survey more complete.

RESULTS AND DISCUSSION

General Survey

Table 1 presents a roster of names and addresses of instructors for courses in aquatic entomology in North American colleges and universities, initially compiled by V.H. Resh and this author and later supplemented by information from questionnaire respondents and symposium participants. The 79 instructors listed teach courses at 71 colleges and universities in Canada and the United States (Fig. 1).

Sixty-eight instructors of aquatic insect courses as defined above responded to one or the other of the two questionnaires. The results of this general survey are summarized in Table 2.

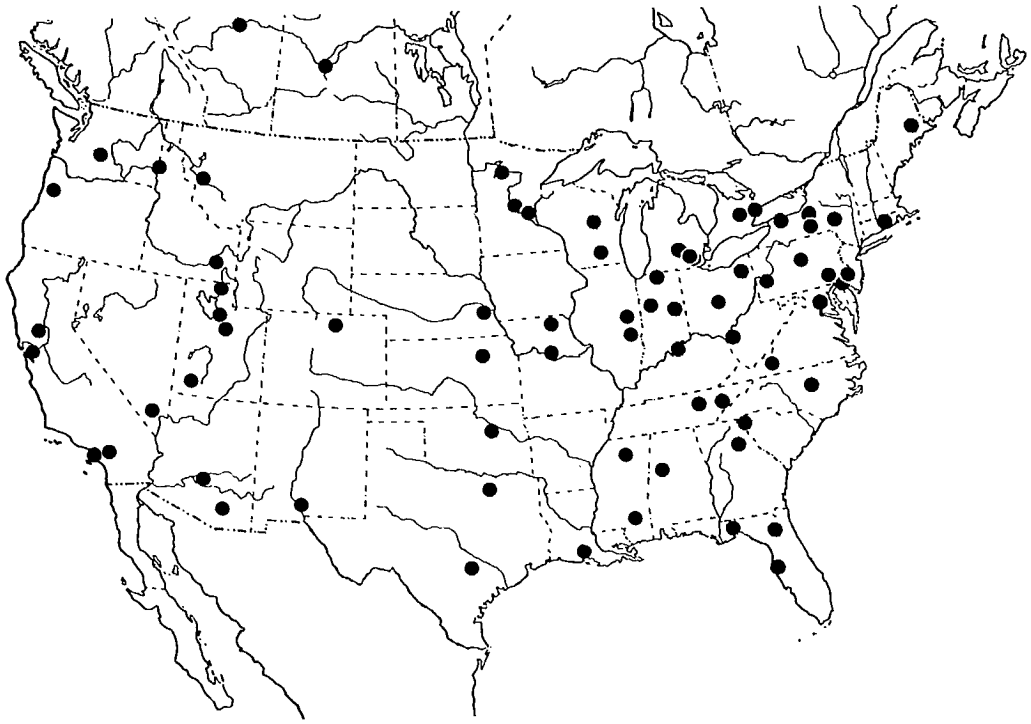

Fig 1. Distribution of North American colleges and universities which offer courses in aquatic entomology.

Table 2. Survey of Aquatic Insect Courses in North American Colleges and Universities

instructor	weekly no. hrs. lecture/ lab.-fld.	total no. hrs. lecture/ lab.-fld.	frequency (per year)	average enrollment	percent of class:[1] fr	so	ju	se	gr	sp	months taught	laboratory asst or instr	relative entomol. emphasis	relative taxonomic emphasis
N. Anderson	2/4	18/36	1	25			40	30	25	5	Apr-Jun	asst	mostly	50%
R. Anderson	3/4	30/40	1	5		10	15	75			Mar-Jun	none	100%	70%
Arnold	2/6.5	27/98	1	12			20	40	40		Jan-May	none	50%	50%
Baumann	2/6	28/76	0.5	B-12			10	40	40	10	Sep-Dec	none	100%	80%
Beck	2/6	---	1	5-6				100			Mar-May	asst	100%	very imp.
Berg	2/6	30/90	1	35		10	30	30	20	10	Jan-May	asst	insects & other macroinv	50%
Berner	2-3/13+	10-15/65+	1	7-10			10	20	70		Jul-Aug	none	100%	heavy
Borchers	3/6	30/60	0.5; variable	---			20	40	20		Sep-Dec; Jun-Jul	none	50%	40%
Brezner & Miller	2/3	26/39	1	25				55	45		Sep-Dec	asst	90%	40%
Brigham & Unzicker	2/4	---	0.5	15				100			Jan-May	asst	100%	75%
Britt	2/4	60/85	1.5	4-16	1	3	7	33	53	3	Mar-Jun; Jul-Aug	occasional asst	100%	70-80%
Brown	2/4	30/60	2	16			5	B5	10		Sep-Dec; Apr-Jul	none	90%	B5%
Brusven	3/4	---	0.5	25			20	40	40		---	asst	10D%	60%
Castle	1/3	---	1	14-22			10	20	70		Jan-Apr	asst	100%	50-60%
Clifford	2/4	---	1	20			40	40	20		Sep-Dec	asst	5D%	60%
Coffman	3/37	12/111	0.5	10			20	20	50	10	May-Jun	asst	100%	60%
Cook	2/4	20/40	0.5	B			20	65	10	5	Mar-May	none	100%	9D%
Covell	2/2	28/28	0.5	8			1	19	79	1	Sep-Dec	none	insects & other arthro	50%
Cowell	2/6	20/60	0.5	10			10	30	50	10	Jan-Apr	none	100%	60%
Dakin	2/2	28/28	0.5	6				50	50		Jan-May	none	100%	50%
Drew	2/4	---	0.5	12				1	97	2	Jan-May	none	100%	50%
Edmunds	2/2	20/80	0.5	10				5D	50		Sep-Dec	instr	10D%	68%
Etnier	2/6	20/60	1	6				5	90	5	Mar-Jun	asst	10D%	60-75%
Foote	8	80+	1	20			5	20	65	10	Apr-Jun	none	100%	70%
Fronk	2/4	30/60	1	30			4	40	56		Sep-Dec	none	100%	75%
Funk	2/2	27/3D	1; variable	B					100		Jan-May; summer	none	100%	75%
Gibbs	2/4	56/112	0.5	10				10	B0	10	Sep-Dec	asst	100%	50%
Grigarick	2/8-11	---	1	27.8			20	64	13	2	Apr-Jun	asst	insects & other macroinv	considerable
Gundersen	2/4	20/40	variable	7				50	50		Jun-Jul	none	90%	55%
Hall	1/3	15/45	0.5	15			30	30	40		Jan-May	asst	100%	60%
Hanson	1/6	10/60	1	6-20			5	10	B5		---	asst	100%	75%
Harman	2/3; 8	15/4B; 120	1	24		20	30	40	10		Jan-Jun; August	none	60%	70%
Hellenthal & Craig	2/3	43/42	0.5	12			5	20	75		Jan-May	asst	100%	66.6%
Hilsenhoff	2/4-6	16/58	1	1B		2	13	25	60		Jan-May	occasional asst	100%	45%
Jacobi	1-2/2-3	---	1	15-20				80	20		Sep-Dec	none	insects & other macroinv	75%
Kim	1/3	---	0.5	25		5	15	50	20	10	Mar-May	asst	arthro	9D%
Lago	2/4	28/56	variable	5				50	50		Jan-Apr; Jul-Aug	asst	100%	50%
Lehmkuhl	3/4	33/44	0.5	7			30	40	30		Sep-Dec	asst	100%	75%
Mackay & Wiggins	2/3	40/108	0.5	20			20	50	25	5	Sep-May	none	100%	10%+
Marzolf	2/6	20/90	0.5	10		10	B0	10			Feb-May	asst	75%	90%
McCafferty	1/6	15/90	1	15			20	50	30		Sep-Dec	asst	mostly	55%

TEACHING AQUATIC INSECT TAXONOMY IN NORTH AMERICA

Table 2. (Continued)

instructor	weekly no. hrs. lecture/ lab.-fld.	total no. hrs. lecture/ lab.-fld.	frequency (per year)	average enrollment	percent of class:[1] fr so ju se gr sp	months taught	laboratory asst or instr	relative entomol. emphasis	relative taxonomic emphasis
Merritt & Cummins	3/3	33/33	1	75-80	4 20 40 34 2	Mar-Jun	instr	100%	50%
Minshal	2/6	--/54	variable	5	50 50	summer	none	insects & other biota	50%
Morse	1/6	14/84	0.5	9	5 25 60 10	Sep-Dec	none	arthro	60%
Mozley	3/3	75/75	variable	7	28 58 14	Jan-Apr	none	insects & other macroinv	40%
Mulla	3/6	30/6D	1	12.5	25 75	Jan-Mar	asst	80%	10%
Murvosh	2/3	30/45	0.5	---	---	Sep-Dec	none	10D%	57.5%
Olson & Smith	1/3	15/45	0.5	13	8 24 38 30	Jan-May	none	100%	75%
Pruess	2/3	30/45	1	10	40 60	Jan-May	none	90%	40%
Radinovsky	3/3	40/60	0.5	12	20 80	summer	none	95%	25%
Resh	3/6	30/60	1	12	20 70 10	Apr-Jun	asst	100%	50%
Roback	1/var.	11/var.	1	4-5	40 60	Jan-May	none	100%	60%
Scheiring	2/3	3D/45	0.5	6	10 15 75	Jan-May	asst	95%	40%
Shaddy	--/4	--/60	variable	---	---	Sep-Dec	none	90%	50%
Sheldon	2/4	20/40	0.5	20-25+	45 45 10	Sep-Dec	asst	100%	60%
Slater	1/4	15/56	0.5	8	20 80	Sep-Dec	none	100%	75%
Sleeper	2/3; 2/3; 1/0	32/48; 32/48+; 16/0	0.5; 0.5; 0.3	15-18; 18-25; 12-15	25 25 25 25; 5 30 30 15 20; 100	Sep-Dec; Feb-May; variable	none; none; none	100%; 80%; 85%	90%; 10%; 15%
Smith	2/6	---	1	5	25 75	Sep-Dec	none	10D%	50%
Stewart	3/3	---	1	8-12	20 80	Jan-Apr	asst	100%	50%
Tarter	2/4	---	1; 0.5	20	20 60 20	Sep-Dec; summer	asst	insects & other biota	most of lab time
Torke	3/2	30/20	1	10	25 25 50	spring	none	arthro	75%
Voshell	2/6	---	1	30	5 45 45 5	Mar-Jun	two instrs	100%	50%
Wallace	3/4	---	0.5	10-15	20 80	---	asst	100%	50-6D%
Westfall	2+/7	15+/75	0.5	10	3 95 2	Jun-Aug	none	---	50%
White	2/6	28/84	1	20	5 5 35 45 10	Jan-Apr	instr	10D%	50%
Winger	1/6	---	1	15	1 99	Jan-Mar	none	insects & other macroinv	80%
Young & Hannan	2/3	36/54	3	15	5 25 20 5	Sep-Dec; Jan-May; Jun-Jul	instr	50%	65%
Zimmerman	2/3	30/45	0.5	8	10 50 40	Jan-May	none	100%	45%

[1] fr = freshmen
so = sophomores
ju = juniors
se = seniors
gr = graduate students
sp = special status or part-time students

Students in these courses receive 1 - 3 h of lecture and generally 3 - 6 h of laboratory or field experience each week in non-summer courses. The total number of hours of lecture in regular session courses ranges from 10 to more than 50 and of laboratory or field trips from 20 to more than 100. Courses taught in summer sessions are more flexible in format than those taught in regular academic months.

Thirty-two courses are taught annually, 32 are taught every other year, two are taught biannually in non-summer sessions, seven (mostly summer) courses are taught on a variable schedule, and one seminar is offered every fourth semester. In some cases, instructors may teach more than one course in any one year or may teach the same course or similar courses more than once in a single year. Average enrollment for the different courses ranges from four to 80. Considering frequency of course offerings and average enrollment for each course, more than 820 students receive training in aquatic insects each year in North American colleges and universities. Less than 1% of these students are freshmen, about 2% are sophomores, 17% juniors, 37% seniors, 40% graduate students, and 3% special status or part-time students.

One course extends over a full 9 mo academic year. Twenty-one courses are taught in the fall quarter or semester, eight in the winter quarter, 14 in the spring quarter, 18 in the winter-to-spring semester, and 13 during the summer months.

Thirty-six courses are taught solely by the course instructor without the help of a laboratory assistant. Twenty-nine courses regularly or at least occasionally have a laboratory assistant. In five courses, most of the responsibility for laboratory instruction is assumed by someone other than the lecture instructor.

Respondents to the August, 1978, questionnaire were asked to mention the textbook used in their courses. The five most-commonly cited books were by Merritt and Cummins (1978, 26 courses); Usinger (1956, 15); Hynes (1970, 4); Pennak (1953, 4); and Hilsenhoff (1975, 2). It should be noted that this poll was taken before the appearance of the revised edition of the volume by Pennak (1978). At least 28 supplementary reference works were specifically listed by the various instructors in addition to their course texts. Instructors also provide mimeographed keys and illustrations from a wide assortment of published and unpublished sources.

In the initial questionnaire, instructors were asked to cite the departments or majors usually represented in their courses. The composite list is long, including biology, entomology, zoology, botany, ecology, fisheries, limnology, environmental education, natural resources, water resources management, sanitation engineering, public health, etc. In general, aquatic entomology courses appear to attract four primary interest groups: fish biologists, insect taxonomists, and freshwater systems ecologists and engineers.

Aquatic Insect Systematics

As is evident in the last column of Table 2, the relative taxonomic emphasis in North American aquatic entomology courses is high, generally more than 50%.

TEACHING AQUATIC INSECT TAXONOMY IN NORTH AMERICA

The 39 instructors who responded to the August, 1978, questionnaire described how they teach aquatic insect systematics by answering various questions, a selection of which are given below along with summaries of the responses.

1. "Do you include any material on biological systematics of aquatic insects in your course, such as phylogenetics, historical biogeography, speciation, etc.?" Most courses in aquatic entomology include very little, if any, subject matter of this sort, despite the fact that many of the leading researchers in biological systematics have been aquatic entomologists. Currently limited class discussion of these subjects is probably due to generally severe time constraints and to the fact that they are often covered in detail in other courses. In those aquatic insect courses where these matters are discussed, the phylogeny of the orders of aquatic insects is often outlined to demonstrate that the freshwater environment has been invaded by insects several times during their evolution, and the phylogenies of certain orders are mentioned to emphasize particular evolutionary trends. Historical biogeography is sometimes discussed and contemporary distribution patterns are often mentioned for the different groups.

2. "Do you generally discuss specific identification characters in lecture? in laboratory? only as students inquire about them? other?" Particular identification characters are generally formally discussed in a laboratory setting, but often during lecture hours in addition. In one course, family-level characters are covered in lecture sessions and generic-level characters in the laboratory. In two of the courses for which students do not collect and identify personal collections, particular diagnostic characters are discussed on an individual basis only as the students inquire about them.

In teaching aquatic insect identification, instructors rely on demonstration materials maintained in special teaching collections or in personal collections or else on material specifically collected by a laboratory assistant or by the whole class in a cooperative effort.

3. "Are the students in your course expected to capture and identify a personal collection of aquatic insects?" In nearly three quarters (29 of 39) of the courses polled, the answer was, "Yes." In three courses such a collection was optional, in five courses the entire class cooperates in the collection and identification of insects, and in three courses no collection is made.

3a. "If yes, are the specimens generally captured during scheduled laboratory sessions or on the student's own time?" For most courses, collections are made both during scheduled field trips and on the student's own time. In seven courses, the students collect mostly or entirely on their own. In nine courses, the collections are generally made during scheduled field trips.

3b. "Are the specimens generally identified during scheduled laboratory sessions or on the student's own time?" Most instructors responded, "Both."

3c. "What is your system for scoring or grading each student's collection?" The quality of the collections is evaluated by about as many different methods as there are instructors. Many courses have a required minimum number of taxa or number of specimens and many are evaluated according to some special scoring system, with points credited to the student for accurate identifications or

debited from him or her for erroneous ones. The collections usually are evaluated for accuracy, quantity, and neatness. In a few courses, the collection is specialized in some way or it is designed as a fundamental part of some larger exercise. For example, in one course the entire collection is made up of reared material with all the associated forms included. In some other courses the collections form the basis for quantitative or qualitative reports comparing different benthic communities.

3d. "What percentage of the final course grade is based on the grade for the collection?" Depending on the course, it is 10% to 60% (average - 29%).

3e. "Are the specimens retained by the student or by the school at the conclusion of the course?" About as often as not the students keep their collections, except that in many cases a few specimens are retained by the school to improve its general reference collection.

4. "As a rule, to what level of identification do you formally teach - order? family? genus? species?" Only seven instructors generally limit formal taxonomic instruction to the family level of identification. All others expect students to learn how to identify genera of aquatic insects with available keys. Students are encouraged to identify their material to the species level where possible in 31 of the 39 polled courses and the instructors assist the students in locating appropriate keys for that purpose.

Instructors were asked to indicate the appropriate response for each of the following:

5a. "Do you cover identification of freshwater organisms only? freshwater and brackishwater organisms only? freshwater, brackishwater, and marine organisms?" Thirty-one, five, and two instructors answered respectively.

5b. "Do you include organisms from lotic habitats? lentic habitats? special habitats such as tree holes, pitcher plants, bromeliads, etc.?" Thirty-nine, 39, and 22 instructors answered respectively.

5c. "Do you cover identification of strictly aquatic stages only? aquatic and semiaquatic stages only? all stages of those insects having at least one strictly aquatic stage? all stages of insects associated with bodies of water sometime during their life cycle?" Seven, eight, five, and 17 instructors answered respectively.

6. "Do you administer a laboratory final examination concerning practical aquatic insect identification?" All but two of the instructors responded in the affirmative. These exams vary considerably in their content and their manner of administration. Generally they require the identification of a certain set of specimens within a particular time limit. Some allow students to view each specimen for a designated time period, others allow the students freedom to spend as much time as they want on any single specimen, but nevertheless expect them to complete the exam on time. Some examinations require strictly closed-book, "sight" identifications; others are open-book; and others include some of each type of determination, often asking for "sight" identification of families and open-book identification of genera or species. At least two instructors who administer open-book exams require citation of the keys and key

couplets chosen in deciding an identity so that the student may be given partial credit if his answer is wrong. A few teachers include brief discussion questions along with the specimens to be identified. On the average, the grade on this laboratory exam constitutes nearly 30% (range 7% - 75%) of the overall final grade for the course.

7. "Do you employ any special identification instructional aids other than printed material, such as audiovisual tapes, computer-based identification keys, etc.?" Many said, "No," but others indicated that they use projection slides, wall charts, and overhead projector transparencies. In my course this past fall, students were introduced to identification characters for genera of chironomid larvae through use of the videocassette developed by Thomas J. Fink and Kenneth G. Wood (Dept. of Biology, State University College, Fredonia, NY 14063). One course makes use of the tape/slide series on *Introductory Entomology* produced at Brigham Young University (Tipton 1973).

8. "Do you employ any other techniques that you consider useful for teaching aquatic insect identification and that you would be willing to have me share with our colleagues at the Houston symposium?" The responses to this question were fascinating! Some examples may be summarized as follows. C.O. Berg, W.P. Coffman, and J.D. Unzicker use projection slides or transparencies to illustrate difficult alternatives in the keys before the students actually look at the specimens. W.L. Hilsenhoff goes a step further by setting out numbered Stendor dishes with two specimens in each, the number marked on each dish corresponding with the number of the key couplet whose alternatives are demonstrated by the specimens.

Many instructors use identified reference collections with which students attempt to work the keys, hoping to arrive at the same answer as is indicated on the determination label. Each specimen in D.S. White's reference collection is identified only by a number. After a student works a specimen through the key, he can check himself by referring to the corresponding number posted elsewhere in the laboratory. W.M. Beck and C. Olson take special effort to include specimens in their reference collections which are not local, thereby broadening their students' training.

Several teachers ask their students to undertake special research projects which are often of a taxonomic nature, such as rearing and associating adult and immature stages, examining longitudinal zonation or other faunistic changes or differences, and studying behavioral or ecological differences between particular taxa. In S.D. Smith's course, students undertake species-level identifications in a group of their choice as a special project.

A number of instructors emphasized the importance of carefully planned field trips because of the enthusiasm that is generated for learning the names of insects and because of the observations of special behaviors and microhabitats that help students remember those names. J.B. Wallace posts a list of so-called "star bugs" which are rare or which live in unusual habitats. The students research these, then go out and capture them, gaining double credit for including them in their collections because of the extra effort involved and obviously learning something more about them in the process.

Lists of the local or regional faunas are provided in several courses to assist students in working through general keys more rapidly and in gaining greater confidence in their conclusions. C.V. Covell's classes provide regular additions to a continuing stream survey of Harrod's Creek, Kentucky. Other instructors similarly update local lists with class-collected data.

Because of the number of fisheries students in their courses, W.J. Hanson and R.W. Merritt include real or contrived "fish gut samples" of broken and mixed insects in their laboratory practical exams. Some others of us administer frequent, short identification quizzes throughout the course in order to encourage students to keep up with the considerable amount of taxonomic information to be learned.

Guest speakers who are specialists in particular groups are regularly invited to address R.W. Merritt's classes. Occasionally, others of us are able to have such experts visit our classrooms for this purpose, also.

CONCLUSION

In summary, it is clear that a large number of students receive special training in aquatic entomology each year in North American colleges and universities (frankly, about four times as many students as this writer estimated before undertaking compilation of the roster and the general survey). The courses vary greatly in (1) the total and the relative amount of lecture and laboratory exposure, (2) class size, (3) extent of prior academic experience and diversity of major interests represented by the students in the classes, (4) season and frequency of course offerings, (5) amount of laboratory assistance provided to the instructor, (6) relative entomological emphasis, and (7) texts and supporting reference literature used.

The amount of emphasis on aquatic insect systematics in these courses also is highly variable, but generally encompasses more than half of the learning experience. Most of this training is in aquatic insect identification rather than in biological systematics and generally is in identification to the lowest taxonomic level possible with available literature. Classroom and laboratory instruction in taxonomy is usually supported with requirements for personal collections and for final laboratory identification examinations, both of which vary considerably from one course to another in their expectations and manner of administration and grading. It is evident from the responses to the survey question regarding novel or other especially effective techniques for teaching aquatic insect identification that North American instructors in these courses are imaginative, resourceful and sensitive to the needs and budding enthusiasm of the annual flock of students who come into their classrooms to learn more about aquatic insects.

ACKNOWLEDGMENTS

The astonishing percentage of return from the two questionnaires (96% - 74 responses to 77 mailouts) is a credit to the vitality of the discipline and to the cooperative attitude among aquatic entomology instructors. I am genuinely grateful to each one. V.H. Resh is to be commended for a superb accomplishment in having organized and moderated the two sessions of the symposium and in having co-edited its proceedings. He was especially helpful to me in our initial

efforts to compile the roster in Table 1.

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SIMULATION OF ECOLOGICAL DATA FOR HYPOTHESIS-TESTING AND EVALUATION OF SAMPLING DESIGNS[1]

Roger H. Green

Department of Zoology, University of Western Ontario
London, Ontario N6A 5B7

ABSTRACT

Data simulation can be used in evaluating sampling designs, determining robustness of statistical methods and for hypothesis-testing, and should be included in quantitative methods courses in benthic ecology. Simulation provides data with known properties, in contrast to data obtained by actual field sampling. Exercises, using simulated data, are presented for: (1) determining the consequences of using different sample unit sizes to sample organisms having different spatial distributions; and (2) testing the H_0 that there is no change in abundance of an indicator species in an impacted area when the data violate assumptions of standard tests. It is also possible to simulate entire multi-species communities on environmental gradients to be sampled for evaluation of multivariate procedures.

This paper recommends, and illustrates, the use of data simulation in quantitative methods courses providing training for benthic ecology. Appropriate areas for such use are evaluation of sampling designs, determining robustness of statistical methods (i.e. their insensitivity to violations of assumptions), and for hypothesis-testing by a method which eliminates dependence on cookbook tests. Students will gain understanding of two different kinds: (1) familiarity with the performance of particular sampling designs and statistical methods on data which violate assumptions in varying degrees, and (2) expertise with a tool - simulation - which they can always use to answer questions such as "What will happen if I sample with this unit size sample?", ...apply this transformation?", ...apply this statistical test?".

Sampling design and statistical analysis methods necessary for benthic studies are often learned in introductory statistics courses without biological orientation, and when the students eventually must apply their knowledge to data derived from sampling organisms in natural environments they may be greatly frustrated. The assumptions underlying the statistics they have learned - linear models and additive, independent, normally distributed errors - are rarely satisfied. Even so-called biometry courses rarely devote much time to the problems which confront ecologists when they must sample natural communities, either for description or for the testing of hypotheses. Nonparametric alternatives to the standard methods are sometimes described, but often in such a way that it suggests to the impressionable student a dichotomy between "good"

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data which satisfy all the assumptions and "bad" data which can only be analyzed by nonparametric methods. The reality is that "the assumptions of most mathematical models are always false to a greater or lesser extent. The relevant question is not whether . . . assumptions are met exactly, but rather whether the plausible violations of the assumptions have serious consequences on the validity of probability statements" (Glass et al. 1972). These authors conclude that the flight to nonparametric statistics is largely unnecessary. "The fact that a particular assumption was used in deriving a test does not mean that violation of that assumption invalidates the test, since the test may be quite robust under . . . violations of the assumptions used to derive it" (Harris 1975). Harris notes that univariate statistical tests appear to be extremely robust under such violations, the major exceptions being for very small and unequal sample sizes and for one-tailed tests. Futhermore, the robustness of nonparametric methods often comes at a price, in that methods with fewer assumptions perform less powerful tests of hypotheses (that is, less ability to reject the null hypothesis when it is in fact false).

Violations of assumptions can be minimized by choosing the most efficient sample unit size and sampling design, with the choice based on data from preliminary sampling. A simple transformation, such as $\log(X + 1)$, may reduce any remaining violations of assumptions to an acceptable level. An excellent inexpensive reference on these subjects is Elliott (1977).

It is of course essential that the student learn the properties of data obtained by actually sampling organisms from a natural community. However, having done so, the distinction between the *estimated* parameters (e.g. mean, variance) from a sample and the true parameters of the population being sampled should be emphasized. It is important that the student understand the limitations of using field data for the evaluation of statistical methods. "Such an approach is unavoidably circular, because the results of a given analysis can only be compared with a preconceived idea of what is in the data, or with what some other analysis method suggests is in the data. The difficulty is that the true properties of the data are not known" (Green 1977). The only way to ensure that data have particular properties is to simulate such data.

For example, the consequences of sampling organisms having different spatial distributions are best illustrated by simulating random and nonrandom distributions and then sampling them in various ways. Consider the question of sample unit size. Most students (and many practicing environmental biologists) intuitively feel that "the larger the better" is a good rule of thumb because they can obtain "more" from each sample. However, there is ample evidence from both theory and practice (Taylor 1953; Elliott 1977) that, for sampling aggregated distributions of organisms, many small samples provide more precise estimates than do a few large samples (given that the same total area or volume is sampled). The following simulated sampling exercise, taken from Green (1979), effectively illustrates this point.

Two simulated spatial distributions are shown in Fig. 1. Square A contains a random distribution of 250 points, each point determined by two-digit coordinates drawn from a table of random numbers. Square B contains an aggregated distribution, for which the first 50 points were allocated at random as before. For the other points it was determined whether each new random point would fall more than a specified distance (the length of a side of the smaller of the two

sample units shown at the top of Fig. 1) from an existing point, and if so it was not entered. If it would fall less than half that distance from an existing point it was entered. If it would fall between half and the full distance from a point then it had a 0.5 probability of being entered. This process was repeated until an additional 200 points were entered, for a total of 250 points.

Each distribution was then randomly sampled using both sample unit sizes. Two replicate sets of 100 samples were obtained from each distribution (A and B) using the smaller sample unit size, and two sets of 25 samples were obtained from each distribution using the larger sample unit size. The expected total number of animals sampled by each set of samples is always 62.5, because the larger sample unit size is four times that of the smaller. The results are shown in Table 1. For the random distribution it makes no difference which sample unit size is used. The standard error to mean ratios ($s_{\bar{x}}/\bar{X}$) are approximately the same. For the aggregated distribution the ratios are much larger for the larger than for the smaller sample unit sizes.

The following, from Green (1979), is an example of the use of simulation for testing a hypothesis where the data violate the assumptions of standard tests. Six replicate samples for abundances of an indicator species are taken before and after an impact occurs, both in an area which will be impacted and

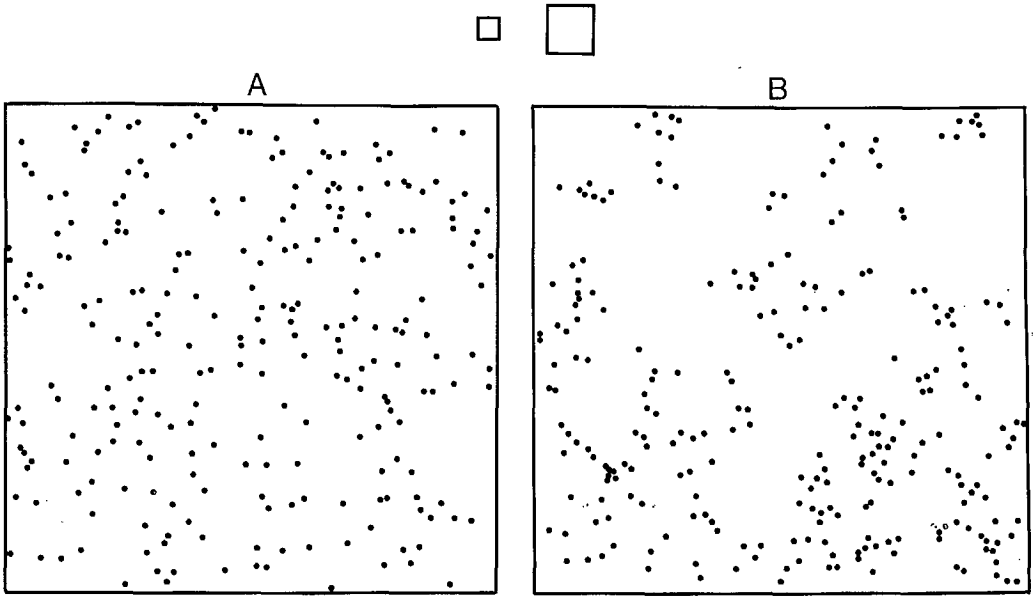

Fig. 1. A random (A) and an aggregated (B) distribution of 250 points. The squares at the top are the two sample unit sizes used for random sampling of these two distributions. (Taken from Figure 3.20 of Green (1979) and used with permission of John Wiley and Sons.)

Table 1. Population parameters and sample statistics for the distributions shown in Figure 1. Sampling is described in the text. (Taken from Table 3.3 of Green (1979) and used with permission of John Wiley and Sons.)

Distribution	Random (A)				Aggregated (B)			
Sample unit size	1		4		1		4	
μ	.625		2.5		.625		2.5	
n	100		25		100		25	
μn	62.5		62.5		62.5		62.5	
Replicate set	1	2	1	2	1	2	1	2
$\bar{X}$	.55	.73	2.80	2.88	.61	.80	1.80	6.8
s^2	.49	.58	2.42	2.69	.81	1.25	4.25	44.3
$s^2/\bar{X}$	.90	.80	.86	.94	1.32	1.56	2.36	6.52
100 $s_{\bar{X}}/\bar{X}$	12.8	10.5	11.1	11.4	14.7	14.0	22.9	19.6

in a control area, for a total of 24 samples (Table 2). The data are simulated to have a mean abundance of $\mu = 50$ except in the impacted area after impact, where there is a reduction in abundance by 75 percent. The standard deviation σ is always half the mean ($\sigma = 0.5\mu$), so that the data are typical of those obtained from sampling benthic organisms: having high variation which varies with the mean abundance. The simulation procedure begins with tabled values of random normal deviates which are then converted to means of log 50 or log 12.5 and standard deviations such that, when antilog'd, $\sigma = 0.5\mu$. See Green (1979) for a more detailed discussion of simulation of these and similar data sets.

To test the null hypothesis (H_0) that there is no change in abundance in the impact area which does not also occur in the control area, we wish to test against an H_0 of no interaction in a two-by-two factorial ANOVA (see Table 2). However, equal within-cell (among-replicate) variances are an assumption of such an analysis, one which we know is violated by these data (because we simulated the data to be that way). With field data of unknown properties, a Bartlett's test (Steele and Torrie 1960) or an F-max test (Sokal and Rohlf 1973) could be used to test the null hypothesis of equal within-cell variances.

How should one proceed, then? It could be demonstrated to the students that a logarithmic transformation stabilizes the within-cell variances, and the ANOVA could then be performed on the transformed data. But would there really be serious consequences if we just ignored the violation and performed the ANOVA on the raw data? It can be shown to the students that they can answer such a question for themselves, by simulating data sets (in a manner similar

SIMULATION OF ECOLOGICAL DATA

Table 2. Abundance of an indicator species in control (C) and impact (I) areas at before-impact (B) and after-impact (A) times. (Taken from Table 2.1 of Green (1979) and used with permission of John Wiley and Sons.)

	C						I					
B	36	67	30	65	40	37	24	60	24	41	95	71
	$\mu = 50$, $\sigma = 0.5\mu$						$\mu = 50$, $\sigma = 0.5\mu$					
	$\bar{X} = 45.8$, $s = 16.0$						$\bar{X} = 52.5$, $s = 28.1$					
A	36	32	49	59	38	32	8	8	20	12	9	6
	$\mu = 50$, $\sigma = 0.5\mu$						$\mu = 12.5$, $\sigma = 0.5\mu$					
	$\bar{X} = 41.0$, $s = 10.8$						$\bar{X} = 10.5$, $s = 5.1$					

to that previously described) which satisfy the null hypothesis of no interaction (all four cells having the same mean, equal to the overall sample mean 37.45) but which have unequal within-cell variances as estimated by the sample variances, which are 255, 792, 117, and 25.5. Of 100 such data sets which I simulated, not one yielded an ANOVA interaction F-value as large as the value (18.5) from the ANOVA on the untransformed data of Table 2. The 0.95 confidence limits on a proportion estimated from zero occurrences out of 100 observations do not include 0.05, and we therefore reject the null hypothesis of "no interaction" at the five percent level.

This exercise demonstrates the robustness of standard ANOVA tests, and also shows how simulation can be used as a verification in any particular instance. A test of any H_0 can be performed by simulating many data sets which satisfy H_0 but otherwise have the properties of the data in hand. Statistical packages such as SAS (Barr et al. 1976) and SPSS (Nie et al. 1975) permit such simulations to be done easily, and then input directly to any statistical analysis procedure.

Simulation of entire multi-species communities on environmental gradients is possible (Gauch 1976), and these simulated communities can then be sampled to generate data for evaluation on multivariate procedures such as ordination and clustering. LaFrance (1972), for example, uses such an approach to evaluate sample number, size and shape, and multivariate statistical analysis methods. Other references where data simulation was used are identified in the bibliography of Green (1979) and a multivariate version of the two-by-two factorial ANOVA presented here, based on simulated data, is also illustrated.

In summary, practice with simulation of ecological data - graphically or with calculators or computers - should be part of the training of benthic and other ecologists. With such techniques they can evaluate sampling designs, verify the robustness of statistical tests, or actually replace the tests by simulation of null hypothesis data sets. For a good general review paper on

simulation, see Raeside (1976).

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ROLE OF CERTAIN BENTHIC MICROORGANISMS AND INVERTEBRATES IN NITROGEN TRANSFORMATIONS IN STREAM SEDIMENTS

N.K. Kaushik, J.B. Robinson, and L. Chatarpaul[1]

Department of Environmental Biology, University of Guelph
Guelph, Ontario N1G 2W1

ABSTRACT

A simple classroom experiment is described to illustrate some aspects of nitrogen transformations in stream sediments and the role that microorganisms and oligochaete worms play in these processes. Plexiglass tubes containing stream sediment with or without oligochaete worms and overlain with water or nitrate solution are incubated for varying periods of time. Changes in nitrate concentrations indicate nitrification and denitrification are occurring. These processes are enhanced by the presence of oligochaete worms. Effects of other factors such as temperature, the oxygen content of the water, amount of energy (carbon) in the sediment and sediment depth can also be examined.

INTRODUCTION

Streams are often regarded simply as channels that transport water and suspended and dissolved materials. However, most streams support an enormous variety of living organisms which are involved in many dynamic processes. For environmental management, it is important not only to know the kinds of living organisms that abound in streams and the effects that various pollutants have on them, but also the effects these organisms have on pollutants. This is especially true for non-point or diffused pollutants (e.g. fungicides, pesticides, nitrogen, phosphorus) which, unlike point source pollutants, may be difficult to prevent or manage.

Studies on nitrogen transformation processes in aquatic environments mostly pertain to lakes (see Keeney 1973; Kamp-Nielsen and Andersen 1977 for reviews). Investigations dealing with flowing waters are not as common (Sain et al. 1977; Van Kessel 1977). The sediment-water interface plays an important role in nitrogen transformation in aquatic environments. Whether or not the water overlying the sediment is anoxic affects these processes. In lakes, especially deeper, eutrophic ones, anoxic conditions often occur at the sediment-water interface. In streams, because of turbulence, flow and shallowness, the water is mostly saturated with oxygen. Despite this, reducing processes may go on just below the sediment surface and can have a dramatic effect on concentration of solutes in the water column. For example, indirect evidence in an English study (Owens et al. 1972) indicated summertime losses of nitrate-N from stream water, probably resulting from the microbial process of denitrification, amounting to 274 g $yr^{-1}m^{-2}$ of river bed. In a small spring-fed brook with only about 2000 m^2 bed area, Robinson et al. (1979) found losses of about 175 g $yr^{-1}m^{-2}$,

[1]Present address: Department of Soil Science, University of Saskatchewan, Saskatoon, Saskatchewan S7N 0W0.

amounting to a total of 350 kg yr^{-1}. The rate of loss was greatest at summer temperatures when about 60% of nitrate input from the spring was lost.

Recently we have been investigating the importance of benthic organisms to some nitrogen transformation processes in stream water and sediment (Sain et al. 1977; Chatarpaul et al. 1979). The laboratory exercise described below uses some of the findings of these investigations for teaching advanced classes in aquatic ecology. The objectives of the experiment are to demonstrate to the student the importance of biological processes in streams vis-a-vis pollutants in transport and to provide an illustration of the interaction of microorganisms and higher benthic organisms.

NITROGEN TRANSFORMATIONS IN STREAMS

Paucity of space does not permit an extensive discussion on nitrogen transformation in streams and freshwater (for details see reviews by Wetzel 1975; and Patrick et al. 1976). Important forms of nitrogen in freshwater are: organic nitrogen (organic compounds with amino acids, amines, proteins), ammonium (NH_4^+), nitrite (NO_2^-) and nitrate (NO_3^-). However, there is continuous input, conversion and loss of N in stream systems and most of the important processes involved are microbial in nature. Organic nitrogen in sediment, originating from leaves, other allochthonous inputs, or stream macrophytes is mineralized as a result of activity of heterotrophic bacteria. The ammonium produced is nitrified, in the aerobic zone near the sediment surface, first to nitrite and then from nitrite to nitrate. The nitrifying bacteria involved in the first step mostly belong to the *Nitrosomonas* group. Microorganisms converting nitrite to nitrate primarily belong to the genus *Nitrobacter*.

Nitrate resulting because of nitrification or entering directly into a stream from ground water or surface run-off diffuses into the sediment. Here eventually it will encounter low redox potentials, be denitrified (i.e. converted to N_2O or N_2) and lost to the atmosphere. The important facultative anaerobic bacteria involved in denitrification belong, among others, to the genera *Pseudomonas*, *Achromobacter*, *Bacillus* and *Micrococcus*.

MATERIAL AND METHODS

Experimental Set Up

Plexiglass columns (Fig. 1) containing stream sediment overlain with nitrate-enriched water are used for the experiments described. We use columns ≈60 cm long, 6.4 cm O.D. and 0.4 cm thick but size is not critical. The water in the columns is constantly aerated to cause turbulence and to maintain high oxygen levels in the water, thus to some extent simulating stream conditions. At least triplicate columns should be used for each treatment to facilitate statistical evaluation.

Preparation of Columns with Sediment

Sediment can be collected by a corer from areas of organic deposits in a stream. Sediment samples thus collected are mixed thoroughly in a plastic bucket, sieved through 0.4 mm mesh to remove larger benthic invertebrates, and refrigerated until used in the demonstration.

NITROGEN TRANSFORMATION IN STREAM SEDIMENTS

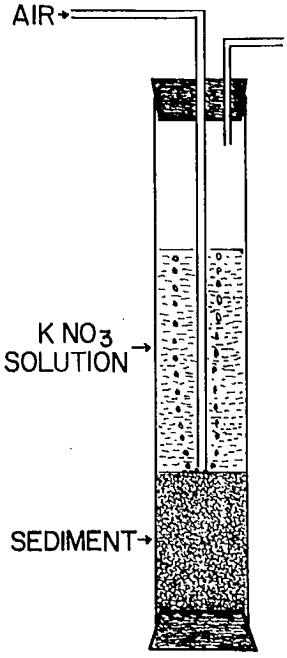

Fig. 1. A plexiglass column with stream sediment and KNO_3 solution for classroom experiments.

Oligochaete worms, *Limnodrilus hoffmeisteri* (Claparède) and *Tubifex tubifex* (Müller) can be collected from streams, rivers or lakes. Generally, they abound in sediments that are rich in organic matter (e.g. downstream of sewage outfalls). The sediment can be washed through a 0.4 mm sieve and the worms collected are stored in sediment at an appropriate temperature (10, 15 or 20°C--depending upon the temperature at which the experiment is to be run). For use in the experiment, a small quantity of the sediment is washed through a 0.25 mm sieve and the worms are collected using a Pasteur pipette.

Ten cm of wet, worm-free sediment is added to each plexiglass column, the lower end of which is stoppered. Air bubbles are purged by lightly tapping the columns. Sediment adhering to the inside of the columns is washed down with deionized water. Twelve columns are prepared with sediment and three are kept without any sediment. The columns are allowed to settle overnight in the refrigerator, the supernatant water is siphoned off, and the following triplicate treatments are used:

i) Columns without sediment but with 500 ml of 10 mg/L KNO_3 solution.

ii) Columns with sediment and 500 ml distilled water.

iii)Columns with sediment, 500 ml distilled water and 40 oligochaete worms per column ($\simeq$15,000 worms m^{-2}).

iv) Columns with sediment and 500 ml of 10 mg/L KNO_3 solution.

v) Columns with sediment, 500 ml of 10 mg/L KNO_3 solution and 40 oligochaete worms per column.

The top ends of the columns are then closed with two-hole rubber stoppers. After 1 day when the sediment is settled, air is introduced through one hole using a sparger located about 1 cm above the sediment-water interface. The columns are then incubated in the dark at the desired temperature (5-25°C). Nitrate concentration in the supernatant is monitored routinely (every other or third day) using the chromotropic acid or phenoldisulfonic acid method or a nitrate electrode (APHA 1975). The latter is the most convenient method for a class. Mean values and standard deviation for each set of samples are then calculated.

RESULTS AND DISCUSSION

As expected, control columns (nitrate solution with no sediment) do not show changes in nitrate concentration during the incubation period. Typical results from the remaining sets of columns are shown in Fig. 2. These data actually were taken from two different experiments and illustrate extreme effects of worms on denitrification and nitrification. In the columns with nitrate solution overlying sediment, the nitrate concentration starts declining and at ≈20°C may attain a value of about 2 mg/L (nitrate N) from the initial 10 mg/L in as little as 2 wk. ^{15}N studies (Chatarpaul and Robinson 1979) have shown that this decrease in nitrate concentration is due to the activity of denitrifying bacteria in the sediment resulting in loss of nitrogen to the atmosphere. The decrease in nitrate concentration is much faster in the columns containing nitrate solution and oligochaetes which indicates an interaction between microbial denitrification and the activity of the oligochaetes.

Our earlier work (Robinson et al. 1979) showed that even when the supernatant was constantly aerated, the oxygenated sediment zone was less than 1 cm thick and the sediment below this depth had reducing conditions where denitrification occurs. Thus, the rate of denitrification is limited by, among other factors, the rate of diffusion of nitrate from the supernatant to the reduced zone in the sediment. It is noteworthy that dentrification goes on in the thin upper layer of reduced sediment and that the concentration of nitrate remains negligible below this active zone.

How do oligochaetes affect this process? It is possible that the burrowing activity of the worms enhances mechanical transfer of both nitrate and oxygen to the deeper zones of sediment. In fact Davis (1974) has shown that tubificid worms deepen the oxidized layer of lake sediment. However, this deepening of the oxidized layer is probably not uniform and microsites with reducing conditions are likely to occur. The greater availability of nitrate at these microsites because of the activities of the worms results in more rapid denitrification. As well, there is some evidence from work in our laboratory that denitrification may occur in the guts of the worms. Pennak (1953) mentioned that some tubificids draw water into the gut through the anus in order to extract oxygen more efficiently from the water. It is possible that nitrate contained in the water may be reduced during this process.

NITROGEN TRANSFORMATIONS IN STREAM SEDIMENTS

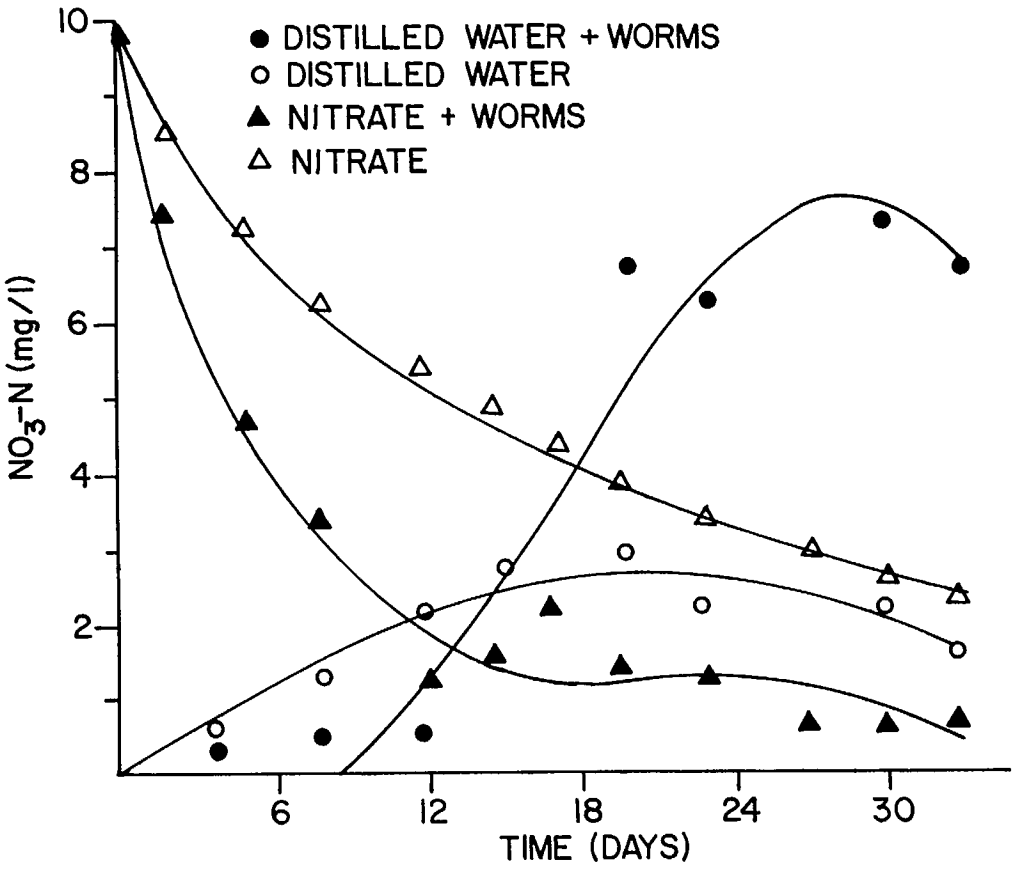

Fig. 2. Changes in NO^3-N concentration in distilled water or nitrate solution overlying stream sediment with and without oligochaete worms.

Oligochaetes also accelerate nitrification. We assume they do so by feeding on N-rich organic matter deep in the reduced zone of the sediment and defecating unused nitrogen in the form of ammonium or easily mineralized organic nitrogen compounds at the aerobic surface of the sediment where the nitrifiers, limited by ammonium, are active. In Fig. 2 the concentration of nitrate-N in columns with distilled water only slowly reaches ≈2 mg/L. In the columns with oligochaetes and distilled water the initial increase in nitrate concentration is slower but the final concentration is much higher. The initial delay in nitrate accumulation presumably results from enhanced denitrification in the presence of the worms.

Effects of other variables can be studied depending upon the class-size. For example, varying sediment depths (1.0, 2.5, 5.0 and 10.0 cm) can be used and columns incubated at different temperatures (5, 10, 15 and 20°C). Temperature is obviously important (Fig. 3) and nitrate disappears slowly at lower temperatures.

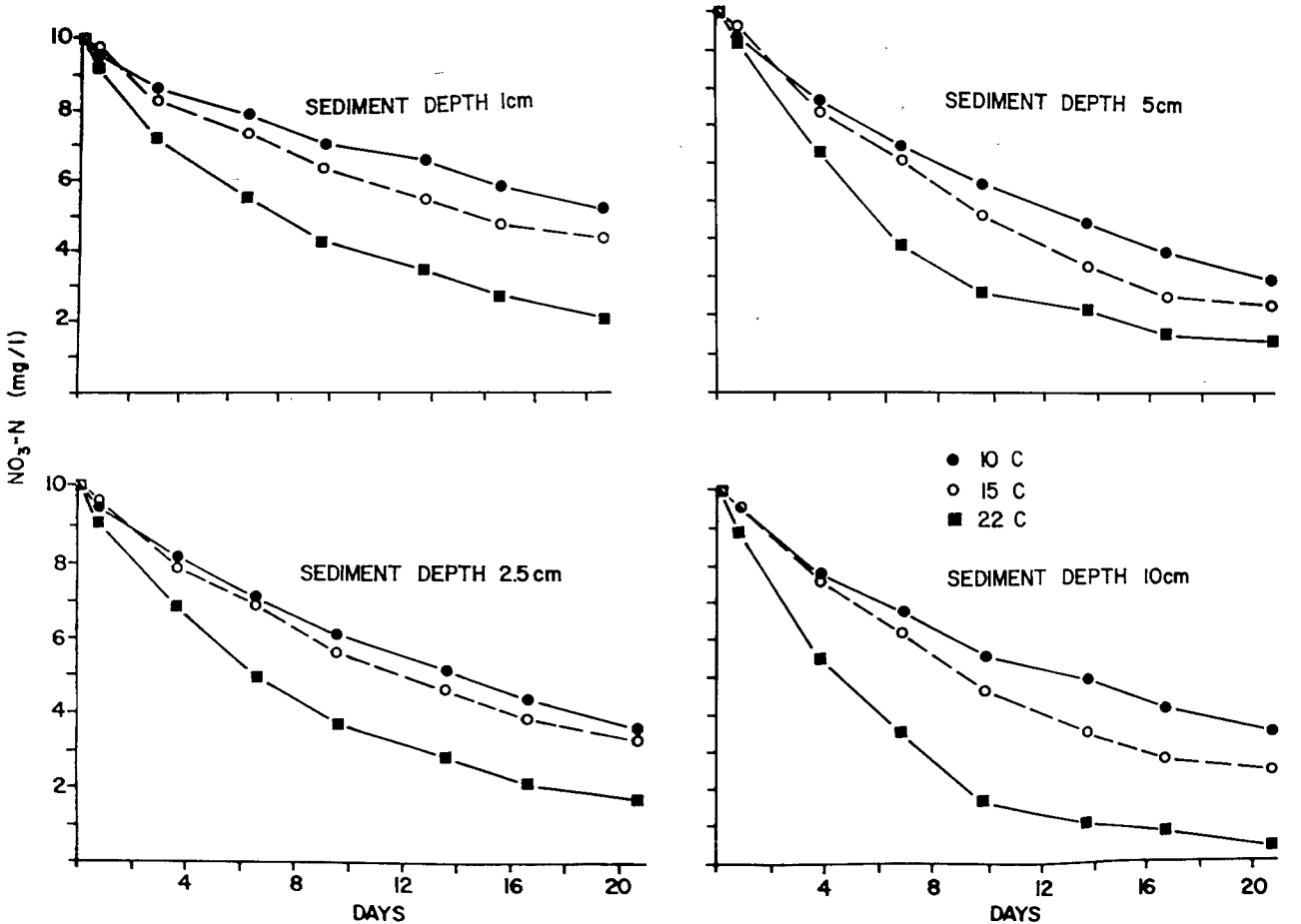

Fig. 3. The effects of temperature and sediment depths on changes in NO_3-N concentration in nitrate solution overlying stream sediment.

However, a positive correlation between nitrate loss and sediment depth does not exist at all temperatures. The importance of sediment depth depends upon temperature. Comparison of mean values of nitrate loss at the end of the experiment shows that at 10 and 15°C there is a significant difference between 1 and 5 cm columns. At 22°C there is no significant difference with sediment depth because denitrification is very rapid at this higher temperature. As the nitrate diffuses from the supernatant into the sediment, it gets denitrified in the top few mm of sediment and, therefore, has little chance to diffuse into the sediment below. Thus, a sediment depth >1 cm at a higher temperature has no impact on the rate of denitrification (for details, see Sain et al. 1977). One could also compare sediments from different streams or study the effects of varying the energy (carbon) content of the sediment by adding different amounts of decomposed and ground leaves (Fig. 4). There is a direct relationship between the amount of added organic matter and rate of nitrate loss. The added organic matter provides an energy source for denitrifying bacteria. Evidently allochthonous organic matter in a stream is not only important as food for stream invertebrates but also influences the rates of biological processes involving nitrogen.

Figure 5 shows results of an experiment in which columns with sediment and nitrate were either left open to the air or oxygen or helium was bubbled into the nitrate solution. We know that anaerobic conditions promote denitrification. The rate of nitrate loss is thus highest when helium is added. However, even when the supernatant is saturated with oxygen, loss of nitrate does occur albeit

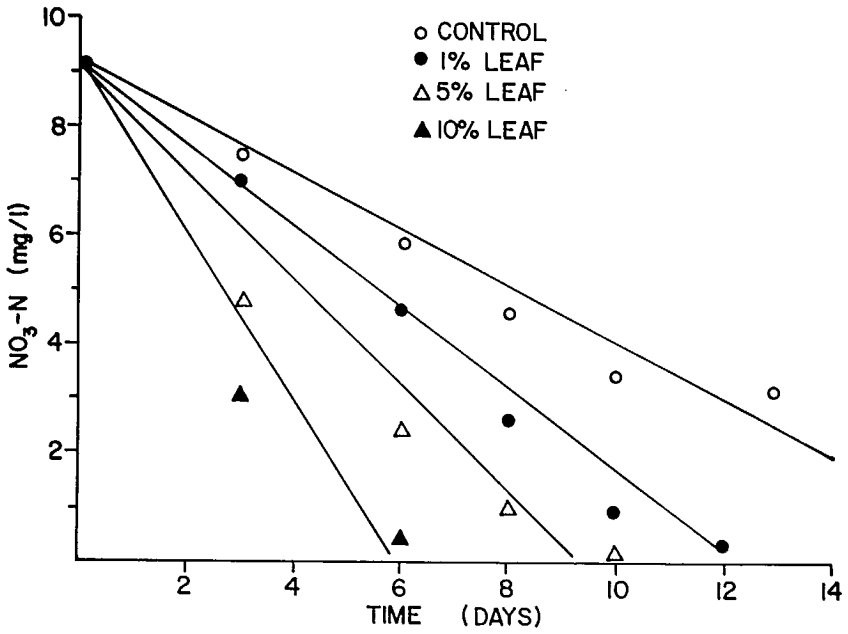

Fig. 4. Changes in NO_3-N concentration in columns following additions of varying amounts of decomposed maple and water cress leaves to the sediment.

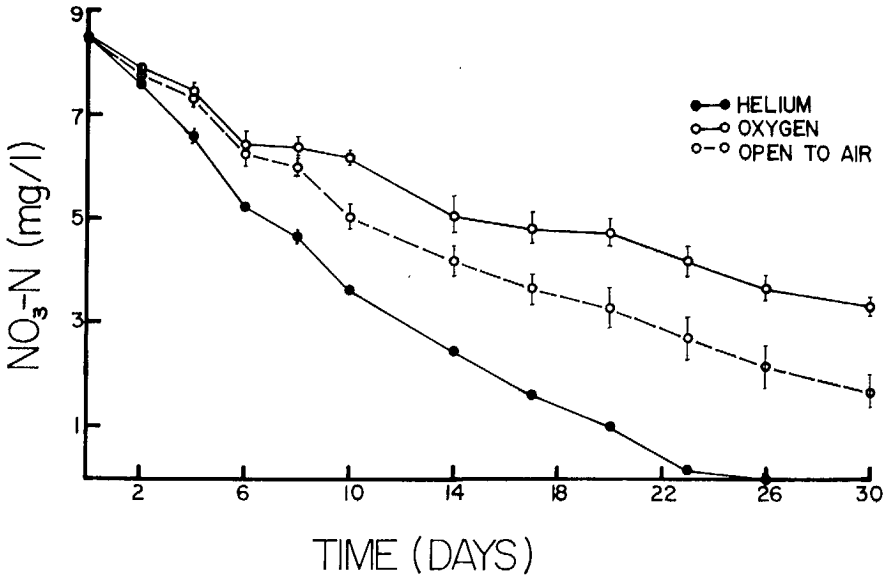

Fig. 5. Changes in NO_3-N concentration in nitrate solution overlying stream sediment in plexiglass columns and either bubbled with helium or oxygen or left open to air.

at a slower rate. It is possible that because of oxygen saturation in water and its diffusion in the sediment, the anaerobic zone does not start just below the sediment-water interface, rather it moves downward. Since for denitrification to proceed, nitrate has to diffuse through this oxidized zone of sediment, the rate of nitrate loss is significantly slower under oxygen-saturated water. Larvae of *Chironomus riparius* Meigen and *C. plumosus* (L.) have been shown to accelerate the rate of denitrification (Edwards 1958; Andersen 1976). Thus some columns can be set up either with these insect larvae or with some other benthic invertebrates that are known to have some burrowing activity. One could also study effects of varying nitrate concentration in the supernatant.

It may be emphasized here that in the column experiments described above, a number of processes involving nitrogen (e.g. ammonification, nitrification and denitrification) proceed simultaneously. In an experiment where ^{15}N is used it is possible to work out rates of such processes. However, for the class demonstrations described here, nitrate concentration at a given time depicts only the final outcome of these interacting processes. For these reasons it is possible that in some instances, depending upon the nature of sediment, effects of some of the variables may be completely masked or altered. However, in doing these column experiments with sediment from a number of streams we have always recorded loss of nitrate from the supernatant, although the rate may have varied widely. Therefore, most natural streams probably do have the capacity for denitrification and loss of nitrogen from the system. While it is difficult to establish nitrogen budgets for streams, we have shown that nitrogen loss does occur in a small headwater stream with high nitrate input (Kaushik and

Robinson 1976) and that this loss probably results from denitrification (Robinson et al. 1979).

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STONEFLY DRUMMING AS A MODEL CLASSROOM STUDY OF AQUATIC INSECT BEHAVIOR[1, 2]

Stanley W. Szczytko and Kenneth W. Stewart

Department of Biological Sciences, North Texas State University
Denton, Texas 76203

ABSTRACT

Stonefly drumming behavior can be used in the classroom to involve students directly with live insects. Instructions are provided for obtaining live material, observing drumming, and recording signals. Possible experiments using combinations of living stonefly nymphs, tape recordings and oscillograph tracings include: (1) illustrating the phenomenon of drumming; (2) demonstrating behavioral species-specificity and potential dialects; (3) studying temperature effects; and (4) examining the capacity of stoneflies to screen their signals through extraneous sounds. Examples are given of other interesting questions to address. Cinemaphotography of drumming stoneflies will allow further classroom investigation of this interesting behavior and is planned for the future.

INTRODUCTION

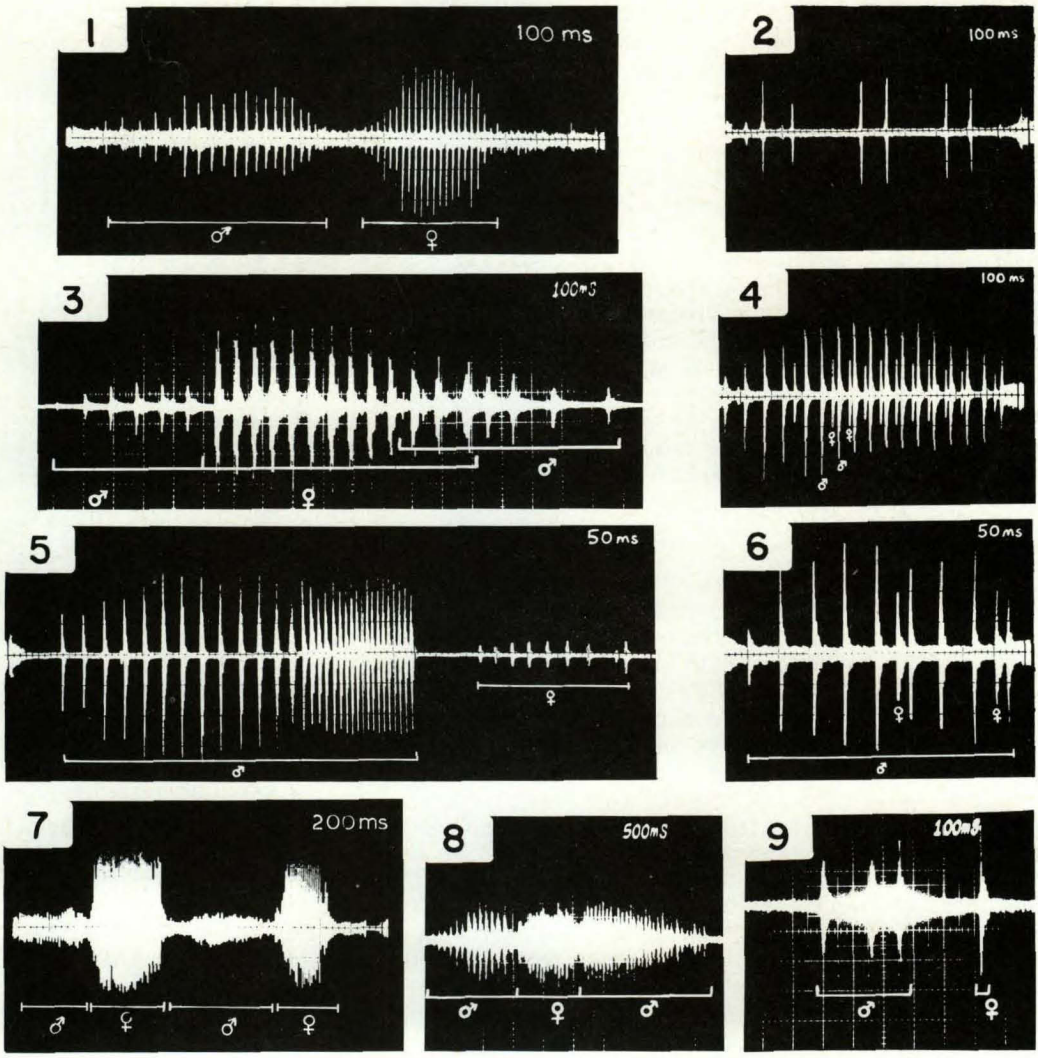
Most adult stoneflies communicate by tapping upon substrate with the terminal-ventral part of their abdomens. This interesting phenomenon, called "drumming" by Newport (1851) was further described by Briggs (1897), MacNamara (1926), Brinck (1949, 1956), Jewett (1959) and Gaufin et al. (1966). Rupprecht (1968, 1969, 1972) pioneered the quantitative and physiological study of drumming in European stoneflies and Ziegler and Stewart (1977) provided the first quantitative accounts of drumming behavior and signals of 11 nearctic species.

We are currently studying drumming, and have found that the accumulated tape data bank and experiments in progress are useful in teaching, and arouse great interest among students in ecology and entomology. Audio recordings and corresponding oscillograph tracings can be used, together with the above background information, for lectures or for laboratory experiments set up to give students "hands-on" interaction with live insects and research tapes.

Male drummers continue their activity throughout their short adult life, but only virgin females respond to male signals. Males almost always initiate the communication and females answer beginning either during the male "drumming

[1]Paper presented in the symposium on *Innovative Teaching Approaches in Benthic Sciences* at the 26th Annual Meeting of the North American Benthological Society, Winnipeg, Canada, May 10 - 12, 1978 and in the symposium on *Innovative Teaching Techniques in Aquatic Entomology* at the Annual Meeting of the Entomological Society of America, Houston, Texas, November 26 - 30, 1978.

[2]Data presented in this paper were collected in part under research support from the Faculty Research Fund of North Texas State University and National Science Foundation Doctoral Dissertation Improvement Grant # DEB 76-15454.



Figs. 1-9. Oscillographs of stonefly (Plecoptera) drumming signals. 1. *Zealeuctra claasseni* male and female (2-way signal); 2. *Isoperla quinquepunctata* female; 3. *Claassenia sabulosa* male and female (3-way signal); 4. *Isoperla quinquepunctata* blending of male and female signals after an intense drumming period; 5. *Isoperla phalerata* male and female (2-way signal); 6. *Isoperla quinquepunctata* male and female (2-way signal); 7. *Zealeuctra hitei* male and female (4-way signal); 8. *Hesperoperla pacifica* male and female (3-way signal); 9. *Perlinella drymo* male and female (2-way signal).

roll" (*Isoperla quinquepunctata* (Banks), Fig. 6) or immediately after (*Zealeuctra claasseni* (Frison), Fig. 1; *Isoperla phalerata* (Needham), Fig. 5; *Perlinella drymo* (Newman), Fig. 9). We have termed this typical sequence a 2-way communication. In some species, a 3-way sequence occurs (as typical or alternating erratically with 2-way sequences), involving males answering back after the female response (*Claassenia sabulosa* (Banks), Fig. 3; *Hesperoperla pacifica* (Banks), Fig. 8; *Pteronarcys californica* (Newport); and occasionally *Pteronarcella badia* (Hagen)). In some *Acroneuria* and *Zealeuctra*, females will answer the second part of the male signal, giving a 4-way sequence (*Zealeuctra hitei* Ricker and Ross, Fig. 7). These signal sequences in most species are separated by a definite time interval, but we have discovered that a "symphony" of intermeshed signals is set up in the drumming of individual or multiple pairs of some *Isoperla* (Fig. 4). Two species we have extensively tested, that apparently do not drum are *Perlesta placida* (Hagen) (Perlidae) and *Hydroperla crosbyi* (Needham and Claassen) (Perlodidae).

We anticipate that many inter-specific variations will be revealed by further research. To date, all studies have indicated that signals are species-specific. It is highly unlikely that drumming is individually learned by stoneflies, since the behavior is practiced perfectly and with little variation by newly emerged adults, obviously not previously exposed to a learning process. This infers a genetic determinant, which, along with the quantifiable nature of drumming, renders it potentially valuable behavioral evidence for phylogenetic analyses and problem-solving in systematics (Zwick 1973; Ziegler and Stewart 1977). The unique nature of drumming in stoneflies can be emphasized to students by comparing it with other insect communication methods such as stridulation (e.g. grasshoppers, crickets) and dancing (e.g. honeybees).

METHODS

Obtaining live material. Since mated females cease drumming, successful experiments usually require rearing of virgins from mature, field-collected nymphs. Males may either be reared or collected as adults. Late-instar nymphs can be successfully reared to adults by maintaining them in stream water at simulated field temperatures, in styrofoam containers. Six-pac styrofoam drink coolers are satisfactory for rearing stoneflies on overnight or longer field excursions. Each compartment accommodates a different collection locality, sex, different species etc., and a small rearing microcosm is created by inserting inverted styrofoam cups into each compartment. Using this method, virgin adults are often obtained in the field, and are held in shell vials until needed for experiments. During hot weather, these six-pacs or other small styrofoam containers can be iced within a larger container.

Since stoneflies exhibit seasonal emergence patterns in most geographic areas, representatives are usually available in most seasons, including the coldest months. Autumnal species (e.g. *Leuctra* sp.) are rare, and therefore would be perhaps the most difficult to obtain. Winter-emerging stoneflies would include members of the Leuctridae (*Zealeuctra*), Capniidae, Taeniopterygidae and some Nemouridae. Most other stoneflies, especially Perlodidae, Chloroperlidae, Perlidae and some Nemouridae (e.g. *Amphinemura*) emerge from early spring through mid-summer. When live material is scarce, or time limited, the taped drumming signals become invaluable supplementary resource material.

Fig. 10. Drumming chamber and recording apparatus.

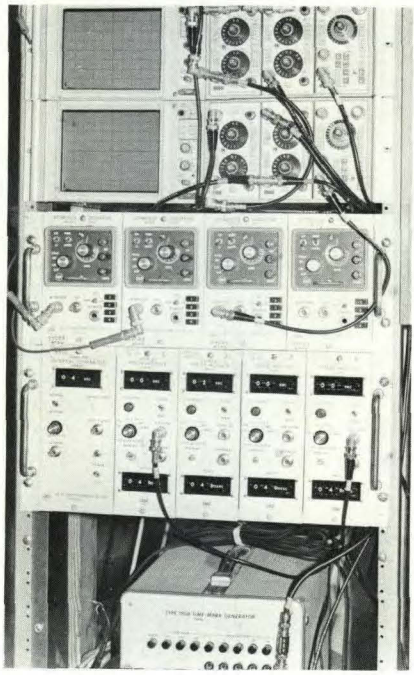


Fig. 11. Oscilloscope set-up.

STONEFLY DRUMMING

Observing drumming and recording signals. Drumming chambers of various sizes (suggested 11 x 7 x 3 cm average size) can be constructed by students using heavyweight manila file folders. This partitioned box is covered by a 14 cm dia. transparent plastic Petri plate cover (Fig. 10). The chamber should rest about 3 mm above a condensor microphone (e.g. a Sony® ECM-955, Fig. 10), on two foam rubber pads. Any high-quality cassette tape recorder and high fidelity cassettes can be used for recording signals (e.g. Sony® TC-142, Fig. 10).

Male and female stoneflies are placed in separate compartments and will often begin drumming after a variable period of acclimation, without artificial enticement. Reluctant stoneflies can usually be induced to drum by gently tapping on the table top or by playing previously recorded signals of the same or sometimes even different species. Taped signals can be displayed as oscilloscope tracings and photographed by playing tapes into one or a group of storage oscilloscopes (Fig. 11). We have used Tektronix 5115 storage oscilloscopes with a Tektronix C-5A oscilloscope camera and Polaroid® Type 107 ASA 3000 black and white film. Once familiar with this procedure, students can be encouraged to use their imagination in designing experiments investigating various aspects of this interesting behavior previously mentioned.

EXPERIMENTS

Background. Probably the most interesting application of this phenomenon to the classroom is the use of live stoneflies collected in the field by the students, in combination with tape recordings. Combinations of the two can be utilized in experiments designed to: (1) illustrate the phenomenon of drumming, (2) demonstrate behavioral species-specificity and potential dialects (geographic or population variation), (3) study temperature effects, and (4) examine the capacity of stoneflies to screen their signals through the natural, extraneous sounds at streamside.

Important parameters for analysis of drumming include: (1) signal length, (2) number of beats in a signal, and (3) signal frequency characteristics. Signal length requires close control, since it is temperature-dependent and could vary if different power sources or recorders are used between original recordings and replay. Number of beats and frequency characteristics are the most important parameters, generally show little variation among individuals of a species, and are species-specific. Number of male beats in the ≈20 nearctic species we have studied have ranged from 3 in *Perlinella drymo* (Fig. 9) to 32 in *Zealeuctra hitei* (Fig. 7). Females generally have fewer beats, ranging from 1 in *Perlinella drymo* (Fig. 9) to 46 in *Zealeuctra hitei* (Fig. 7).

It is therefore possible for students to observe, record, and analyze the 2-way, 3-way and 4-way sequences. Also, signals may be characterized as monophasic, in which all beats have the same frequency (Figs. 1-4, 6-9), or multiphasic, in which the frequency of beats changes as the signal progresses (Fig. 5). Other interesting questions that can be addressed include: (1) Which part of the abdomen do the sexes of different species use to strike the substrate? (2) Is the signal transmitted as sound waves and/or substrate vibrations? (3) What movement patterns are exhibited by males and females of different species once a drumming sequence is initiated?

Examples: Experiment 1. Species-specificity can be demonstrated by playing recorded male signals of several species to live females of a given species. Temperature at which the males were recorded should be matched with that at which the live females are being held. Females may respond to a portion of the signal of a different male species, signal for a few times, and then "recognize" the lack of feedback and cease responding (e.g. *Perlinella drymo*, in Ziegler and Stewart 1977).

Experiment 2. Geographic variations or dialects can be detected by recording signals of specimens collected from different areas. Dialect specificity could then be tested by using live males and females from different areas, or by utilizing taped signals of males from several geographic areas, played to live females from other areas.

Experiment 3. Temperature effects on signal characteristics can be demonstrated in short-term experiments in the lab. Males and females can be recorded at different temperatures, then signals compared (e.g. *Perlinella drymo*, in Ziegler and Stewart 1977).

Experiment 4. The ability of stoneflies to receive and respond to the proper signal frequency and screen out extraneous streamside noise demonstrates the efficacy and importance of signal transmission in natural habitats. This can be demonstrated by playing the recorded roar of a large riffle during drumming of males and females. The drumming chambers should be rested on foam rubber pads or suspended from a wire.

In the future, we plan to supplement the audio recordings and oscillograph tracings with Super-8 cinemaphotography of drumming pairs. Accumulated film would be invaluable in classroom situations, when experiments are not working as planned, or when live stoneflies are unavailable. Film would also enable slow-motion reviewing for analysis, and student discussion of behavioral aspects such as searching movements, body position and body structures utilized during drumming. Further study of this unique method of insect communication will undoubtedly reveal a great diversity of drumming propensity and signal characteristics in stoneflies. Thus, students can be encouraged to provide innovative experimental designs and more exciting and thought-provoking presentations in the classroom.

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AN IMPROVED TECHNIQUE FOR PROJECTING IMAGES OF LIVING AQUATIC INSECTS[1]

Clifford O. Berg

Department of Entomology, Cornell University
Ithaca, New York 14853

ABSTRACT

Two kinds of small aquaria are described that can be inserted into a 2 in x 2 in slide projector in order to project the images of living insects and other freshwater invertebrates. A framework of mirrors that rectifies images from the aquaria also is described. The apparatus can be used in the classroom, for example, to demonstrate respiratory, locomotory and feeding movements as well as differences in morphological features among species.

My previous paper on projecting the images of living insects and other freshwater invertebrates (Berg 1948) includes instructions for making small aquaria that can be inserted into a standard lantern slide projector in place of 2 in x 2 in slides. It also addresses the problem that the aquaria cannot be inverted, as slides always are, to make images appear right side up on the screen. It illustrates a method of rectifying inverted images "by pointing the projector away from the screen and reflecting the image back over the lantern by means of two good quality, first-surface, plate glass mirrors". This technique had been demonstrated only by the use of hand-held mirrors. I had not designed a supporting framework that would hold the mirrors rigidly and yet permit adjustment of their vertical and horizontal inclinations.

I have recently designed and constructed such a framework. The aquaria have been redesigned to adapt them for use in currently available projectors, and two types of aquarium inserts that appreciably improve visibility have been constructed. This paper includes a brief discussion of the most advantageous uses of these aquaria and the types of invertebrates best displayed in them, followed by descriptions of the redesigned aquaria, aquarium inserts, and framework for supporting the mirrors.

Projection of images of living aquatic insects stimulates student interest by bringing the life we are studying right into the classroom. It disseminates information far more rapidly and certainly than the methods traditionally used in biology laboratories. Students' individual laboratory exercises produce some successes, some failures, and many intermediate results. We cannot assume that every student in the class has watched a hydra ingest a cladoceran, even if considerable laboratory time has been spent on that study. In contrast, this technique, like the showing of a motion picture, provides simultaneous

[1]Paper presented in the symposium on *Innovative Teaching Techniques in Aquatic Entomology* at the Annual Meeting of the Entomological Society of America, Houston, Texas, November 26 - 30, 1978.

viewing for all students in a class, with optimum visibility. Discussions of the observed activities and responses thus acquire understandability and meaning.

In some important respects, good motion pictures would be preferable if they were available. A movie can be shown in any season, and there is no need for a successful collecting trip beforehand. A properly exposed film also shows colors of some animals much better. Images of completely opaque subjects project as dark, colorless silhouettes regardless of their color. However, there are no movies of most species of aquatic insects, and the films available usually do not include either the combination of organisms or all of the activities that one wants to show.

The chances of getting a desired response or activity from living insects in an aquarium can be multiplied by using several individuals. If an aquarium is stocked with 5 or 6 damsel fly naiads before mosquito larvae are dropped into it, the lightning-fast action of odonate prey capture will almost certainly be demonstrated in a few seconds. Several individuals usually prove more satisfactory than one, even when no dramatic action is involved. Because each usually positions itself somewhat differently, the group presents a clearer and more complete concept of the structure and posture of a species, and of its respiratory, locomotory, and feeding movements. Displays that focus the viewers' attention on contrasts can be used to strengthen this concept. An aquarium containing mayfly naiads of 3 or 4 families permits contrasts as well as comparisons (especially of gill structure and movements) and thus provides opportunities for quick family recognition. When displaying limnephiloid caddis larvae, I usually stock the aquarium with 3 or 4 larvae, then add 1 or 2 larvae removed from their cases. The naked larvae provide excellent visibility of gills, appendages, and other morphological details, and their efforts to get into a larval case often result in spirited contests for possession.

Insects that display normal activities even when confined are especially desirable subjects. Many chironomid larvae will take up residence in pieces of capillary glass tubing (Fig.1), which can be placed to hold the larvae in the field and in focus. Their elaborate cycle of feeding motions (spinning tiny plankton nets, pumping currents through them, devouring the nets and their contents, and constructing new nets) then projects clearly and convincingly.

Relationships best observed in lateral view usually are seen more clearly by this technique than by viewing living organisms through a dissecting microscope. The gross morphological differences between larvae of *Culex*, *Aedes*, and *Anopheles*, and differences in the ways they and other pond insects orient to the surface film, project very well. Dorso-ventrally flattened animals usually are not so well observed by this technique, although some frequently position themselves on one of the aquarium inserts so as to cast images comparable to a dorsal view. Many benthic species remain on bottoms of the miniature aquaria and clump so closely together that even their lateral silhouettes are obscured.

Most inhabitants of stream headwaters are unsatisfactory subjects because they display more responses to unnatural stresses than natural activities. I have tried to show them in refrigerated water saturated with oxygen, but visibility is obscured by the accumulation of oxygen bubbles on the sides of the aquarium even before it is exposed to the heat of the projector.

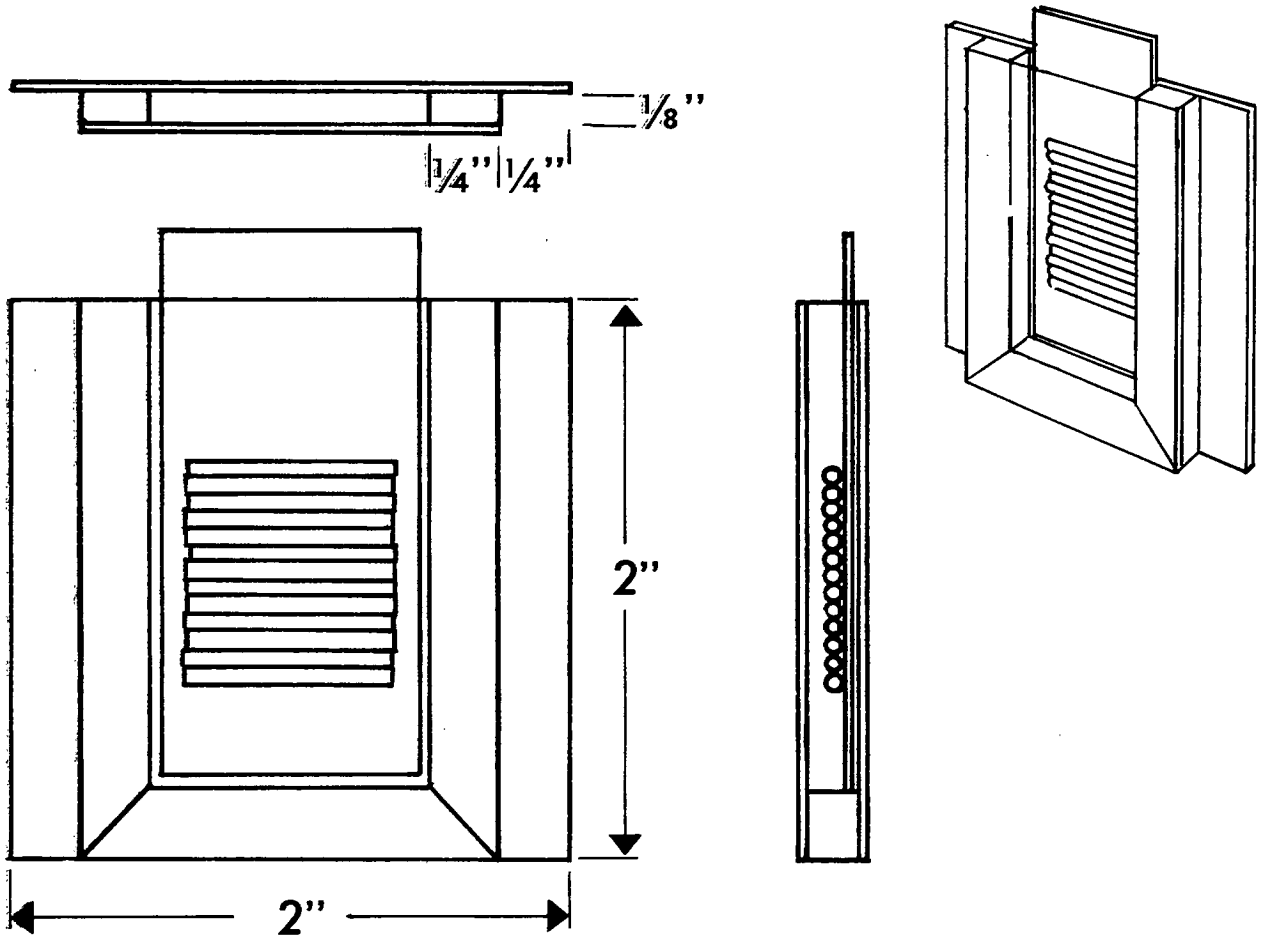

Fig. 1. Thin aquarium, with insert of capillary tubes cemented to piece of slide cover, for displaying activities of chironomid larvae.

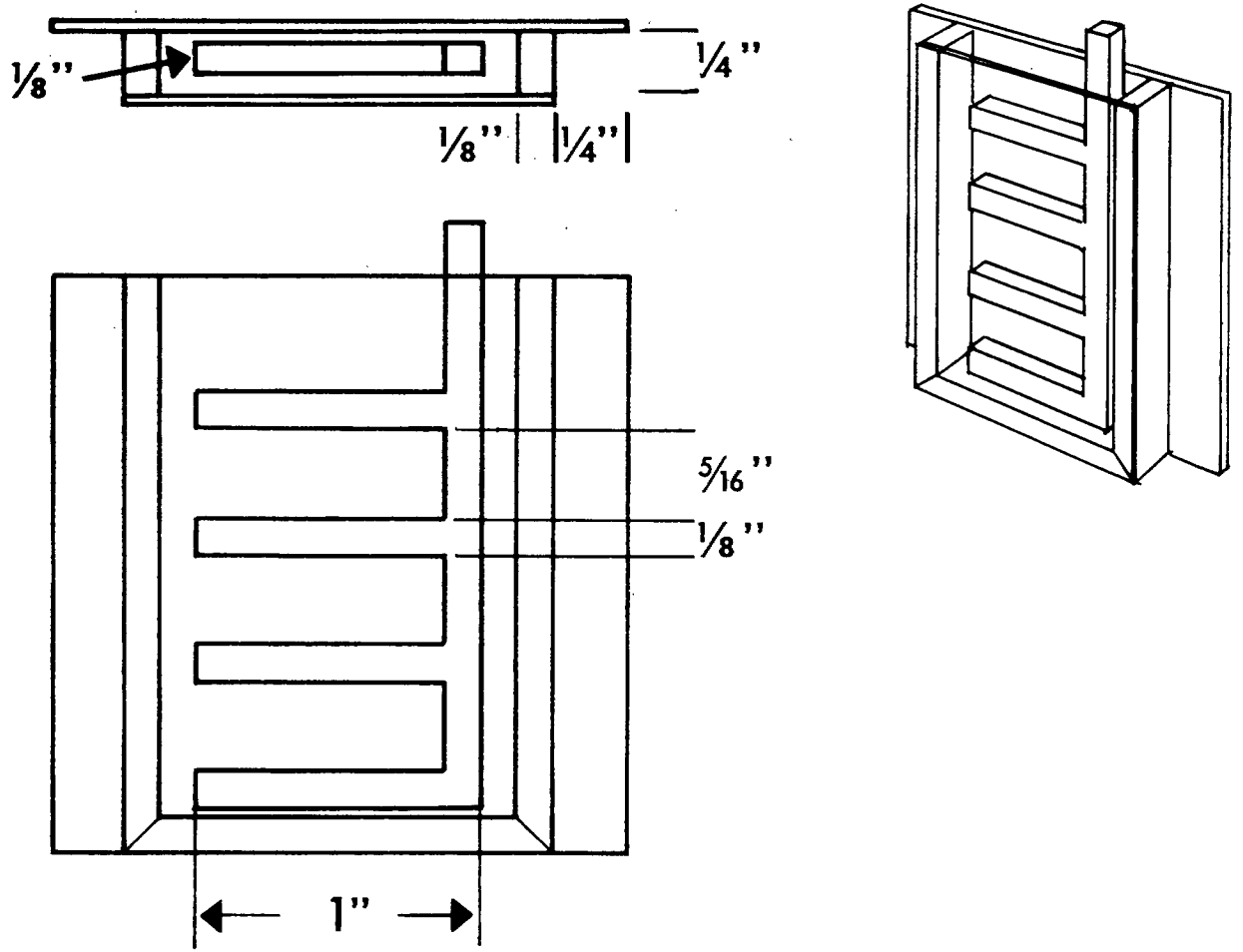

Fig. 2. Thick aquarium, with insert of 1/8 in Plexiglas, cut to provide resting places at different levels for sedentary pond invertebrates.

PROJECTING LIVING AQUATIC INSECTS

Aquaria already at room temperature are not heated in the projector enough to cause heat shock to most pond insects. A Kodak Carousel® projector operating at moderate light intensity required 3 min (longer than most aquaria would remain in the projector) to heat the water in an aquarium (Fig. 2) from 23.0 to 25.0°C.

More problems are created by high light intensity than by heat. As long as they remain in the light path, Gyrinidae, Dytiscidae, and Corixidae swim too rapidly and constantly to provide good visibility of either their form and structure or their normal orientation to the surface film. Other insects may become immobile as if in shock. The phantom larvae of *Chaoborus*, seemingly ideal subjects because of their transparency, open-water suspension, and feeding activities, maintain natural orientation and activity in aquaria until the projector is turned on. Then they suddenly appear lifeless, sink, and clump abnormally on the bottom. If removed from the light path in a few seconds, most larvae resume normal orientation and movements.

In summary, the best subjects for projection by this technique are pond or lake invertebrates that are relatively sedentary, have translucent or semitransparent bodies, are observed to good advantage in lateral view, and do not react too abnormally to brilliant light.

Both types of aquarium shown here (Figs. 1, 2) can be inserted manually, directly into a Kodak Carousel projector. A carousel can be modified to accommodate both by partial removal of septa separating the slide slots. The aquaria are made from 2 glass covers for 2 in x 2 in (50.8 mm) slides and 3 strips of Plexiglas® , Lucite® , or window glass 1/8 in (3.2 mm) thick and 1/4 in (6.3 mm) wide. These strips are laid flat to make the thin aquarium and set on edge to produce the thicker one. The thinner model (Fig. 1) is structurally stronger, and it confines animals closer to the proper focal plane. The thicker model (Fig. 2) demands constant attention to proper focus, but it accommodates larger animals. If Plexiglas or Lucite is used for the sides and bottom, the mitered corners shown (Figs. 1, 2) are suggested. If only glass is available, the bottom strip can be either extended beneath the side pieces or cut shorter and fitted between them. For both types of aquarium, one of the glass covers must be cut down to 1 1/2 in (38.1 mm) in width.

I have cemented the pieces together only with Canada balsam thinned with xylene. This material flows readily by capillarity, thus spreading throughout the area of contact and sealing off joints that would otherwise leak. Before gluing, I draw the front view of an aquarium (lower left of Fig. 1 or 2), actual size, and lay the 2 in x 2 in glass cover on this pattern. Then I spread balsam on the side and bottom strips on the edges to be cemented to that cover and lay them on the cover in the positions indicated. The 2 bottom corners and all surfaces of contact with the glass cover should then be checked to make sure they are completely sealed. Air pockets can be filled by applying a small droplet of xylene followed by balsam. However, the presence of air pockets indicates that the balsam needs thinning. Finally, the uppermost surfaces of the bottom and side strips are covered with a thin layer of balsam and the glass cover cut to a width of 1 1/2 in (38.1 mm) is gently lowered onto them. I have dried each new aquarium at 45 - 50°C for at least 24 h, and then held it at room temperature for 2 or 3 days before filling it with water.

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The aquarium insert shown in Fig. 1 is a strip of glass cover 2 in x 3/4 in (50.8 x 19.0 mm) with pieces of capillary tubing (glass melting point tubes) cemented to it. If this insert is laid flat in a shallow dish of water, and larvae of various genera of Chironominae are removed from their tubes and dropped in, many will move into these tubes. After a few hours or perhaps overnight, the insert can be placed in a small aquarium, and respiratory and feeding movements of the larvae can be projected effectively. Similar inserts can be made with glass tubing of larger diameters for display of caddis larvae and aquatic Lepidoptera larvae.

The insert shown in Fig. 2 provides "perches" at various levels for caddis larvae, mayfly nymphs, and damselfly nymphs, to avoid clumping at the bottom. It is made from a 1 in x 2 in (25.4 x 50.8 mm) piece of 1/8 in (3.2 mm) Plexiglas cut to create the structure illustrated. Plexiglas inserts provide much better visibility than any structure made of opaque materials.

The mirrors used to rectify images must be made of quality plate glass, silvered on the front side. First-surface mirrors must be handled with great care; they are easily scratched and fingerprinted. They should be cleaned only with a non-abrasive glass cleaner such as Windex® , applied with soft cloth or facial tissue. The lower mirror is 4 in x 5 1/2 in (10.2 x 14 cm); the upper, 6 in x 6 in (15.2 x 15.2 cm). A 3 in length of piano hinge is glued to the back of the lower mirror along its short upper edge. A 5 in length of piano hinge is glued along the upper edge of the larger mirror. A piece of sheet metal glued to the back of the larger mirror extends 1 in beyond its lower edge, equidistant from the two sides. It has a 3/16 in hole near its free edge to adjust mirror inclination.

The framework to hold the mirrors (Figs. 3, 4) is built on a 6 in x 9 in ringstand base. A 1/2 in hole is drilled near each corner, with centers 3 3/4 in apart on the short side and 7 1/4 in apart on the long side. The upright rods from this and 3 other ringstands are cut to a length of 15 1/2 in, and threaded (1 in long) at their lower ends. These rods are secured firmly in the 4 corner holes by turning a hexagonal nut as far as possible onto each, inserting the threaded end through the hole, and tightening a second nut onto each from the lower side of the ringstand base. To obtain tripod support and adjustable horizontal inclination, a bolt (1/2 in x 1 1/2 in) is screwed into the threaded hole originally provided for the upright rod of the ringstand (Fig. 4), and the 2 corner feet at that end of the ringstand base are ground away. This bolt is screwed through the ringstand base just far enough to level it.

Three horizontal rods for suspending the mirrors, each 8 1/2 in long, can be obtained from the ringstand rods when the upright corner rods are cut. The lengths of piano hinge attached to the mirrors are glued to two of these rods, allowing equal lengths of rod to extend at each end of each hinge. These rods are attached to the vertical rods with clamp holders whose thumbscrews have been replaced with short bolts. The rod suspending the lower mirror is attached to the vertical rods farthest from the projector, 4 1/2 in above the ringstand base. For adjustment of its inclination, an L-shaped, 1 in x 3 in piece of sheet metal is glued to the ringstand base. A 3/16 in hole drilled through the end that projects upward has a nut glued over it, and a screw (1/8 in x 2 1/2 in) passing through that nut pushes against the back of the mirror, tilting it more horizontally when it is tightened (Fig. 4).

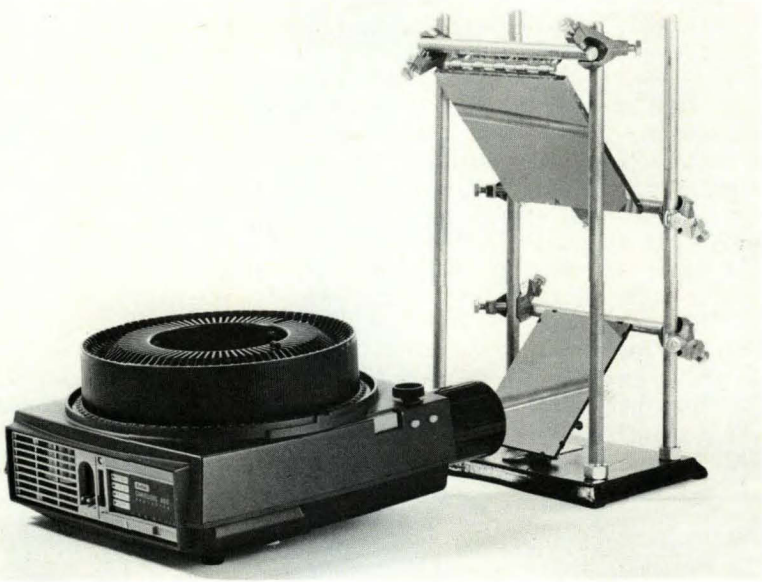

Fig. 3. Projector and framework for mirrors to rectify inverted images (piano hinge suspending top mirror clearly visible).

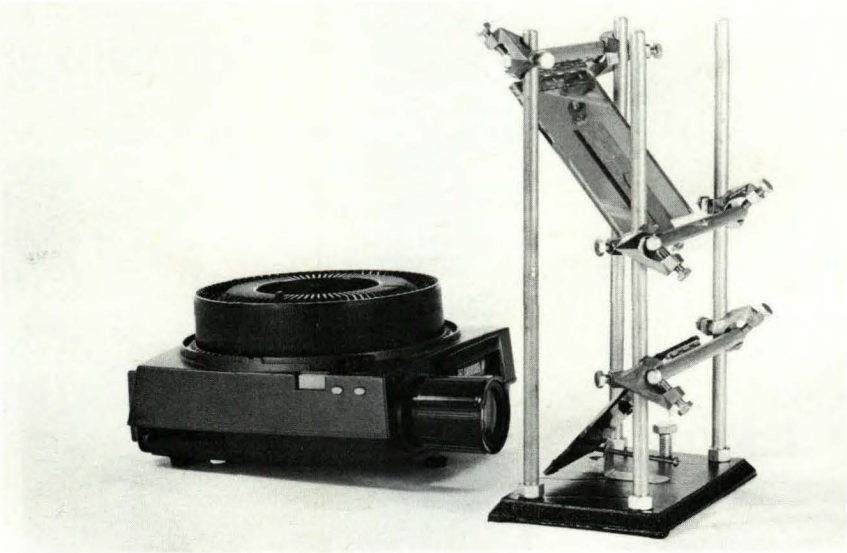

Fig. 4. Projector and framework for mirrors, showing bolt to adjust level and screws to adjust inclination of mirrors.

The adjustment screws illustrated (Figs. 3, 4) should be replaced by screws with knurled heads, eliminating the need for a screwdriver and the attendant risk of scratching a mirror.

The rod suspending the upper mirror is attached to the vertical rods nearest the projector, 13 in above the ringstand base. A hole large enough for a loosely fitting 1/8 in screw (2 1/2 in long) is drilled through the middle of the third horizontal rod. This rod is attached to the corner rods farthest from the projector 8 in above the ringstand base. The screw that goes through it passes through the hole in the free edge of the sheet metal glued to the back of the upper mirror and into a nut glued over that hole. When this screw is tightened, it pulls the mirror back into a more nearly horizontal position.

Inclination of the lower mirror is first adjusted to direct the whole image onto the upper mirror. Then elevation of the image on the screen is adjusted by inclination of the upper mirror. Images are centered on the screen by a slight clockwise or counterclockwise turn of the entire apparatus.

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BRINGING LIVE INSECTS INTO THE CLASSROOM[1]

N.H. Anderson

Department of Entomology, Oregon State University
Corvallis, Oregon 97331

ABSTRACT

Four laboratory demonstrations or exercises are described which expose students to the activities of insects: (1) functioning of the physical gill, using corixids or notonectids; (2) physical properties of the surface film of water, using the staphylinid beetles *Dianous* and *Stenus;* (3) molting, using a large laboratory culture of, for example, craneflies or dragonflies; and (4) behavior and feeding of the larval limnephilid *Clistoronia magnifica* (Banks).

INTRODUCTION

I give a 10 wk upper division course in aquatic entomology. It is primarily a service course for students in fisheries but also includes graduates and undergraduates in other biological sciences, naturalists, and even fly fishermen. The subject matter is not conceptually difficult but, because of the wide scope of the field, the limited entomological background of the students, and the small number of contact hours in an academic quarter (lecture - 18 h, lab - 36 h), it is a challenging course. In this paper, I give examples of fieldwork and lab exercises or demonstrations that have kindled further interest or curiosity in students so that they pursue aquatic entomology on their own.

I believe that information, facts, or ideas that students obtain by their own initiative are retained longer than is lecture or laboratory material. Thus, *learning* rather than teaching is important and we should provide stimuli for learning. Futhermore, an instructor does not have to be *innovative* to be successful because the subject matter of aquatic entomology is inherently interesting to those who have some biological curiosity. So, I do not feel obliged to be an entertainer, replete with gimmicks, gadgetry, new-fangled audiovisual hardware, or programmed learning kits.

COURSE STRUCTURE

A: Field work

Field experiences are important because they provide a stimulus for learning. There is much to be gained by observations of actions and interactions of aquatic insects in their natural surroundings.

We provide standard aquatic collecting nets but often I feel that this is a mistake because collecting by net obscures more than it reveals about an

[1]Paper presented in the symposium on *Innovative Teaching Approaches in Benthic Sciences* at the 26th Annual Meeting of the North American Benthological Society Winnipeg, Canada, May 10 - 12, 1978.

insect's life history. Arming students with nothing more than a tea strainer or a guppy net would certainly encourage a closer look at micro-habitats in ponds and streams.

An insect collection comprises a major component of the grade in my course. I also require (and grade) a field log book to accompany the collection. It includes microhabitat data and behavioral observations to prevent the exercise from becoming just entomological "stamp collecting".

B: Laboratory aspects

An understanding of taxonomy or classification is necessary for discussing physiological, ecological or behavioral adaptations to the aquatic environment. Lectures are the most expedient method of presenting this information but such lectures are boring if filled with scientific or common names of organisms of which the students do not have a mental image. Thus, recognition, or identification (at least to the family level) is the first step in learning about aquatic insects. The continuous keying of preserved specimens in laboratories is somewhat tedious but can be enlivened by including activities using living insects. For example:

1. Functioning of the physical gill (after Popham 1954). --The exercise compares survival of corixids or notonectids in 2 flasks of water, one nitrogen-saturated, the other oxygen-saturated and both having air in the top of the flask displaced by an atmosphere of nitrogen gas.

The bugs in the nitrogen-saturated water return to the surface quickly to renew their bubble, then dive and return to the surface for a second or third time. After 5 - 10 min they are all floating motionless at the surface and eventually die from lack of oxygen. With oxygen-saturated water, the insects remain at the bottom for some time before surfacing for more gas. Though they can obtain only pure nitrogen, they swim to the bottom and behave normally for the ½ - 1 h duration of the experiment.

The exercise demonstrates that dissolved oxygen can diffuse into the nitrogen bubble and then into the tracheal system. Many variations, including temperature comparisons, timing the periods of submergence, and preventing surfacing with a layer of oil, have been added at the suggestion of students.

2. Physical properties of the surface film of water (Hynes 1970). --The staphylinid beetle, *Dianous*, and a related genus, *Stenus*, can traverse the water surface at 60 - 70 cm sec^{-1} for several meters. For *Dianous*, this speed is many times faster than it can run on land. The movement is caused by a surface-tension-lowering secretion exuded from glands at the tip of the abdomen. By watching a *Dianous* adult swim to safety, comments about physical properties of the surface film gain meaning. I suggest that *Dianous* races would bring out the best of ingenuity and enterprise as a classroom exercise!

3. Molting. --Molting to the adult is an aspect of the life cycle that is not commonly observed in the field or laboratory. However, with a large laboratory culture available, and considerable luck, occasionally a dragonfly or cranefly will emerge at the appropriate time for a laboratory demonstration.

LIVE INSECTS IN THE CLASSROOM

Observations of molting, of the wings expanding and hardening, of the wings folded above the body before hardening and then extended laterally, and, especially, the ectodermal lining being pulled out of the tracheae are remembered if observed but have little impact when described in a lecture.

4. Behavior and feeding (Anderson 1978). --I have available a continuous culture of the limnephilid caddisfly, *Clistoronia magnifica* (Banks). For behavior and feeding exercises, I provide the students with 3 or 4 larvae and ask the unstructured question: "What can you learn about these caddis?" (e.g. What happens if the larvae are pulled out of their cases? What if part of the case is broken off? What will they eat? What will they use for case material?). This evokes a range of responses from "nothing of significance" to many interesting observations (e.g. novel ideas for potential case materials and foods). A myriad of unorthodox approaches are used to answer the questions.

To conclude, the most important aspect of lab exercises is exposure to the activities of living insects. Behavior of insects in the laboratory may be atypical of that in their natural habitat, but student observations and experiments lead to questions, to a heightened interest, and to *learning*. These desirable attributes are obtained without elaborate equipment.

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UTILIZATION OF STUDY STREAMS[1], [2]

David S. White

School of Natural Resources and Great Lakes Research Division
University of Michigan
Ann Arbor, Michigan 48109

ABSTRACT

The annual University of Michigan Aquatic Entomology field trip to the Pigeon River area, an innovation begun by Justin Leonard in 1965, is described. Data accumulated from previous trips (e.g. taxa which have been consistently collected by classes over the past 13 yr) help prepare the student for the trip and are used in year-to-year comparisons. Similar study sites nearer Ann Arbor are used to familiarize students before the Pigeon River trip. Guidelines to aid in selecting streams for aquatic entomology field trips are presented.

Aquatic Entomology has been offered at the University of Michigan, School of Natural Resources, yearly since 1965. The course was originated and taught by Justin (Doc) Leonard until his death in 1975. Leonard was a well-known aquatic entomologist not only for his teaching, but for his many publications, books, and radio and television appearances (Ross 1976; Gloyd 1976). His excellence in teaching is reflected in the number of students he influenced and the new text *Aquatic Insects of North America* (Merritt and Cummins 1978) which was dedicated to him.

Through a long association with entomology, aquatic ecology and fisheries, Leonard brought many innovations to teaching including state-wide species lists, excellent use of photography, and a thorough knowledge of particular stream systems. The latter has provided unique opportunities for demonstrating the ecology and distribution of aquatic insects.

Aquatic Entomology at the University of Michigan was designed by Leonard primarily as a supplement to the Resource Ecology Program with particular emphasis on natural history and ecology. Of equal importance has been the teaching of taxonomy which is supplemented by student collections. To combine various aspects of the course, Leonard made extensive use of the Pigeon River area, located in the north-central portion of the lower peninsula of Michigan, about 200 mi (320 km) from Ann Arbor (Fig. 1). The area consists of a variety of streams, rivers, and small lakes located in three state forests. The Black, Pigeon, and Sturgeon Rivers in the Pigeon River State Forest, Hunt Creek and

[1]Paper presented in the symposium on *Innovative Teaching Techniques in Aquatic Entomology* at the Annual Meeting of the Entomological Society of America, Houston, Texas, November 26 - 30, 1978.

[2]Contribution No. 252 from the Great Lakes Research Division, University of Michigan, Ann Arbor, Michigan 48109.

the Hunt Creek Experiment Station in the Thunder Bay State Forest, and the Au Sable River and Big Creek in the Au Sable State Forest are of particular interest.

The rivers, streams, and lakes of the state forests are controlled and managed by the Michigan Department of Natural Resources. They have been the sites of thesis research, utilized by classes from various universities in the state, as well as the subject of controlled research and manipulation by the Department of Natural Resources over the past several decades. Leonard was Director of the Hunt Creek Experiment Station until 1942, many years before joining the faculty at Michigan. Using his knowledge of the area, combined with reports from other investigations, Leonard began an annual, 2 - 3 day, Aquatic Entomology field trip to the Pigeon River area. The trip, started in 1965, has been continued to the present because of the opportunity given the students to examine a wide variety of aquatic insects in natural habitats. The instructor is able to discuss what will be seen before a trip, rather than after, because of the previously compiled information.

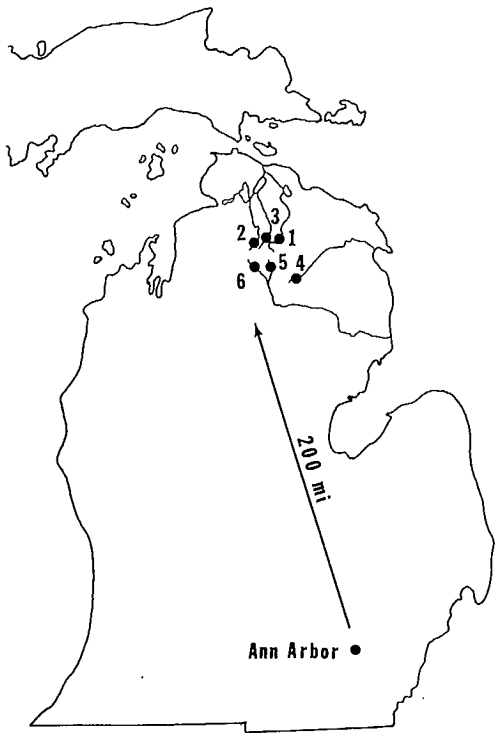

Fig. 1. The lower peninsula of Michigan showing approximate locations of study streams in the Pigeon River area: 1, Black River; 2, Sturgeon River; 3, Pigeon River; 4, Hunt Creek; 5, Big Creek; and 6, North Branch of Au Sable River.

STUDY STREAMS

Aquatic Entomology is offered in the winter term because, in Michigan, streams are most stable in late winter before the first spring thaws. The Pigeon River field trip is held near the first of April allowing observation and collection of the greatest diversity of aquatic insects. At that time, some of the ice and snow is gone from the streams and lakes, and students are reasonably assured of seeing and collecting certain taxa, several of which either are not found in the Ann Arbor area or are difficult to collect locally. Table 1 gives examples of taxa which consistently have been collected over the past 13 yr. The sites listed represent small lakes and first to fifth order streams having a varity of habitat types including extensive areas of leaf litter, shifting sand, gravel and large rubble riffles.

Each type of habitat is discussed in light of the species which will be present and their relations to substrate, flow, depth, and other physical factors before the students start collecting. Students then remove portions of the substrate and examine the habitats, habits, and location of the various taxa. Finally, working in groups of two or more, qualitative and quantitative samples are collected and preserved for analysis in the laboratory. Each student is required to record all observations in a field notebook. The student prepares a short paper which, for each site, includes a description of the habitat, a list of taxa and their relative abundances, and notes on ecology using the field data and information from the samples analyzed in the laboratory. Comparisons then are made between the various sites and their respective faunas. In past years, comparisons have been purely descriptive; however, we are now including some mathematical analyses such as various diversity and biotic indexes (e.g. Hilsenhoff 1977). Copies of papers from past years allow the student to make comparisons between their data and what has been collected previously at the same sites.

The trip also provides a basis for the required taxonomic collection and gives the student an eagerly sought intensive period of study away from normal distractions.

Similar study sites have been established near Ann Arbor. These sites are used primarily for supplementing the taxonomic collection and are sampled early in the semester. The local trips allow students to become familiar with collecting techniques and observation before the extended trip. Species lists are kept for these sites; however, extensive analysis of the data is not required.

The following guides have been developed to aid in selecting particular types of systems for aquatic entomology field trips:

1. Sites should be free of natural and man-made disturbances unless highly polluted waters specifically are sought.

2. Sites should have available species lists to provide comparisons with lists prepared by the students.

3. A wide variety of habitat types should be available for extended trips.

4. Easy access to sites both locally and on the extended trip will allow students to get the most out of the limited collecting time usually available.

Table 1. Selected taxa to be collected at study streams.

SITE	EPHEMEROPTERA	PLECOPTERA	TRICHOPTERA	COLEOPTERA	DIPTERA
Black River			*Micrasema*		
Sturgeon River	*Siphloplectron*				*Atherix*
Pigeon River	*Ephemerella*	*Phasganophora* *Cultus* *Soyedina* *Allocapnia*	*Glossosoma* *Brachycentrus* Hydroptilidae *Frenesia* *Lepidostoma*		
Hunt Creek		*Amphinemoura* *Isogenoides* *Paracapnia*	*Parapsyche* *Psychoglypha* *Wormaldia* *Dolophilodes* *Hydatophylax* *Hesperophylax*	*Stenelmis* *Dubiraphia* *Optioservus*	*Epoicocladius*
Big Creek	*Litobrancha* *Baetis*	*Paragnetina* *Isoperla* *Alloperla*	*Ptilostomis*		
North Branch Au Sable R.	*Baetisca* *Isonychia*	*Pteronarcys* *Acroneuria*			
Hardwood Lake			Phryganeidae		
Tee Lake			*Molanna*	*Donacia* *Omophron*	

STUDY STREAMS

5. Sites should be visited when conditions are the most stable and are most similar from year to year.

The nature of the study sites chosen will depend on the goals of the particular course and instructor and the time of year; however, a well-conceived program for the utilization of study sites will provide a base and continuity for a course in aquatic entomology. Justin Leonard was innovative in selecting and utilizing study sites, particularly streams. The sites have proven valuable for student collections and for observing a wide variety of aquatic insects.

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WINTER FIELD WORK[1]

Rosemary J. Mackay

Department of Zoology, University of Toronto
Toronto, Ontario M5S 1A1

ABSTRACT

Winter field work is described for students specializing in freshwater biology and aquatic entomology at the University of Toronto. Exercises include observation of growth, feeding, processing of autumn-shed leaves, case-building, and migrations within a stream by members of winter communities. Logistical problems such as locating suitable stream sites, optimal numbers of students, adequate winter dress and food, and efficient sampling are discussed.

Considering how often we use the word "winter" when describing seasonal events in freshwater habitats, it is a remarkably imprecise term. In the more southern United States, winter field work is hardly innovative - it is done as a matter of course. But in the northern States and Canada, winter is more than just the coldest season. Field work from November through March is often extremely uncomfortabe because of cold temperatures, snow, and ice.

In Canada, the academic year in many universities runs from mid-September to the beginning of April. Courses typically are 24 wk long with one laboratory period per week. How can we teach practical field ecology when mild weather will be encountered for only 6 - 8 wk?

At the University of Toronto, two courses involve field work on stream-living invertebrates. The introductory animal ecology course concentrates all field work into the mild period, using the practical example of a stream ecosystem to illustrate some of the general features of ecosystems. While the stream ecosystem is atypical in that it receives allochthonous inputs and also exports energy and materials, I find that the exceptions serve to emphasize the general features of more characteristic ecosystems. With an enrolment of up to 150 students, logistics are complicated; but by taking 30 - 40 students at a time, we ensure that each student samples the headwaters (2nd order), midregion (3rd order) and mouth (4th order, at Lake Ontario). The advantages of running waters over other ecosystems are as follows:

1. An aquatic animal community in late September and October is a dynamic and diverse natural system; most terrestrial communities at this time are becoming inactive.

2. Macroinvertebrate communities in running water are more diverse than those in lakes, and a river bottom offers more spatial heterogeneity than that of a

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pond. Substrate differences plus current velocity, depth, and distance from stream source are all easily measured factors. A stream or small river is also more suitable than a pond because sampling can be spread over a considerable distance and thus sampling impact is minimized.

3. In a large city (Toronto is 80 km wide)the only other active autumn community - that of forest litter - is rare.

Unfortunately, many interesting features of the ecosystem are neither apparent in autumn, nor easily perceived by a large introductory class. For example, although the three stations (each separated by 8 - 12 km) do have markedly different fish and invertebrate communities, differences in temperature are no longer as obvious as they were in summer when the distinctive species composition of the headwater community could be correlated with summer-cool conditions. However, it is possible to point out that the water temperature is relatively warm when compared to air temperatures in autumn which introduces the idea of a potentially active aquatic community.

At this intermediate season, some vegetation is beginning to die back, but there is still a general greenness of rocks and banks. Primary production may still be important. However, the dead leaves contributed by autumnal leaf fall are still largely undecomposed; if they are noticed at all, it is during their export downstream rather than while accumulating in leaf packs and deposits. Most of the detritivores whose life histories are synchronized with this autumn influx of food are still too small to be detected by our inexperienced students and the class is too large for the few experts among our teaching assistants to spend much time pointing out these inconspicuous invertebrates. In this introductory course we therefore have to concentrate on the general resources of the running water ecosystem and the adaptations that invertebrates have evolved to exploit these resources.

For students who are specializing in freshwater biology and aquatic entomology we offer a more advanced course where the class is exposed to the challenge of real winter field work. Many of the events and processes which occur between October and March are of vital importance to the stream ecosystem and include some of the more interesting features of invertebrate life history and ecology.

I have done a lot of winter work myself and have frequently heard the surprised remark: "But surely the stream is frozen?" Third or 4th order streams may indeed have a complete covering of thick ice and be almost impossible to sample unless one leaves artifical samplers *in situ* for later retrieval. But in Quebec and Ontario, the small woodland streams (1st or 2nd order) remain open over much of their length (Fig. 1) and provide a wealth of information for an advanced class. Even 3rd order streams may not freeze from one bank to the other in a mild winter; any ice on slow-flowing marginal water can be carefully lifted up and floated downstream (Fig. 2).

The most exciting feature of the winter stream, in my opinion, is its dynamic nature. Leaves are decomposing, invertebrates are feeding, and caddis larvae can be seen cutting out discs from leaves to make cases. There are no signs of hibernation and, if samples are taken at monthly intervals, many species can be seen to have their major growth periods at this time.

WINTER FIELD WORK

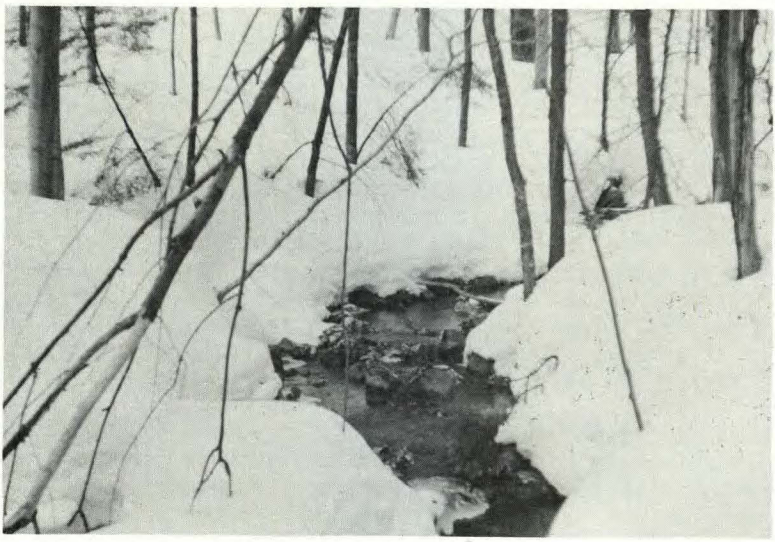

Fig. 1. A woodland stream near Montreal, Quebec, in early March.

Fig. 2. Removing ice cover before sampling the benthos, at the margin of a 3rd-order stream in Ontario.

Of course, all these features can be seen in spring and summer too, but the uninitiated student doesn't *expect* this sort of activity in winter; therefore it is a stimulating experience.

Energy in woodland streams flows mainly through heterotrophic organisms. This is a key feature of the stream ecosystem and one which we all emphasize. However, the impact of this concept on a student will be far greater if he can follow the processing of autumn-shed leaves by the winter community. Because green algae and other green plants are often absent during winter, the importance of coarse particulate organic matter as food material is more obvious. Much can be learned about the food values of different leaves by observing the type and extent of decomposition over time (see Merritt et al. 1979). These observations can be extended to include studies of case-building by caddisflies, many of which cut discs from tough leaves such as beech or oak in the autumn but change to sand or bark in late winter when these leaves are beginning to decay (Mackay 1972; Mackay and Kalff 1973).

Several species of stoneflies and caddisflies make good subjects for a study of growth rates. Because temperatures are cool, growth rates are slow enough to be followed by monthly sampling without the risk of missing an instar or group of instars, as can happen in summer. Even winter stoneflies do not grow rapidly until February or March. The usual method of studying rates of growth is to measure the widths of head capsules in the population, and then to observe proportions of different instars or age classes at intervals (e.g. Harper 1973 a, b; Mackay 1969, 1972). In stoneflies, length measurements of the mesothorax, metathorax or wing pads provide a more obvious indication of approaching emergence. The sight of emerging stoneflies crawling over the snow on a sunny February day is a dramatic, never-to-be-forgotten experience. Emergence can also be anticipated as stonefly larvae become more concentrated in marginal areas of the stream. Samples taken from central and marginal areas during the preceding months reveal such a trend (Table 1). *Pycnopsyche lepida* (Hagen) migrations may be observed in the opposite direction (Table 2). Larvae occur in organic cases among detritus at the margins in late fall but during the winter there will be increasing proportions of older larvae in central regions where the case is changed to one of sand grains.

Most students are surprised that such obvious changes in size and distribution can occur when the water temperature remains just a few degrees above freezing point. Again one should emphasize that habitat temperatures are warmer and food resources probably more available here than they would be in a nearby terrestrial habitat at this season.

Winter field work should never be attempted with a large group; 20 students is the maximum number that can be supervised effectively by one instructor, and best results will be obtained by 10 or less. To remind students to dress warmly will not guarantee suitable clothes especially in the case of students from other countries. Warm underwear and several layers of wool shirts or thin sweaters should be specified. Rubber gloves with cotton liners are essential; I encourage rubber gloves even on fall field trips. Students should have windproof jackets with down vests too, if possible. A spare set of clothes is essential in case of emergency. Warm mitts are necessary for work out of the water.

WINTER FIELD WORK

Table 1. Distribution of stonefly larvae over a fall-winter sampling period in a 3rd-order stream.

	Numbers (%) per habitat per collection date	
Date	Marginal detritus and gravel	Central gravel and pebbles
1 Nov.	5	95
1 Dec.	2	98
3 Jan.	10	90
3 Feb.	80	20
2 Mar.	100	-

Table 2. Distribution of caddisfly larvae over a fall-winter sampling period in a 3rd-order stream.

	Numbers (%) per habitat per collection date	
Date	Marginal organic detritus	Central gravel and pebbles
1 Nov.	100	-
1 Dec.	100	-
3 Jan.	80	20
3 Feb.	60	40
2 Mar.	40	60

ROSEMARY J. MACKAY

Students from urban areas do not realize that air temperatures a few miles out in the country will be considerably cooler than in the city. Nor do they realize how quickly they will feel cold while standing around the sampling site. Hot drinks at hand and high energy snacks in the pocket are invaluable for maintaining energy and good spirits.

For quick processing of samples and to minimize ice formation on containers, I suggest that nets be emptied into deep dish-washing basins rather than shallow trays, coloured basins being less likely to be left behind in the snow. Damp gravel and organic detritus should be transferred to large plastic bags, and quickly stored in a pack sack or vehicle near (but above) 0^0C.

Because the vehicles in which students travel are the most reliable quick source of warmth, a stream should be selected which is accessible by car. If snow shoes have to be used, everyone should be familiar with the harness before leaving room temperature; straps and buckles are much harder to manage when fingers are cold. Snow shoes can easily be fitted to waders. Do not emphasize the unpleasantness of cold weather work. Treat the conditions as just another set of factors for which special attitudes, equipment, and preparations are necessary, as for any field exercise.

Once students have experienced this kind of field work they will have a far deeper understanding of how streams and their communities function as well as a feeling of having achieved an unusual and often exciting piece of work.

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ASSOCIATION ANALYSIS, SPECIES INTERACTIONS, AND THE STRUCTURE OF BENTHIC INVERTEBRATE COMMUNITIES

David D. Hart
W.K. Kellogg Biological Station
Michigan State University
Hickory Corners, Michigan 49060

ABSTRACT

Interactions between species contribute to the structure of ecological communities. A preliminary assessment of the importance of these interactions is achieved by quantifying patterns of species association. Methods are presented for using both presence-absence data and population abundance data to describe co-occurrence patterns for species pairs. The statistical analysis of these patterns receives particular attention. Significant associations may result from either active ecological interactions or habitat heterogeneity. Distinguishing between these alternative biological explanations will frequently require the use of supplementary, experimental methods. Taken together, these descriptive and experimental approaches can provide a powerful analytical method for determining the role ecological interactions play in structuring benthic invertebrate communities.

INTRODUCTION

An ecological community is composed of a set of species that co-occur in a given habitat. One characteristic of these communities is their "structure", which Tyler (1974) defines as "the functional relationships within and among species that bring about species population changes" (cf. Pielou 1975). Connell and Slatyer (1977) view community structure similarly, stating that "it is the interactions [among species] that make a community a worthy unit of study." These authors stress the importance of understanding the degree to which the distribution and abundance of a given species is dependent upon the other species with which it co-occurs.

This exercise evaluates the importance of species interactions in structuring benthic invertebrate communities, using methodologies suitable for undergraduate courses in ecology. It begins by briefly presenting methods for collecting data which can be used to describe the spatial co-occurrence patterns of benthic species. Several methods for statistically analyzing these patterns of species co-occurrence are then developed. Particular attention is directed towards the calculation and interpretation of these statistical tests. Associations resulting from active ecological interactions vs. habitat heterogeneity are discussed and the importance of analyzing community structure using both descriptive and experimental approaches is stressed.

DATA COLLECTION

To examine patterns of spatial co-occurrence, it is first necessary to select a habitat and decide upon a sample unit size and sampling procedure. In certain environments, species occupy discrete habitat units (e.g. leaf-mining insects; aquatic invertebrates inhabiting pitcher plants; etc.). The size of

individual sampling units in such environments is dictated by the habitat unit itself. The choice of a sample unit size in less discrete habitats (e.g. many benthic environments) becomes more arbitrary. In streams, individual stones can serve as relatively discrete sampling units (e.g. McGinniss and Trush 1979; Hart 1978; Stout and Vandermeer 1975), as can decomposing leaf packs (e.g. Merritt et al. 1979; Reice 1977). Alternatively, the sample unit size can be arbitrarily defined by the choice of sampling equipment (e.g. Surber samplers, bottom grabs, or artificial substrates). However, the co-occurrence pattern generated by such plot sampling is likely to be sensitive to the sample unit size chosen (see p. 67, "Influence of Sample Unit Size").

Having chosen a sample unit size and a habitat to be sampled (e.g. stream riffle; lake littoral zone; etc.), a set of replicate samples of the benthic fauna is collected. The number of samples to be taken and the sampling procedure to be used (e.g. random or stratified) will depend upon the degree of variability across samples relative to the level of precision required to estimate population abundances (Elliott 1977). In this regard, some prior familiarity with the habitat should allow the instructor to estimate the expected range of densities for given taxa. When sampling, construct a map of the habitat (cf. Welch 1948), and record the locale from which each sample is collected. This often proves useful for later interpretation of species distribution patterns.

Once the samples are returned to the lab, the invertebrates should be sorted into the lowest possible taxonomic categories (preferably to species), so that a list of the taxa in each sample can be prepared. If time permits, the number of individuals per species in each sample can also be determined. These data provide either a qualitative list of the species present in each sample or a quantitative record of the abundance patterns of the species in the samples. Patterns of interspecific association can now be analyzed.

DATA ANALYSIS AND INTERPRETATION

Patterns of Presence and Absence

Association can be measured by constructing a 2 x 2 contingency table for presences and absences of any pair of species (cf. Cole 1949). For example, 48 Surber samples were taken from a stream riffle. The association pattern of two perlid stonefly species in these samples is given in Table 1. This table records the number of sample units in which both species are present (a), absent (d), and either *Hesperoperla pacifica* (Banks) occurs alone (b), or *Calineuria californica* (Banks) occurs alone (c). The total number of samples equals n. If these two species are positively associated, most observations should occur in cells a and d. A negative association would produce higher frequencies in cells b and c. A statistical test for association can be made by formulating the following null hypothesis (H_0): the distribution of *Hesperoperla pacifica* is independent of the distribution of *Calineuria californica* in the 48 samples. If H_0 is true, the probability of obtaining a single sample in which the two species occur together should equal:

Table 1. The co-occurrence of two perlid stonefly species in 48 Surber samples collected from a stream riffle. The patterns of presence and absence are illustrated in a 2 x 2 contingency table.

Hesperoperla pacifica (Banks)	*Calineuria californica* (Banks) Present	Absent	Total
Present	24	5	29
	(a)	(b)	(a+b)
Absent	9	10	19
	(c)	(d)	(c+d)
Total	33	15	48
	(a+c)	(b+d)	(n=a+b+c+d)

$$\left[\begin{matrix}\text{Probability of finding} \\ \textit{H. pacifica} \text{ in a sample}\end{matrix}\right] \times \left[\begin{matrix}\text{Probability of finding} \\ \textit{C. californica} \text{ in a sample}\end{matrix}\right] = .42$$

$$(29/48 = .60) \qquad (33/48 = .69)$$

Out of 48 samples, (48 x .42 =) 19.9 samples should be found in which both species are present. In fact, 24 are found, suggesting that the two species co-occur more frequently than expected by chance. The expected frequencies for the other three cells can be similarly determined. Then, a χ^2 goodness of fit test can determine whether the observed values in the four cells differ significantly from those expected under the null hypothesis of independence:

$$\chi^2 = \frac{(ad-bc)^2 n}{(a+b)(c+d)(a+c)(b+d)} = \frac{(240-45)^2 48}{(29)(19)(33)(15)} = 6.69$$

For very small sample sizes ($n < 20$), this equation should include Yates' correction (Sokal and Rohlf 1969). This adjusts for the fact that X^2 is based on discrete data while χ^2 is a continuously distributed variate (cf. Grizzle 1967), where X^2 is approximately distributed as χ^2 with 1 degree of freedom. The greater the deviation between the observed and expected frequencies (i.e. the larger the value of X^2), the greater the likelihood that the null hypothesis is incorrect. A standard decision rule for evaluating H_o states that when X^2 exceeds 3.84, H_o has a very small probability ($P < 0.05$) of being correct. In the above example, $X^2 > 3.84$, so H_o can be rejected at the 5% significance level. The use of a χ^2 test to evaluate 2 x 2 contingency tables requires that no cell have a frequency < 5. If this assumption is not met, Fisher's exact test for independence should be used (Sokal and Rohlf 1969). Cox (1970) and Sokal and Rohlf (1969) provide more detailed discussions of these statistical models and their assumptions.

The X^2 equation can be manipulated to provide an index which discriminates between positive and negative associations, both of which produce high X^2 values. The point correlation coefficient (V) measures this aspect of association:

$$V = \frac{ad-bc}{[(a+b)(c+d)(a+c)(b+d)]^{\frac{1}{2}}}$$

(Note that $|V| = \sqrt{X^2/n}$)

The value of V ranges from +1 (absolute positive association) through 0 (independence) to -1 (absolute negative association) and Pielou (1977) provides an estimate of the sampling variance of V. Thus, two measures of association (X^2 and V) based on presence/absence patterns of species occurrence can now be calculated.

Patterns of Abundance

The qualitative analyses presented above ignore the abundance patterns of species. The presence of 100 individuals of a species in a sample differs markedly from the occurrence of just a few individuals, but the contingency table analysis does not discriminate between these two situations. Spearman's coefficient of rank correlation (r_s) can test for a correlation between the abundance patterns of any given species pair. Its value ranges from a maximum of +1 (direct correlation) to a minimum of -1 (inverse correlation). This statistical test is non-parametric, thus avoiding the assumption that the abundance patterns of species are normally distributed variables. Sokal and Rohlf (1969) and Conover (1971) present examples of the calculation of r_s.

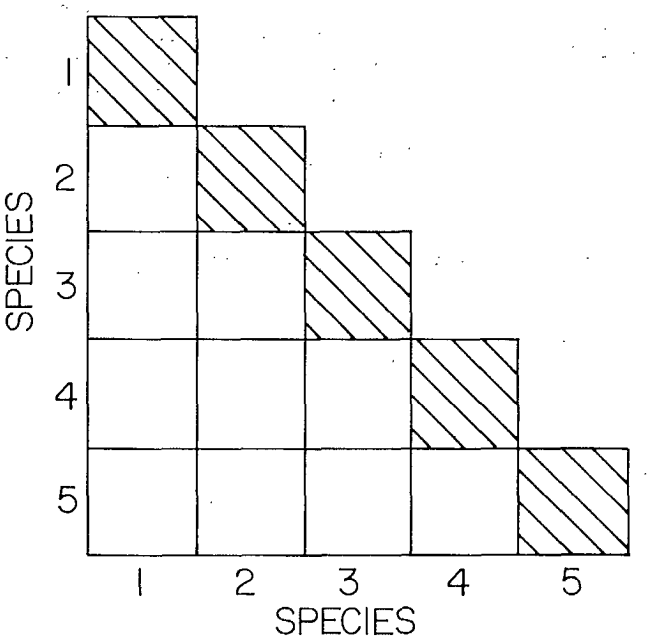

Fig. 1. Hypothetical association matrix for 5 species.

STRUCTURE OF BENTHIC INVERTEBRATE COMMUNITIES

Association Matrices

An association matrix (Fig. 1) displays data from analyses where many species pairs are to be examined. Point correlation coefficients (V) or Spearman's rank correlation coefficients (r_S) can be listed for each unique species combination. The other half of this matrix need not be represented, since $V_{ixj} = V_{jxi}$, and the association of a species with itself (V_{ixi}) equals 1 (shaded elements).

Patterns of association within these matrices must be interpreted with caution. Imagine a 10 species matrix in which values of V are listed for the $\left[\frac{(n)(n-1)}{2} = \frac{(10)(9)}{2} = \right]$ 45 unique species combinations. Next, the X^2 values for some of these species pairs might be calculated, but for which pairs? The results can be easily biased if only some pairs are analyzed relative to the null hypothesis of independence. For example, by choosing only those species pairs with high absolute values of V, the test is biased in favor of rejection. Analyzing only those pairs with values near zero will have the opposite effect, lowering the probability of rejection. Alternatively, the null hypothesis can be tested for all 45 species pairs but the probability of committing a Type I error will be greatly increased (cf. Sokal and Rohlf 1969). Thus, it is necessary to use a lower confidence level in claiming that an entire pattern of association is significant, relative to the confidence level stated for any single, significantly associated pair. For example, consider three species pairs, each of which is positively associated at the 95% confidence level. The probability of correctly rejecting H_o for all three species equals $(0.95)^3 = 0.86$, if the association of each species pair is independent of the other two pairs. Sokal and Rohlf (1969) discuss methods by which the probability levels for a series of hypothesis tests can be adjusted.

Influence of Sample Unit Size

Association analyses may be profoundly influenced by sampling procedures. Consider the hypothetical distribution of two species (W and X) in a heterogeneous habitat (Fig. 2). Using the small sampler in the top section, only one individual of either species could be collected in a single sample. Thus, no samples will contain both species, and the observed frequency in cell a of a 2 x 2 contingency table will equal 0, suggesting a negative association. The larger sampler would either include both species or miss a clump altogether, effectively raising the product of cells a and d, indicating a positive association. The sample unit sizes shown here purposely exaggerate this bias, but more subtle aspects of this methodological problem are relevant to many analyses of association. A number of methods have been proposed to evaluate the effect of sample unit size in quadrat or plot sampling. Grieg-Smith (1964) and Kershaw (1973) advocate the use of grids of contiguous quadrats in order to measure association over a scale of quadrat sizes. Pielou (1977) recommends the use of nearest-neighbor sampling techniques. These plotless methods are likely to be especially useful in the analysis of relatively sedentary benthic invertebrates.

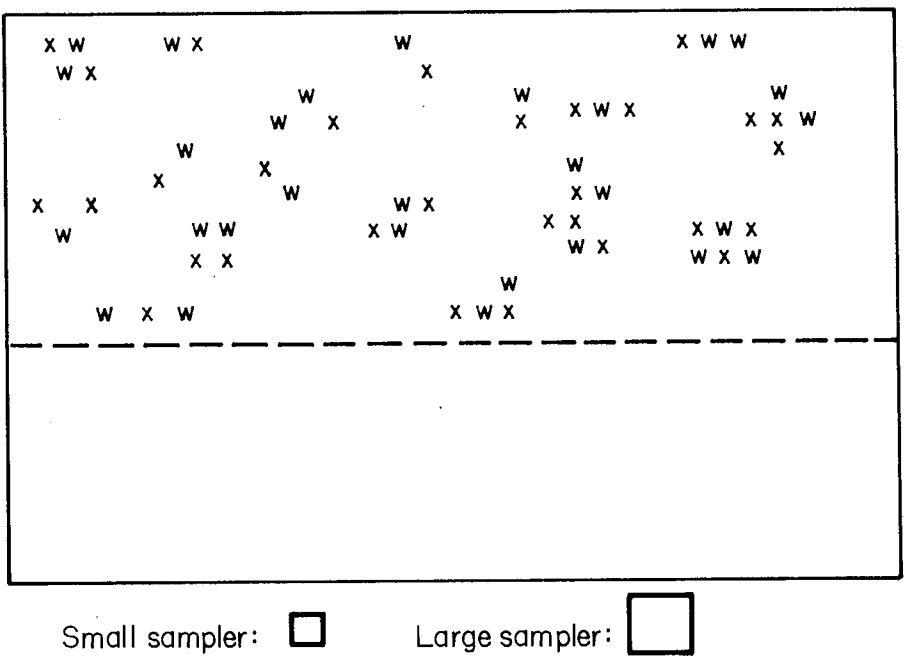

Fig. 2. Distribution of Species W and Species X in a heterogeneous habitat. The two species occur only in that portion of the habitat (rectangle) above the dashed line. Two different sizes of quadrat samplers are used to analyze the pair's association.

Habitat Heterogeneity vs. Ecological Interaction

If the area to be sampled in Fig. 2 includes the section below the dashed line, a large number of samples will be collected in which neither species occurs. This might lead to the conclusion that the species are positively associated, an artifact of enlarging the sampling area. Stratified random sampling (Poole 1974; Elliott 1977) in each "section" of the habitat may sometimes help to decipher the influence of environmental heterogeneity on association patterns. Nevertheless, this raises a critical issue regarding the interpretation of associations as measured by these techniques. If two species are intense competitors which react aggressively to each other's presence, the species should seldom co-occur in a local area. However, co-occurrence would also be minimal if the two species are each adapted to different microhabitats in a patchy environment. In the former case, the species are negatively associated as a result of active ecological interactions, whereas the latter case represents negative association resulting from habitat heterogeneity. A similar problem holds for the interpretation of positive associations. The methods of analysis proposed here largely fail to distinguish between associations resulting from these two factors. Thus, experimental studies must be conducted, both in the lab (e.g. Hildebrand 1974) and the field (e.g. Peckarsky 1979; Connell 1974), in order to identify the causal pathways responsible for the maintenance of particular patterns of interspecific association.

STRUCTURE OF BENTHIC INVERTEBRATE COMMUNITIES

RELATED STUDIES

The data set from this exercise can be analyzed in a variety of ways, serving to demonstrate many ecological concepts. For example:

1. Other aspects of community structure (e.g. richness, evenness, and heterogeneity) can be quantified for each sample unit (cf. Peet 1974).

2. The similarity of different samples (or sites) can be compared, based on the degree to which sites possess a similar composition of species (cf. Hart and Richerson 1977; Sneath and Sokal 1973).

3. If samples are collected on more than one date, temporal as well as spatial patterns of interspecific association can be analyzed (cf. Turkington and Harper 1979).

THOUGHTS FOR DISCUSSION

1. Measures of interspecific association are influenced by biological interactions, habitat heterogeneity, sample unit size, and mathematical characteristics of the particular index used. Thus, association analysis can at best provide only a rough sketch of the degree to which communities are structured by species interactions. Assuming that significantly positive or negative associations are found in your data set, propose a series of follow-up studies (including field experiments) that would allow you to confirm or reject the existence of true biological interactions between species.

2. In ecological communities, many species may be interacting simultaneously via complex feedback networks. This fact suggests that the form of association analysis proposed here (i.e. analyzing species pairs) may give a rather unrealistic view of patterns of species association. Some authors recommend that more complex associations be evaluated using multidimensional contingency tables (e.g. Cole 1957; cf. Fienberg 1970). However, both the derivation of appropriate mathematical models and the interpretation of multi-species association measures can be difficult. Discuss how you would approach the study of complex species interaction networks.

3. Is it possible to predict which species pairs in a community are most likely to be positively or negatively associated? What sorts of information might be useful in making such a prediction? For example, are species that are closely related phylogenetically more likely to compete with each other, and thus be negatively associated, than distantly related species?

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ISLANDS IN THE STREAM: AN ANALYSIS OF THE SPECIES-AREA PATTERNS OF STREAM ROCKS[1]

Matthew J. McGinniss[2] and William J. Trush[3]

Biology Department and Center for Environmental Studies
Virginia Polytechnic Institute and State University
Blacksburg, Virginia 24061

ABSTRACT

The species-area relationship, an aspect of the theory of island biogeography, can be explored in the field and laboratory to help the student associate certain ecological concepts with an observed phenomenon. Procedures are described for selection of the stream and rocks, removing organisms from the rocks, estimating numbers of species, estimating the surface area of rocks, and constructing species-area curves. A simplified key to the orders of major groups of immature aquatic insects is included. Inherent sources of variability (e.g. variable surface texture of rocks, exposure to different physical and biological influences in the stream, sampling variability) are also investigated. Possible expansions of the exercises are presented.

INTRODUCTION

An ecological concept that has attracted recent attention is the theory of island biogeography. Since the early nineteenth century, naturalists have observed that larger islands support more species than smaller islands. MacArthur and Wilson (1967) proposed that the number of species on an island is a dynamic balance between immigration and extinction[4]. Pianka (1974) and Wilson and Bossert (1971) give a general introduction to the theory. We will focus on one of the many aspects of the theory of island biogeography that can be explored in the classroom laboratory, namely, the species-area relationship. We will provide an outline for examining and interpreting species-area relationships, utilizing stream rocks as islands. We will also comment on the effectiveness of using this exercise in a freshman teaching laboratory.

Islands are not necessarily land masses surrounded by water. Ecologically, islands may be defined as discrete patches of similar habitat difficult to reach by potential colonists (Simberloff 1974). For example, lakes have been considered habitat islands (Lassen 1975). The land mass between lakes serves

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[2]Present address: Metcalf & Eddy, Inc., 50 Staniford St., Boston, MA. 02114.

[3]Present address: Dept. of Forestry and Conservation, University of California, Berkeley, CA. 94720.

[4]Actual extinction or emigration. We define "extinction" as the total disappearance of a species from an island, not precluding recolonization (MacArthur and Wilson 1967).

as a barrier to the dispersal of freshwater snails. Snails can reach new lakes through passive dispersal (e.g. transport by waterfowl). Coral heads (Abele and Patton 1976), flowers (Seifert 1975), oak trees (Opler 1974) also are examples of habitat islands. One can easily think of other habitat islands such as rock outcrops and bogs.

Stream rocks have also been considered as habitat islands (Stout and Vandermeer 1975; Trush 1978). Stream rocks are insular since current can present a barrier to immigration (Trush 1978). Each rock supports an assemblage of macrobenthic organisms composed primarily of immature aquatic insects, gastropods, water mites, and to a smaller extent, oligochaetes and planarians. Species of macrobenthos drifting in the current are subjected to predation and physical damage but they are good immigrators. Stream substrates are quickly colonized at any time of the year. For example, hemispherical concrete molds ($\sim$ 13 cm in diameter), left in Sinking Creek (Giles Co., VA.) for 30 days, accumulated an average of 27 macrobenthic taxa (Trush 1978).

PROCEDURE

Selection of Stream and Rocks

This exercise may be done in most streams. The best habitat would be a shallow riffle characterized by a cobble bottom and turbulent flow. Rocks sampled should be from 4 to 30 cm in diameter. Variability will be minimized if rocks of uniform color and surface texture are selected and the sample areas of the riffle are of uniform depth and velocity. Rocks should be gently lifted from the stream bottom and then removed from the water with a slow downstream motion to avoid excessive loss of organisms. Some organisms may be lost in this fashion but the use of a downstream net may include taxa not directly associated with the rock surface (e.g. those in bottom gravels and detritus; Trush 1978). Rocks can be placed in suitably sized plastic containers for transport. Cold storage will maintain samples for up to two days. Ideally, the students should collect their own rocks.

Removing Organisms from the Rocks

Organisms are best removed from each rock by scrubbing and rinsing over a large funnel fitted with $\sim$ 500 µm mesh bolting cloth. Plastic gallon milk containers with the bottoms removed make ideal funnels. The original screw cap, with the center removed, will secure the bolting cloth to the neck of the funnel. Additional scrubbing may be necessary to remove net-spinning caddisflies and other attached forms. After the rock has been cleaned, the organisms can be rinsed from the bolting cloth into a dish.

Constructing Species-Area Curves

Species number estimates. Samples are best sorted live (with a magnifying glass or, ideally, a dissecting microscope) because a moving animal is readily seen by the untrained eye. Insects should first be separated into taxonomic order (Appendix I; see also Borror and DeLong 1971; Needham and Needham 1962; Pennak 1953; and Usinger 1967). Keying to order should be a valuable introduction to stream ecology. (It allows the instructor to point out various life histories and adaptations to flowing water.) Once keyed, all members of each order can be placed into individual Petri dishes and then sorted into a collect-

ion of taxa based on differences in shape and colour. Although we use the terms species and taxon synonymously, taxon is probably more appropriate.

Rock surface area estimates. Area can be roughly estimated by calculating the area of the sphere, cube or solid rectangle which best approximates the actual shape of the rock. For example, the surface area (A) of a cube-shaped rock with an average length (l) of one side would be:

$$A = 6l^2$$

A more accurate method would involve painting the rocks with several layers of rubber latex (Trush 1978). After drying for 24 h, the molds can be peeled from the rock and stretched over graph paper to be outlined. A count of all squares within the outlined portion is an accurate estimate of the rock surface area.

Presentation and analysis. Students should then plot the number of taxa versus the area of each rock on regular graph paper. The best line, fit by eye, can then be drawn through the points revealing the nonlinear nature of the species-area relationship. Generally, this relationship has been described by the following equation (Wilson and Bossert 1971):

$$S = CA^z$$

where S is the number of species or taxa, A the area and C and z are constants. These species-area curves can be tested for goodness of fit by linear regression techniques where log A is the independent variable and S or log S is the dependent variable (Trush 1978). However, the data we discuss below were plotted on regular graph paper and a line, fitted by eye, was drawn.

RESULTS AND DISCUSSION

We have completed the exercise in two, 2 h lab periods. During the first lab, the instructor familiarized the students with the natural history of stream animals and the students identified the benthic animals previously collected. For the second lab, rocks were measured and the species-area relationship calculated and examined. Groups of three to four students were assigned to each rock. The use of latex molds to estimate rock surface area increased the length of the second lab.

Species-area data collected by students, compared with the authors' data from the same site, showed that while both curves have similar shapes, the students' curve was displaced toward a lower number of taxa for any given area (Fig. 1). The students may have overlooked some of the smaller species and may have failed to distinguish similar species.

A wide range of surface areas are needed to adequately describe the species-area relationship. For a narrow range of small rocks (e.g. 16-200 cm^2), the species-area trend may not be clear due to variability in species number (Fig. 1). For a narrow range of large rocks (e.g. 600-1000 cm^2), the differences in species number with area are small and the asymptotic nature of the curve is evident (Fig. 1).

What does the higher number of species on larger "islands" tell us about these "islands"? An ecological definition of species can be: a population of organisms, sharing a common gene pool, surviving through time in a manner different from other populations. Therefore, each species in an ecosystem utilizes the environment in a manner differently from its neighboring species. If seven different taxa are found on a rock it may indicate that the rock environment is being utilized in as many as seven different ways. We do not want to suggest that stream rocks are real ecosystems or that macrobenthos spend a significant portion of their life cycle on one rock. Rather, we have found that viewing stream rocks as model ecosystems allows the student to associate fundamental terms such as diversity, species, individual, and abundance with something they have intimately observed. We had found that while most students claimed complete understanding for these terms it was not until they worked through the exercise that they truly understood them.

Variability inherent to stream rocks should not be attributed solely to sampling error. Rather, analyzing these inherent sources of variability can be used to generate other questions. For example, rocks in Sinking Creek are of variable surface texture. Some are smooth while others have contorted

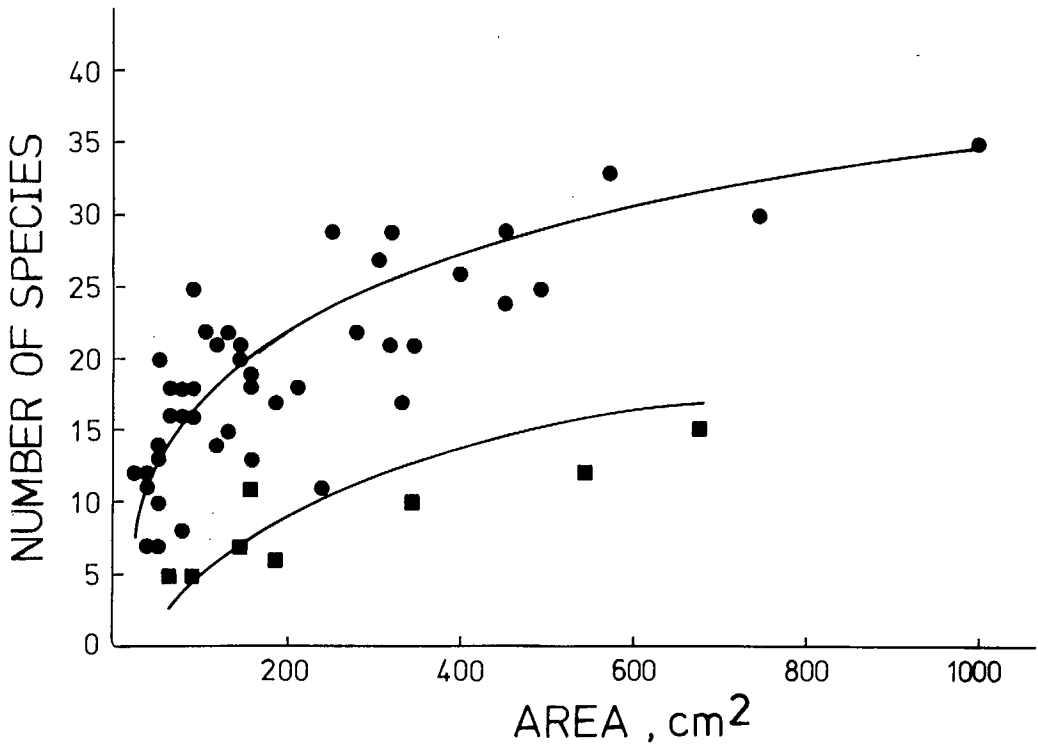

Fig. 1. Species-area relationship for rocks in Sinking Creek, VA.
● - data of authors; ■ - data obtained by students.

surfaces (e.g. crevices and pores). Since many stream insects are substrate specific (Hynes 1970), you might expect that rocks with both smooth and rough surfaces would support the greatest number of taxa. Students may contrast the number of taxa found on smooth and rough rocks of similar area. They will find that more complex surfaces may support more taxa (Trush 1978).

One assumption implicit in this analysis is that all rocks sampled have been exposed to identical physical and biological influences. Any change, however, can shift the balance between immigration and extinction causing variability in species number for rocks of similar area (Trush 1978). Such factors as velocity, depth, nearness to other rocks and objects trapped against the rock all affect the number of species present. For example, one factor might be a leaf trapped against the upstream face of a rock. The dead water space behind the leaf would create an ideal hiding place for organisms requiring slow current. In addition, the leaf may bring in new species (the "Kon-tiki effect"). Furthermore, trapped leaves may shade and prohibit growth of algae, which may have been a food source for other organisms. The list could continue.

Students can investigate the magnitude of sampling variability due to observer bias. Estimating species number is related to a student's powers of observation and interest. Allowing different sets of students to estimate the number of taxa found on the same rock would be a good way to demonstrate this sampling variability. This exercise can be expanded easily. For example, rocks from polluted and nonpolluted streams or rocks from fast and slow currents could be compared. It would be interesting to compare the number of taxa on one large rock with the number on two or more small rocks whose combined total area equalled the area of the large rock. The small rocks will probably have a higher total number of taxa. In addition, the numbers of macrobenthos per unit area can be compared for rocks of different sizes. Students may be surprised to find that density decreases with increasing area (Trush 1978).

The main advantage of this exercise is that it helps the student associate some ecological concepts with an observed phenomenon. Also, the inherent variability of the information collected is utilized to ask other questions rather than being ignored as often happens with routine lab exercises.

ACKNOWLEDGMENTS

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APPENDIX I: A SIMPLIFIED KEY TO THE ORDERS OF MOST IMMATURE AQUATIC INSECTS EXCLUSIVE OF PUPAE

(Composed by Dr. F. Sherberger. There are also aquatic and semi-aquatic Orthoptera, Hymenoptera and terrestrial insects that you may encounter in the water.)

1a With external wing pads[1] 2

1b Without external wing pads 5

2a Labium (lower lip) folded back beneath head, capable of being extended forward. . . . . .ODONATA (Dragonflies and Damselflies)

2b Labium not folded beneath head 3

3a Mouthparts in the form of a pointed beak, for sucking and piercing[2] . . . . . .HEMIPTERA (True bugs)

SPECIES-AREA PATTERNS OF STREAM ROCKS

3b Mouthparts not as above, mandible usually present, distinct. 4

4a With only *one* tarsal claw, usually with 3 long, many-segmented tails[3] . . . . . EPHEMEROPTERA (Mayflies)

4b With two tarsal claws, with 2 tails. . . . . .PLECOPTERA (Stoneflies)

5a A springing apparatus present on the venter of the abdomen, small insects with mouthparts withdrawn into the head. . . . . COLLEMBOLA (Springtails)

5b No such device on abdomen, mouthparts exposed. 6

6a No *jointed thoracic* legs present[4]. . . . . DIPTERA (True flies)

6b Jointed thoracic legs present. 7

7a Some abdominal segments bearing fleshy "legs" which are armed with a series of sclerotized hooks. . . . LEPIDOPTERA (Butterfly and Moth larvae)

7b No abdominal legs with series of hooks 8

8a Anal segment of abdomen with a *single* pair of sclerotized hooks, which may be on "legs" . . . . . .TRICHOPTERA (Caddisflies)

8b No such hooks, or two pairs. 9

9a Abdomen with 7 or 8 pairs of lateral filaments, labrum visible from above[5] . . . . . MEGALOPTERA (Hellgrammites and Alderflies)

9b Abdomen without filaments, labrum not visible from above. . . . . COLEOPTERA (Beetles)

[1]Very early instars will have small pads or none, however, they generally resemble later instars.

[2]Corixidae (Hemiptera) have a blunt "nose".

[3]A couple of genera (*Iron*, *Ironopsis*) have two tails. Also check to make sure that some tails are not broken off.

[4]Beetle larvae of the Curculionidae (weevils) do not have legs.

[5]A few beetles (Gyrinidae, some Dytiscidae and some Hydrophilidae) have lateral filaments.

DEVELOPING MODULES FOR FIELD EXERCISES IN AQUATIC ENTOMOLOGY[1]

Jeffry Gottfried
Group in Science and Math Education
University of California
Berkeley, California 94720

and

Vincent H. Resh
Division of Entomology and Parasitology
University of California
Berkeley, California 94720

ABSTRACT

A self-instructional field module was developed at the University of California, Berkeley, to provide students with concrete field experiences before dealing with abstract counterparts in lectures. The module was designed to introduce undergraduate students in aquatic entomology to benthic sampling, species diversity, and the use of benthic invertebrates as indicators of water quality. The module provides background information, specifies tasks, asks questions, and gives immediate feedback through an answer sheet. Advantages offered by the use of modules are presented. Additional modules on selected topics in aquatic biology should be developed and a module exchange program organized.

INTRODUCTION

The ancient Chinese proverb: "I hear and I forget...I do and I understand", has been reinforced by modern researchers in science education who have found that people learn scientific skills and concepts better when they have had actual experiences with the skills and/or activities that illustrate the concepts, than when they are asked to learn on an abstract level only (Lawson 1975; Jaus 1977; Schneider 1978). In fact, for many situations an experiential base is essential before abstract learning can take place.

The progression in human intellectual development from the "concrete" stage (i.e. dealing only with tangible objects and situations) to the "formal" stage (i.e. having the ability to deal with abstract and symbolic ideas and problems) is controlled by both growth and experience (e.g. Lawson and Wollman 1976; Piaget 1972; Schneider 1978). The potential for formal thought appears at about age 12 but studies suggest that as many as 50% of college freshmen are still concrete thinkers and require experiences to learn (Keasey 1970; Lawson and Renner 1974; McKinnon 1976). According to Piaget (1964), whose pioneering work in intellectual development forms the basis for much current research in science educa-

[1]Paper presented in the symposium on *Innovative Teaching Approaches in Benthic Sciences* at the 26th Annual Meeting of the North American Benthological Society, Winnipeg, Canada, May 10 - 12, 1978.

tion, a person has *learned* something when s/he can *act* upon the knowledge and *do* something with it, as opposed to merely being able to repeat the correct words or select the correct answer from a list of alternatives (see also Renner 1976).

How can instructors provide students with concrete experiences before dealing with abstract counterparts in lecture? In demonstrating ecological principles, how can we get students into the field, earlier and more often, and have them take samples and work with the "substance" of biology given the time and resource constraints of a one quarter or semester class?

We wanted beginning students in an aquatic entomology course to sample aquatic organisms, determine the relative diversities of these organisms in freshwater habitats, and to start thinking about the variables responsible for the observed diversity before this was discussed in class. We decided to develop self-instructional field modules that would be appropriate because we have had positive experiences using and developing instructional modules and we were aware of their educational potential. We define "module" as a self-contained educational program on a specific topic that provides background information, specifies tasks and questions for the student, and gives immediate feedback through an answer sheet (Commission on Undergraduate Education in the Biological Sciences 1971).

METHODS

In developing our module on sampling and diversity, we first formulated overall objectives to be met by the module and then enumerated specific facts, concepts, and skills that we wanted to teach through this module. Next, we outlined a brief sequence of instruction to prepare students for a field exercise. A module should neither be a textbook nor a transcript of a class lecture but rather it should be a series of bits of information and exercises for the student to do and respond to. Generally, modules are divided into frames, each of which teaches one idea or concept. Because the module was intended for use outside of class, independent of an instructor, it was important that it be clear; therefore we decided to involve the students in developing it. We gave the module to undergraduates in the aquatic entomology course at the University of California, Berkeley, to use and criticize. Then, we observed them doing the field exercise to see if they were using the module as we had envisioned. Such observations are an essential step in developing a module; based on them, we clarified and revised the module. The module is entitled, "An Introduction to Benthic Sampling and Species Diversity", (Appendix 1) and was specifically designed for undergraduate students in an aquatic entomology course but could be used in a wide variety of courses and for a diverse array of students (including college freshmen or advanced high school students). The Sequential Comparison Index was developed by Cairns et al. (1968). Assigned readings for the sampling component of this module include Merritt et al. (1978) and Resh (1979) and for the species diversity components include Wilhm and Dorris (1968), Goodnight (1973), Peet (1974), and Resh and Unzicker (1975).

OBSERVATIONS

We learned from our observations in the field that most students could

carry out the field exercise as we had envisioned it. However, we observed that some students who could verbally *describe* sampling procedures correctly, could not actually do them correctly in the field. Thus, the ability to give correct answers in a lecture-discussion setting differed from actually *doing* the procedure correctly. This is in agreement with Piaget's theories discussed above.

We have reason to be optimistic about the module's usefulness based upon student responses after the exercise. Students asked such questions as: Would you get the same results if you repeated the sampling? How many times should the SCI (diversity index) be repeated? When should you use one of the other diversity indices? Questions were aired, points clarified, and a lively discussion followed. But even more importantly, the students who were in their first week of the aquatic entomology course were asking questions based upon their own aquatic entomology experiences. Having had concrete experience in the field, they were better prepared for the more abstract (formal) discussion in class. They were actively participating and sharing ideas, experiences, and observations; the instructor was not the sole source of knowledge for the class. We consider this outcome of the field module extremely important.

We also gave written tests before and after completion of the module on the information contained in it (Appendix 2). Grades improved from an average of 66% before to 97% after completion of the module.

CONCLUSION

Modules offer a number of advantages to an instructor: (1) Class time is more efficiently used because students synthesize ideas and concepts from their own experiences. (2) Students can be expected to be more active in class discussion as they attempt to make intellectual sense out of their concrete experiences. (3) There is decreased dependence upon lectures and increased student responsibility for learning. (4) Easy evaluation can be made of student progress in a course. (5) Modules, unlike texts, can easily be kept up to date.

We are pleased with our first field module and plan to continue using and modifying it. Also, we plan to develop further modules on selected topics in aquatic biology (e.g. rearing insects in the laboratory). We would like to encourage others to do the same. A module exchange program could be organized between various instructors in aquatic biology or aquatic entomology courses. This would enable the time-consuming module development process to be shared and, at the same time, students could benefit from working with modules prepared by experts in their respective specialized fields.

JEFFRY GOTTFRIED AND VINCENT H. RESH

APPENDIX 1: AN INTRODUCTION TO BENTHIC SAMPLING AND SPECIES DIVERSITY

TO THE STUDENT

This is a self-instructional module on *species diversity* and *sampling*. Do the exercises, answer the questions, and then check your answers (see answer sheet, last page) to be sure that you understand the material presented.

When you have completed this module you should be able to:

1. Explain the rationale for using aquatic organisms as water quality indicators.
2. State the characteristics of a good sample.
3. Define species diversity.
4. Set up a line transect across a stream and sample a riffle.
5. Set up a depth contour transect and sample a pool, pond, or lake.
6. Calculate a sequential comparison index (SCI) and determine the relative diversities of benthic communities in a particular habitat.

Bear in mind that the information provided in the field part of the module is the minimum amount necessary to get you started in the field exercise. Undoubtedly, you will be making observations and discoveries that will lead you beyond the information provided. Remember your questions, observations, and comments for later discussions in class with your instructor and fellow students.

MODULE INTRODUCTION

Traditionally, the analysis of water quality has relied heavily on physical and chemical measurements. However, these measurements are *instantaneous*; they only reflect conditions that exist at the time the sample is taken. They do not really indicate anything about past conditions and may offer little in terms of predicting what would happen if an accidental spill of sewage, a toxic chemical, or any other potentially damaging substance was discharged into a particular aquatic habitat.

Organisms that occur on the bottom of lakes and streams offer an added dimension to water quality analysis because they are more than just instantaneous measurements. Since they are present through a range of environmental conditions, they may provide valuable clues about past water quality conditions and may also be used predictively in analyzing the effect of perturbation on the functioning of aquatic ecosystems.

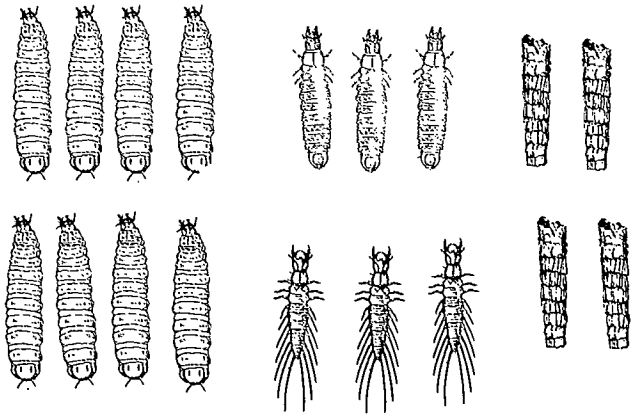
For many years, aquatic macroinvertebrates have been studied and their use as indicator organisms explored. One approach has been to analyze the *diversity* of the benthic (bottom) community and relate this diversity to changes in water quality. In this module you will examine one method of analyzing community diversity which can then be used to provide information about the environment in which these organisms live.

DEVELOPING MODULES FOR FIELD EXERCISES

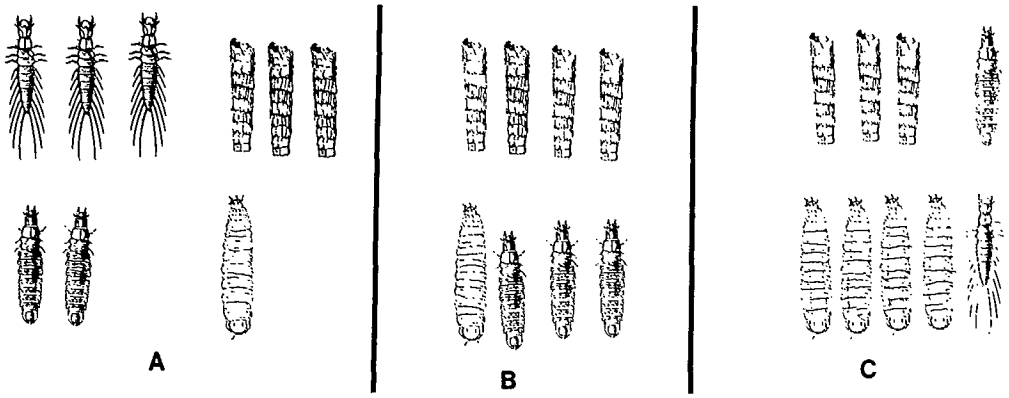
EXERCISES

I. Since we cannot examine the whole community, we must draw a *representative sample* of the organisms as they occur in the real environment. What constitutes a good sample? Basically, it is one collected so that analysis of its composition gives accurate information about the relative abundances of the species present in the total community. The aquatic biologist cannot look at every organism, even in a very small stretch of stream. Therefore, s/he must depend upon a series of samples for information about the total population.

Three samples (A, B, C) were selected from the *known population* of an artificial stream located in a laboratory. Which would you say was the best sample?___

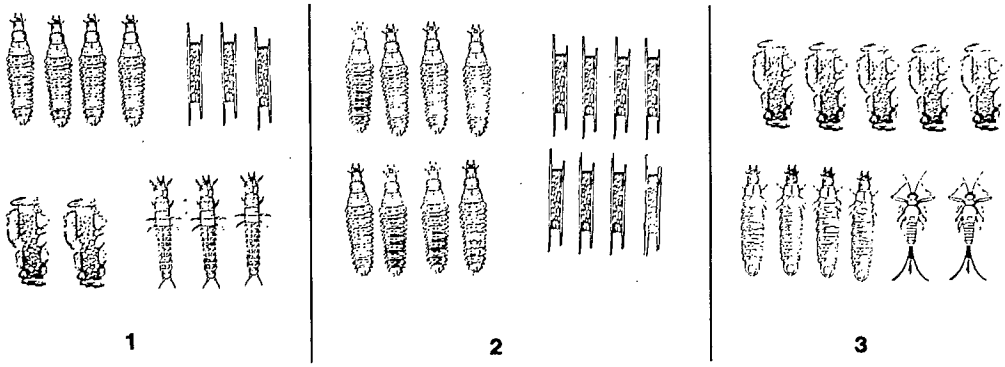


KNOWN POPULATION

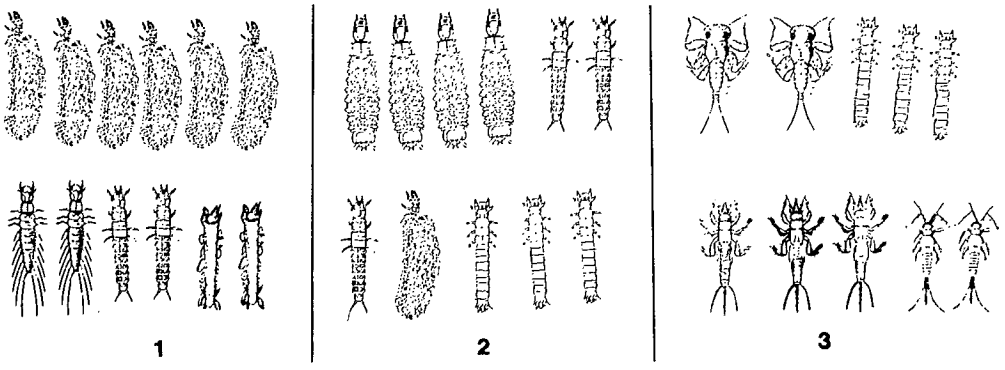


II. Diversity

In general, rivers and lakes with more diverse benthic communities are considered less disturbed than similar systems that have less diverse communities. (The exceptions to this general rule will be discussed in class.) But what do we mean by diversity when applied to aquatic communities? Suppose we had three communities. Which do you think is the most diverse?___ Why?



IIA. The number of species present is one factor considered in the diversity measurement. However, diversity can mean more than this. Diversity also refers to the relative *evenness* of the abundances of species present in the community. In the three populations below each contains the same number of species but which one has the most even distribution in terms of numbers of individuals per species? (i.e. Which is the most diverse?)_____



IIB. Based upon the following samples taken from three ponds, which one would you say has the most diverse insect community?

Sample-pop. #1		Sample-pop. #2		Sample-pop. #3	
Species	Numbers	Species	Numbers	Species	Numbers
A	87	A	23	A	19
B	3	B	70	B	21
C	5	C	7	C	25
D	3			D	22
E	2			E	13

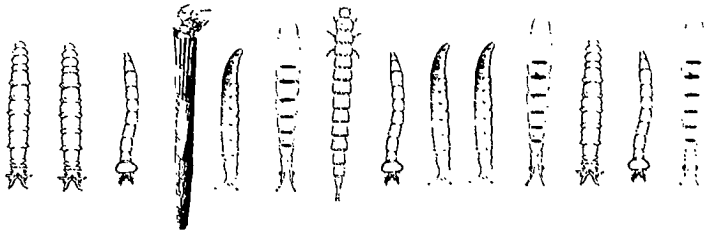
DEVELOPING MODULES FOR FIELD EXERCISES

III. The Sequential Comparison Index (SCI)

In the above examples, the relative diversity of the samples is fairly clear-cut and one could tell the most diverse communities merely by looking at the samples. However, in most cases the situation is more complex. Also, it is often necessary to quantify the diversity of an insect community so that it can be compared to those of other streams or lakes. There are a number of methods devised for calculating a *diversity index* for a community. The one that we will use is called the *Sequential Comparison Index* or *SCI*.

The use of the SCI does not require a computer as do many other types of diversity analyses, nor does it require that the organisms present be identified to species. The index is simply based upon recognizing if an organism looks like another organism (i.e. similar shape, body structure, overall appearance). In order to calculate SCI's, the specimens from your collection are gently agitated in a jar or vial, poured out into a flat tray and then arranged into rows, at all times trying to keep the organisms as close to the position in which they came to rest in the pan as possible (i.e. randomized).

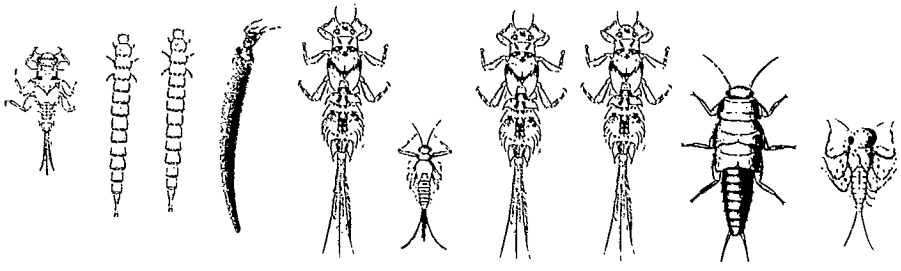
Let's imagine that one of the rows looks like this:



A. Place an X on your paper to represent the first organism. If the organism next to it appears to be the same species, place another X next to the first one; if it looks different and you think that it is a different species then place an O next to the X. Then compare the third specimen to the second. If it is the same as the second then repeat the symbol that you wrote to represent the second specimen. Proceed in this way across the rows comparing each organism with the one before it and recording the appropriate symbols for each one.

Does your sequence of X's and O's agree with the sequence given in the answer section? If not, go back and check again.

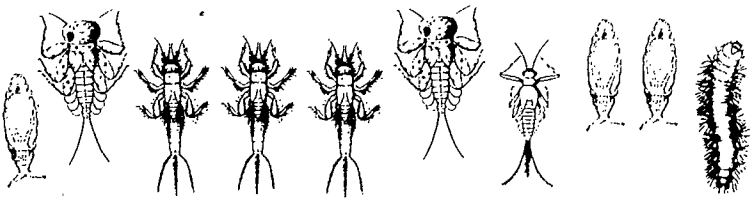
B. Let's try a second example. Again, check your results with those given in the answer section.



C. When two organisms are compared and are found to be similar, they are said to be a part of the same *run*. If they are different, they are members of different runs. Each time you change from X to O you are starting a new run. The greater the number of runs per given number of specimens, the greater the diversity. Now count the total number of individuals in the sample. The SCI is calculated by dividing the number of runs by the number of individuals and is expressed as a decimal.

Now go back to the previous examples and calculate the SCI's. Check your answers with those given in the answer section.

Now try this one.



IV. Transect sampling in running water environments

Very shortly, you will use your knowledge of sampling and calculating SCI's in a field exercise but first it is important to consider exactly how you will take your samples. In order to get good samples, it is important to make collections systematically. The specific sampling plan will vary with the type of habitat (pond, stream, lake, river etc.) and the type of information desired.

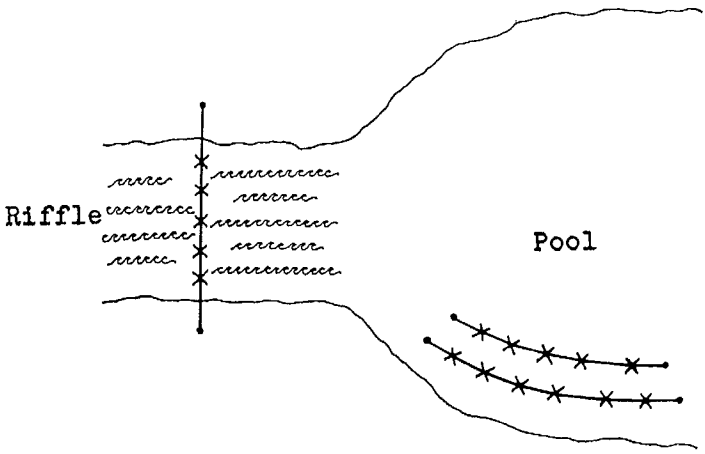


Diagram of a transect line across a riffle and a depth transect in a pool.

In the first part of your field exercise you will be taking samples of aquatic organisms in a *riffle*, a stretch of stream characterized by flowing water of relatively even depth. You will be stretching a line (or making an

imaginary line) across a selected riffle, from bank to bank, and then taking samples at predetermined intervals along the line. At each sampling point place your net on the bottom facing into the current and brush off all organisms from the substrate (rocks, pieces of wood etc.) *within a foot* in front of the net so they are carried by the current into the net. Be sure to check crevices for organisms. If the substrate is gravel or small rock, agitate it in front of the net.

V. Get out there and get your feet wet!

Select a small riffle in a stream of your choice. Establish your sampling transect line and take samples as described above. Be sure to work directly in front of the net so you can catch organisms that get washed off the substrate.

Be prepared to discuss this collector's "techniques" in class.

Take the organisms collected at each sampling point along your transect (e.g. ≈2 - 3 feet apart, depending on the width of the riffle) and place them in a jar or vial. Repeat this procedure until you have 150 - 200 organisms in your jar. (This "lumping" of samples will not be done in our later quantitative sampling exercise.) Then gently agitate the jar, dump the specimens into a white pan, arrange them into rows and calculate an SCI.

VI. Transect sampling in pools, ponds and lakes

In slow moving or standing water where the substrate may be soft, samples may be taken at similar depths (see diagram of pool transect in IV) or across different depths.

VII. No sooner said than done!

Select a pool on the same stream and take a series of samples according to depth contour. Place your net in the water at the first sampling point, scoop up some bottom material and place it in a white pan; sort and calculate an SCI.

VIII. Repeat the above procedure in the standing waters of a pond or lake and calculate an SCI. How do the SCI's of the riffle, the pool and the pond compare? Do you think that it is valid or meaningful to make these comparisons? Why?

Now that you have had some experience in taking samples and calculating diversity indices you have some basic tools necessary to study aquatic insect diversity in different habitats. Think of problems in this area that you might like to pursue in the future.

ANSWERS

I. C is the best sample because it *most* accurately reflects the species present and the relative numbers of individuals per species.

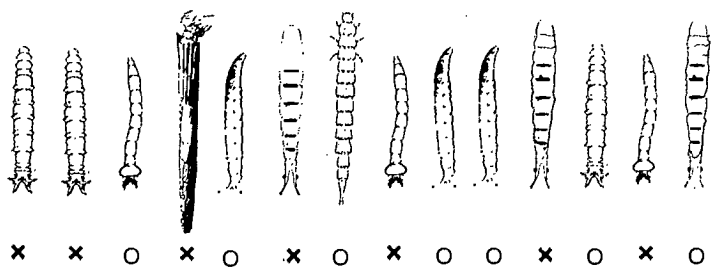
II. Diversity - Community #1 is the most diverse because it is composed of four species, while #2 and #3 are composed of two and three species, respectively.

IIA. Population #3 is the most diverse because no one species dominates. In populations #1 and #2 one species or another far exceeds at least one other species in numbers of individuals. Therefore, #3 is said to have the most *even* distribution and (because all populations contain the same number of species) is the most diverse.

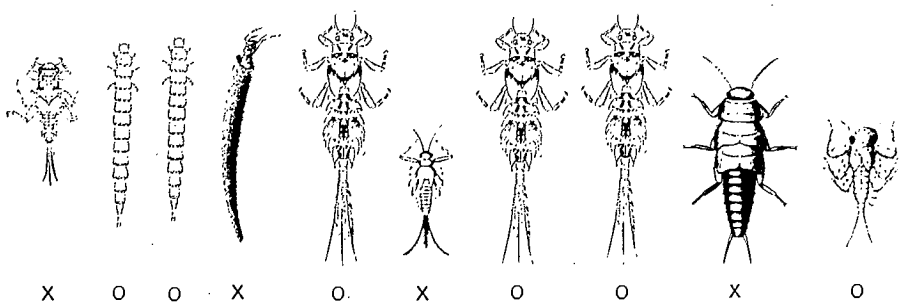
IIB. Of course, one would never try to establish which pond had the most diverse insect community based upon one sample. However, based upon the one sample, population #3 (pond #3) would appear to be the most diverse due to a *greater number of species* than population #2 and a *higher degree of evenness* than population #1.

III. SCI

A.

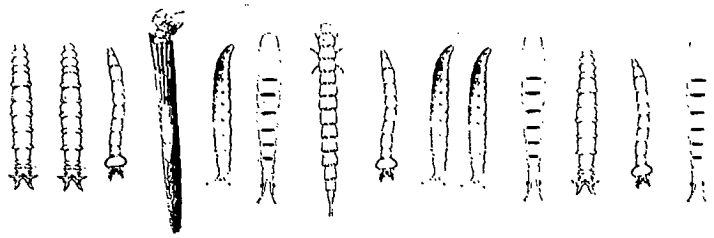


B



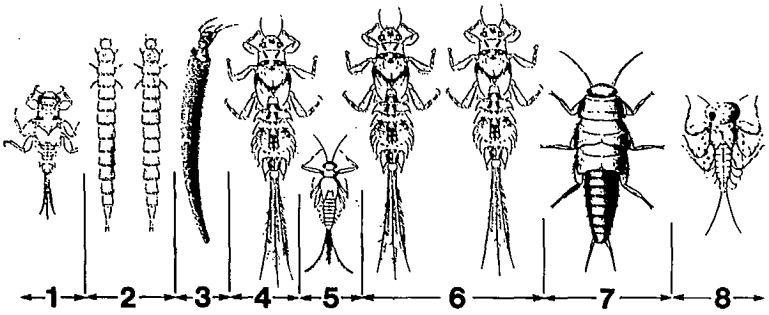
C.

SCI for IIIA.



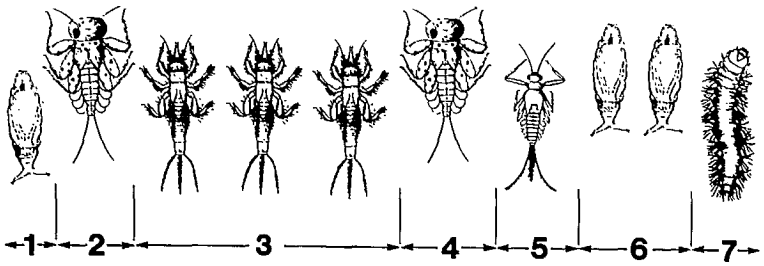
$$SCI = \frac{\text{Number of runs}}{\text{Number of individuals}} = \frac{12}{14}$$

SCI for IIIB.



$$SCI = \frac{\text{Number of runs}}{\text{Number of individuals}} = \frac{8}{10}$$

SCI for IIIC.



$$SCI = \frac{\text{Number of runs}}{\text{Number of individuals}} = \frac{7}{10}$$

APPENDIX 2: TEST GIVEN BEFORE AND AFTER COMPLETION OF MODULE TO EVALUATE STUDENT PROGRESS

1. If asked to, could you systematically sample a riffle for aquatic insects and compute a diversity index?

2. What does it mean that one community of insects is *more diverse* than another?

3. A sample is taken from a known population of an artificial stream. What criteria would you use to judge whether it is a good sample or not?

4. Is it necessary to identify insects to species in order to compute a diversity index for a community? Why?

5. What can a representative sample of aquatic insects tell you about the water quality of the stream from which it came, that a water sample cannot?

6. I prefer to:

   Learn by doing..1....2....3....4....5..Learn by lecture-demonstration (Labs, projects, field trips).

7. In ecology classes that you have taken thus far how would you rate the relative amount of theory to research skills taught?

   Theory..1....2....3....4....5..Research skills.

DEVELOPING MODULES FOR FIELD EXERCISES

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A MODULE FOR ADVANCED FIELD EXERCISES IN AQUATIC ENTOMOLOGY

K. W. Stewart

Department of Biological Sciences, North Texas State University
Denton, Texas 76203

ABSTRACT

A module for graduate-level students taking "Ecology of Benthic Organisms" is described. The module provides students with supervised field and laboratory experience in lotic microhabitat sampling (leaf packs), aquatic insect census and diversity, and trophic/life cycle considerations. Structure of the module encourages innovation, self-reliance, and cooperative problem-solving among the students.

INTRODUCTION

The importance of encouraging student initiative through actual field or laboratory interactions with live aquatic insects is emphasized by Anderson (1979) and Gottfried and Resh (1979). Such activities, within the context of professorial enthusiam and thought-provoking guidance, are as stimulating to more advanced or graduate students as to those getting their "feet wet" for the first time in an ecology or entomology class. Just as it is challenging to an instructor to stimulate basic inquiry in undergraduates, it can be exciting to do the same with graduate students, whose previous experience and academic enlightenment we sometimes overestimate.

I have tested several "modules" or graduate exercises, designed to give students direct experience with aquatic insects. For example, Szczytko and Stewart (1979) emphasize experimental use of stonefly drumming behavior by students in a laboratory. The module considered here provides students with supervised field experience in lotic microhabitat (leaf pack) sampling (Merritt et al. 1979), aquatic insect census and diversity, and trophic/life cycle considerations. It basically provides the attributes of a module (i.e. a self-contained educational exercise on specifically related topics) with specific tasks and questions to provide student feedback, and de-emphasizes background information, encouraging the more advanced students to search the literature.

Additionally, the module invites a group of students to act cooperatively as a research team, and to present their coordinated findings to the instructor in the form of a research paper. Most of the students taking the course (Ecology of Benthic Organisms) have taken Limnology, Limnological Methods, Chemistry of Water and Pollution, Entomology, and perhaps Aquatic Entomology. Because traditional limnology courses often give disproportionate attention to characterizing and measuring physical-chemical parameters, and because relatively unpolluted habitats are selected for study, biological phenomena are emphasized.

Students are required, after a week's exposure to the module, to present the instructor with a list of equipment needed for the exercise. This promotes further self-reliance, cooperative problem-solving, and innovation on the part of the class.

K. W. STEWART

A MODULE ON SAMPLING ANALYSIS AND BIOLOGY OF STREAM INSECTS INHABITING LEAF PACKS

Coarse particulate organic matter (CPOM), in the form of leaves and other plant material from riparian vegetation, represents a major allochthonous energy source for most stream systems (Cummins 1974). This module is intended to give you experience in sampling natural aggregations of leaves during an active period of their processing and to arouse your curiosity as to the basic biology and interactions of insects inhabiting them.

The suggested procedures are intended only to lead your team into basic inquiry regarding leaf pack biology, and to your recognition of logistical sampling problems often encountered in stream habitats. Literature searching and team innovation should make you aware that these procedures and methods of analysis can be (or have been) improved upon. Some of the major questions to be brought out by the procedures include: (1) How can natural leaf packs be retrieved with a minimum of insect washout? (2) How can insects be most effectively separated from leaves? (3) Can most or all immature stream insects collected be identified to the species level? (4) How do we determine and express abundance and/or biomass of insects in leaf aggregations? (5) How evenly are various species or groups distributed in the community? (6) How evenly are sizes of individuals within species represented and what does this suggest relative to life history and larval growth patterns? (7) What are trophic roles and possible food preferences in these insects, and how much dietary overlap (potential competition for food) exists among the species?

Samples should be taken over a 3-month period during late fall or winter. The class as a whole, or in assigned groups, should work as a research team with cooperative allocation of ideas, labor and report preparation. The instructor is available on a consultation basis and for group discussion of questions and problems arising out of the exercise.

A: Procedures

(Develop a condensed set of forms to accommodate data generated from 2, 7, 8, 9, 10, 11, 12 and 13 below.)

1. Obtain leaf and debris packs from or adjacent to riffles. Can your group innovate methods to reduce washout of organisms as the leaf packs are being retrieved? Fill small-sized styrofoam chest(s) of measured dimensions for transport back to laboratory.

2. Record stream temperature, flow, substrate type, date and other pertinent data.

3. Use luke-warm water from laboratory faucet (with attached rubber tubing) to wash insects from leaves-debris into a stack of 4 sieves with successive mesh sizes of 2800, 1180, 425 and 180 μm. This takes advantage of a thermal shock that very successfully causes the insects to lose their hold and become separated from the vegetable material.

4. Rewash all material on top sieve until only animals and inseparable debris remains.

MODULE FOR ADVANCED FIELD EXERCISES

5. Preserve contents of sieves in 50 - 70% ethyl alcohol.

6. Separate all insects into "apparently different taxa" per sample. All subsequent numerical, volumetric or gravimetric measurements can be expressed either *per cubic volume of leaf pack,* or perhaps, more usefully, *per g dry weight of leaf pack.*

7. Identify animals to lowest taxon possible and enumerate each taxon/sample. A counting device will be necessary to increase efficiency.

8. Dry insects from ½ of each sample taken in the field (by taxon) for 24 h at 120 - 150°C. Express these data as total g/taxon and $\bar{X}$ g/individual/taxon for these particular samples.

9. Calculate the Index of Dominance, $c = \Sigma(n_i/N)^2$ for each sample. n_i = importance value of each taxon; N = total of importance values. Importance value can be estimated by the relative proportion of a taxon to all taxa numerically (from 7) and/or gravimetrically (from 8). Higher values indicate dominance by fewer taxa.

10. Calculate species diversity of each sample (Simpson's, $\bar{d}$).

11. Using a calibrated micrometer in scope eyepiece (know how this calibration is made), measure the head capsule width of the first 25 individuals (or fewer if sample is represented by less than 25) per taxon. Express in $\bar{X}$ width/taxon and take particular note of variation in individuals. Speculate, on this basis, how much brood overlap exists in each taxon and therefore how spontaneous (or spread) you would expect emergence to be (unless development is somehow synchronized between the time this sample was taken and the emergence). Speculate on possible difficulty in establishing cohorts of each taxon for production determination.

12. Make a fore-gut analysis of at least 10 individuals of the 5 most numerous (or largest in biomass) species. Make your own dissecting needles and scalpels and use short-cut methods (other than complete dissection)--see literature. Express relative fullness of gut and number and relative volume of each identifiable fraction or dietary item.

13. Calculate Forage Ratio and Ivlev Index for each dietary component of carnivores or omnivores. How could we have made similar estimations for shredders, detritivores, and herbivores (e.g. by altering initial sampling procedure?)? Consult original literature on these indices or Vaught and Stewart (1974). Also calculate *Coefficient of Dietary Overlap* (Horn 1966) for two species that seem to be competing for food.

a) Forage Ratio (Hess and Swartz 1941)

$$FR = \frac{n/N}{n'/N'}$$

where:

- n = no. of any organism in stomach
- N = total no. of organisms in stomachs
- n' = no. of same organism in environment
- N' = total no. of food organisms in environment

FR (range of values = 0 plus)
1 = random feeding
>1 = selected (preferred) or more available food
<1 = less preferred or less available food

b) Ivlev Index (Ivlev 1961)

$$E = \frac{r_i - P_i}{r_i + P_i}$$

where:
r_i = % abundance of a food item in total ration
P_i = % same food item in environment

E (range of values = -1 to +1)
0 = random feeding
>0 = selection for (preference) or greater availability of food item
<0 = selection against or less availability of food item

c) Coefficient of Dietary Overlap (Horn 1966)

$$\hat{C}\lambda = \frac{2\sum_{i=1}^{s} X_i Y_i}{\sum_{i=1}^{s} X_i^2 + \sum_{i=1}^{s} Y_i^2}$$

where:
X_i = that proportion of total diet of species X taken from a given category of food
Y_i = that proportion of total diet of species Y taken from a given category of food
s = total no. of food categories

14. Make microphotographs of each taxon studied to emphasize pertinent identifying characters.

15. Review literature and cite any papers that suggest methods for quantifying debris-inhabiting lotic insects.

16. Write a research report based on above sampling and procedures. Include Introduction, Objectives, Techniques (expand procedures above), Results, Discussion (or combined Results and Discussion).

17. Be able to repeat above procedures, and be aware of application of each step for examination purposes.

B: Thought questions

1. Why is the above method of separating insects from leaf packs an advantage over hand-picking them in the field or lab?

2. Explain why the improved elutriator (Stewart 1975) would not be useful in insect-substrate separation. Could the elutriator possibly be modified to efficiently achieve separation?

3. Why were you not able to identify all insects to the species level?

4. How would you expect the numerical and gravimetric expressions of density to vary seasonally in the groups of insects encountered (procedures 7 and 8)?

5. Of what value are diversity indices in lotic ecology (procedure 10)?

6. How do you account for numerical or biomass dominance of particular species observed in this exercise?

7. What could quantification and study of mid- and hind-gut contents of insects elucidate (even though specific identification of contents might be impossible)?

8. For what reasons should head capsules be measured and size class determinations made?

9. How and under which circumstances can forage ratios, electivity indices etc. mislead and cause misinterpretation of food preference (or electivity) (procedure 13)?

10. How would the Competitive Exclusion Principle relate to studies of food habits, preference and coefficient of dietary overlap (procedures 12 and 13)?

11. What limitations are imposed on biological interpretations of data collected over such a short time span?

CONCLUSION

Students have responded to this module very favorably. They become more receptive to the instructor's suggestion that much basic research is needed on the questions addressed early in the module when they find that some logistical problems and questions they encounter have no firm answers.

The combination of practical field-laboratory experience, literature searching, and report preparation gives students exposure to graduate-level inquiry. This stimulates their thinking and encourages innovation. A major achievement of the module is usually increased student awareness of the advantages and adjustments of cooperative team effort.

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DEMONSTRATION OF STREAM WATERSHED COMMUNITY PROCESSES WITH SOME SIMPLE BIOASSAY TECHNIQUES[1]

Richard W. Merritt[2]

Department of Entomology, Michigan State University
East Lansing, Michigan 48824

and

Kenneth W. Cummins[3]

Department of Fisheries and Wildlife, Oregon State University
Corvallis, Oregon 97331

and

James R. Barnes

Department of Zoology, Brigham Young University
Provo, Utah 84602

ABSTRACT

Two simple bioassay techniques are described to illustrate stream watershed community processes. Leaf litter processing can be examined using experimental leaf packs in streams and litter bags on floodplains and other terrestrial sites. The analysis of invertebrate functional groups or the categorization of communities based on morphobehavioral food-gathering adaptations can also be examined using the above techniques. Leaf litter processing and invertebrate functional feeding group analyses have proven to be useful tools for field classes examining stream and floodplain community structure and function.

INTRODUCTION

There has been strong interest in recent years in developing bioassay techniques which integrate stream community and related watershed processes (Cummins 1974) for comparing streams within and between basins and identifying disturbed reaches of running water systems. We define "disturbed" as abnormal

[1]Paper presented in the symposium on *Innovative Teaching Approaches in Benthic Sciences* at the 26th Annual Meeting of the North American Benthological Society Winnipeg, Canada, May 10 - 12, 1978 and in the symposium on *Innovative Teaching Techniques in Aquatic Entomology* at the Annual Meeting of the Entomological Society of America, Houston, Texas, November 26 - 30, 1978.

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rates of biological activity (e.g. change in the rate of leaf litter processing and associated, disrupted community structure) given a particular physical and chemical setting.

Two general procedures for assessing stream community structure and function are described: 1) the leaf pack or litter bag technique (e.g. Petersen and Cummins 1974; Crossley and Hoglund 1962; Merritt and Lawson 1979) which integrates the biotic (including decomposition) and abiotic processing of coarse, particulate organic matter (CPOM) in a stream or on its floodplain (upper bank) where leaf litter has accumulated; and 2) the analysis of invertebrate functional groups which categorizes stream (and floodplain) macroinvertebrates on the basis

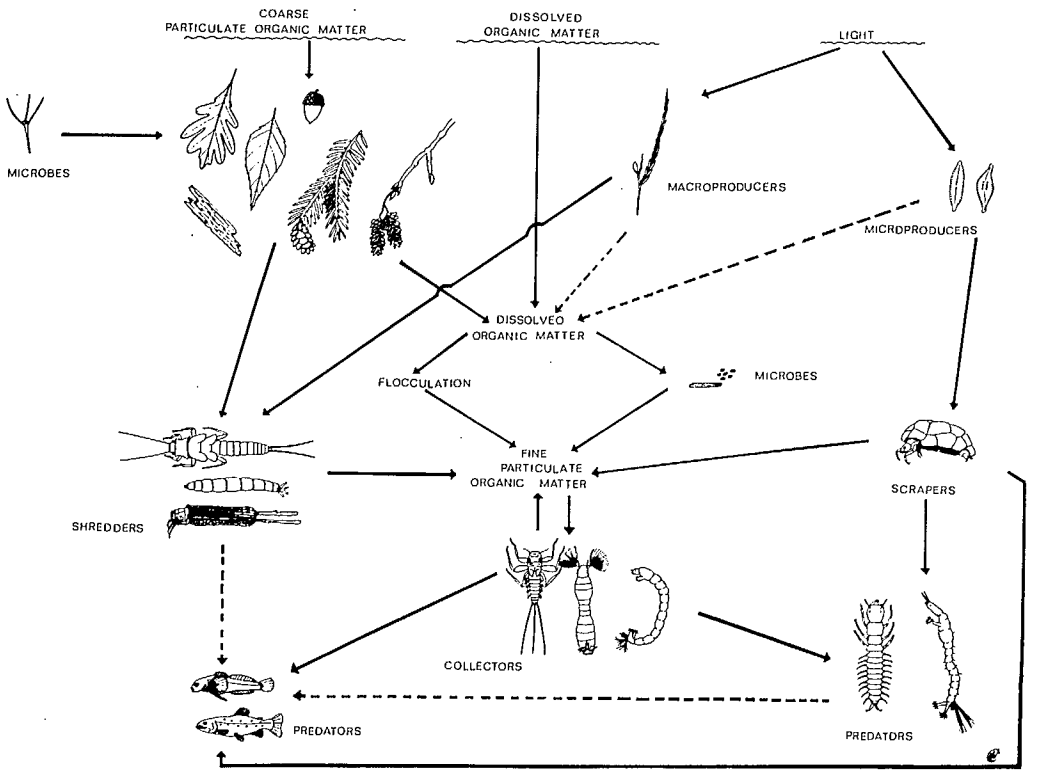

Fig. 1. A conceptual model of stream ecosystem structure and function (modified from Cummins 1973) emphasizing the processing of particulate and dissolved organic matter. Coarse particulate organic matter (CPOM) includes deciduous leaves, coniferous needles, twigs, bark, nuts, and flowers. CPOM initially is colonized by aquatic hyphomycete fungal spores. Dissolved organic matter (DOM) is utilized by rod-shaped and spheroid bacteria. The plant community is represented by diatoms (microproducers) and moss (macroproducers). The animals shown are: shredders-crane fly and caddisfly larvae and a stonefly nymph; collectors-black fly and midge larvae and a mayfly nymph; scrapers-a caddisfly larva; predators-fishfly and midge larvae and fish (sculpin and trout).

of morphobehavioral food-gathering adaptations (Merritt and Cummins 1978).

Within the context of general stream structure and function (Fig. 1), the leaf pack method deals with the leaf litter (CPOM)-microbe (especially aquatic hyphomycete fungi)-shredder (macroinvertebrates feeding on leaf litter) system. Fig. 1 emphasizes the separate functional roles played by shredders, collectors (filterers and gatherers of fine detrital particles), scrapers (algal grazers), and predators. The litter bag method deals with a similar association of organisms and related processes that occur in the foodplain and upland areas of the watershed.

MATERIALS AND METHODS

Leaf Pack Construction

Leaves collected at the time of abscission and air dried ($\sim$ 1 wk) are made into 5 or 10 g packs. For class exercises requiring a large number of packs, 2.5 to 5.0 g packs are sufficient. Packs are made by soaking the dried leaves until softened and assembling them in loose aggregations fastened together with long shank plastic "buttoneers"[1] or monofilament line (Fig. 2). Conifer packs

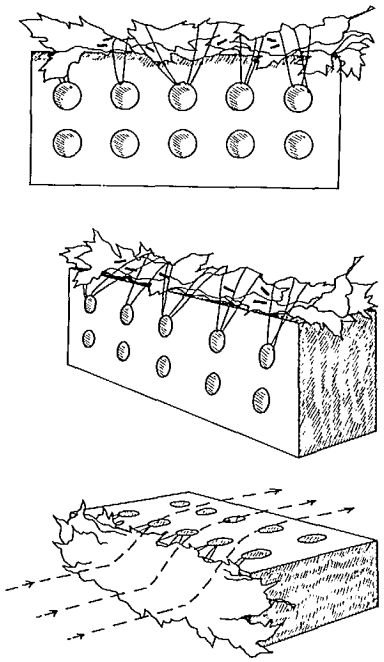

Fig. 2. Leaf packs held together with plastic "buttoneer" fasteners (Dennison Co.) and lashed loosely to brick with monofilament fishing line. Bottom drawing shows packs in position in the stream with direction of current indicated.

[1]Available from Dennison Mfg. Co., Framingham, Mass. 01701, and many variety stores.

can be made by stringing needles on monofilament line. One or two packs are lashed loosely with monofilament or attached with elastic bands to a brick (the latter allows easy use of packs in the field) and then placed in the stream facing into the current to simulate accumulations at the leading edge of an obstruction (Fig. 2). The importance of uniformity of leaf pack construction should be emphasized. Also, packing the leaves too tightly yields widely varying results.

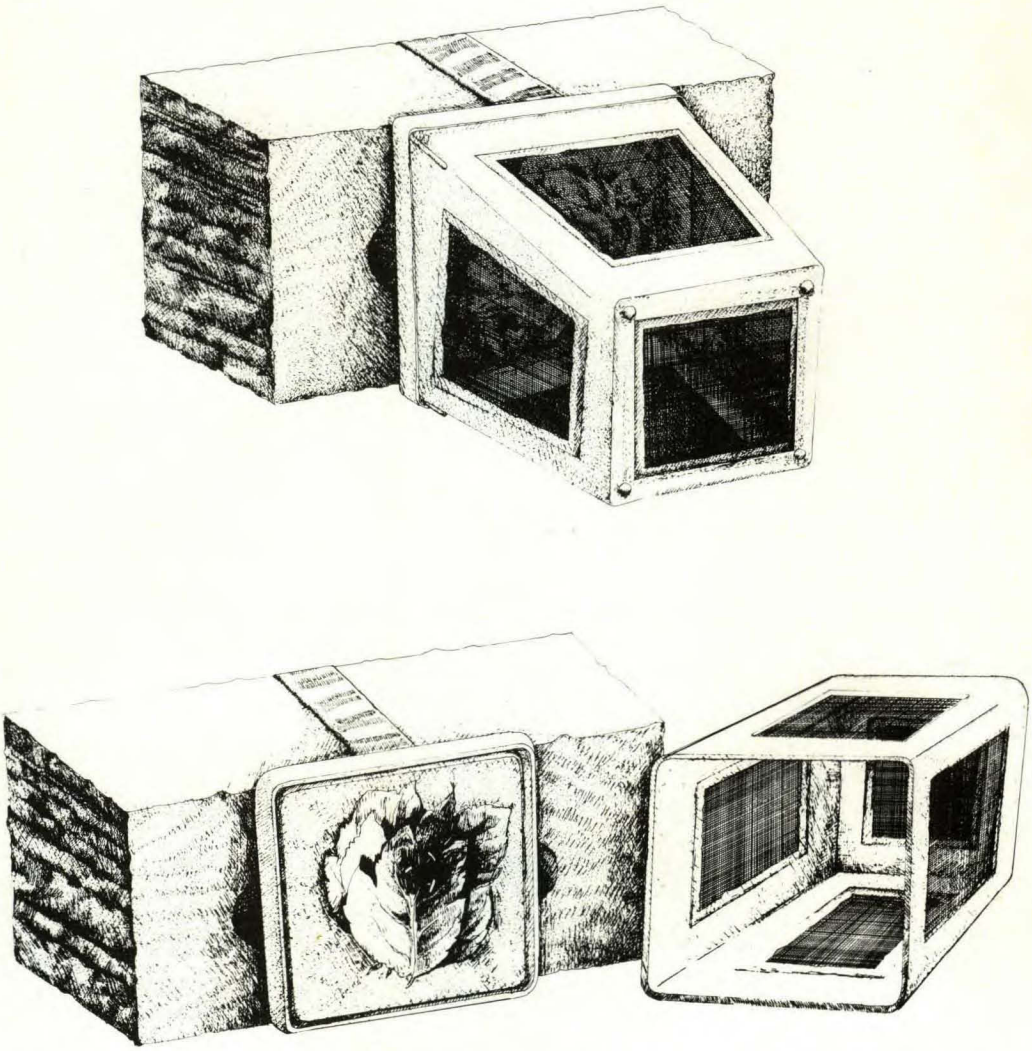

Fig. 3. Leaf pack system utilizing plastic containers or "cages" with different mesh sizes to exclude or enclose selected organisms. Lower drawing shows open cage with leaf pack attached to cage cover and brick. Upper drawing shows closed cage.

The role of shredders in leaf litter processing can be demonstrated also through the use of in-stream cages which enclose leaf packs and shredders (Fig. 3). The cages are constructed from plastic freezer containers from which most of the sides and bottoms have been removed and the open areas covered with Nitex® netting[2]. The netting is attached to the inside of the cage with hot glue. Leaf packs are constructed as described above, except the lid is placed between the pack and the elastic band. The bottom of the container is placed over the leaf pack and snapped into place (Fig. 3). Cages are positioned in the stream with the bottom facing into the current (Fig. 4). If the netting becomes clogged, the bottom of the cage can be removed quickly and cleaned in the stream. The mesh size chosen depends on the size of organisms to be kept in the cages or selectively admitted in natural or artificial stream systems, and the concentration of suspended particulates in transport. For example, in the Michigan streams investigated, the high particulate loads necessitate cleaning of the screens (even 1 mm mesh) at least once per day, whereas in the Utah streams, even 250 µm mesh requires little cleaning. To demonstrate the effect of shredders, different shredder taxa and numbers can be placed in the cages. For example, the processing of leaf packs can be compared in cages that contain 0, 1, 2, 4, and 8 *Tipula* larvae. Also, experiments with various combinations of shredders, collectors, and predators can be set up.

Litter Bag Construction

Following the initial procedures described for leaf pack construction, 4 - 8 g amounts of softened leaves are placed in 10 x 10 cm nylon or fiberglass mesh bags, labeled and sealed. One or several different mesh bag sizes can be employed, depending on the objectives of the class experiment. The following three sizes (Fig. 5), similar to those used by Heath et al. (1964), were chosen to separate components of leaf litter degradation in a woodland floodplain:

a) 50 µm - to exclude all soil invertebrates and allow only leaching and microbial decomposition.

b) 500 µm - to admit microarthropods (mites, Collembola and enchytraeids) and exclude most macroarthropods.

c) 8000 µm - to admit all soil fauna (microarthropods, macroinvertebrates, earthworms and Mollusca).

Since estimates of sample decay rates in confined litter bags have been substantially lower than those observed with individually tethered non-confined leaves (Witcamp and Olson 1963), both techniques can be used to estimate upper and lower rates of litter degradation (Fig. 5). Litter bags and tethered leaves are buried between the litter and humus layers in the field and secured by long nails or spikes. A number of different leaf species can be compared to determine preferences of the litter fauna and/or differential breakdown rates.

[2]Nitex (nylon monofilament cloth) ranging in mesh size from 20 µm to 1800 µm can be purchased from E.A. Case, P.O. Box 45, Andover, N.J. 07821. Less expensive coarse and fine mesh can be obtained from most commercial net and twine companies and retail fabric stores.

Fig. 4. "Caged" leaf packs in position on stream bottom. Current flowing from bottom to top.

Fig. 5. Different sized mesh bags and tethered leaves used for leaf litter processing experiments.

ANALYSIS

At least 3 to 5 leaf packs or bags should be retained to determine initial dry weights if quantitative data are required. Each collection from the field should include a minimum of 3 packs to allow for statistical analysis. Packs should be collected initially from the stream at 48 - 72 h to account for handling breakage and loss due to leaching of soluble organic matter. Packs should be sampled $\sim$ 1 wk^{-1} at 20 to 30°C or $\sim$ 1 mo^{-1} at 0 to 20°C (i.e. collections about every 150 to 300 degree days). In the floodplain, a subsample of litter bags should be collected immediately after being placed in the field to assess weight loss due to leaching, breakage and handling during litter bag preparation. If possible, bags should be evaluated monthly during fall and winter and twice a month during spring and summer. However, sampling frequency will also depend on the duration of the course.

The rate of leaf material loss can be expressed as the percentage remaining after a given time and calculated by:

$$\% R = W(t_f)/W(t_i) \times 100$$

where $W(t_i)$ is the initial weight of leaf material and $W(t_f)$ is the amount of material remaining after time t. The values can be expressed as % loss/day for comparison of sites, species and seasons.

Estimates of the amount (%) of leaf remaining, observations of the type of feeding activity that has occurred, and counts of animals--especially shredders--can be done in the field if only qualitative analyses are required. For example, leaf packs can be separated and washed in the field over a 250 µm mesh sieve and the shredders picked from the screen. Leaf packs or litter bags recovered from reaches of streams or rivers and floodplains having the type of community associated with CPOM leaf litter processing will be obviously different from habitats lacking these organisms.

EXPERIMENTAL RESULTS

Leaf packs were made from representative species taken from along Augusta Creek (Barry and Kalamazoo Counties, MI), placed at two sites and collected after processing 40 days during late spring-early summer. The amount of processing at the two sites was strikingly different (Fig. 6). A much higher processing rate occurred at the headwater site (1st order stream, Strahler 1957) compared to the larger (3rd order), more open location due primarily to the presence of a species of shredder (*Lepidostoma costalis* (Banks)) at the former site. This site was actually colder and less subject to physical abrasion. The processing rates for leaves of a number of common trees and shrubs of riparian zones have been compared by Petersen and Cummins (1974). The rate of processing depends on how fast a leaf is colonized by stream invertebrates which, in turn, selectively feed on leaves with the most microbes.

Differences in the processing rates of ash leaves in coarse mesh bags placed on an elevational gradient on the floodplain can also be demonstrated (Fig. 7). Only petioles and larger leaf veins remained after 6 mo in the point bar of the

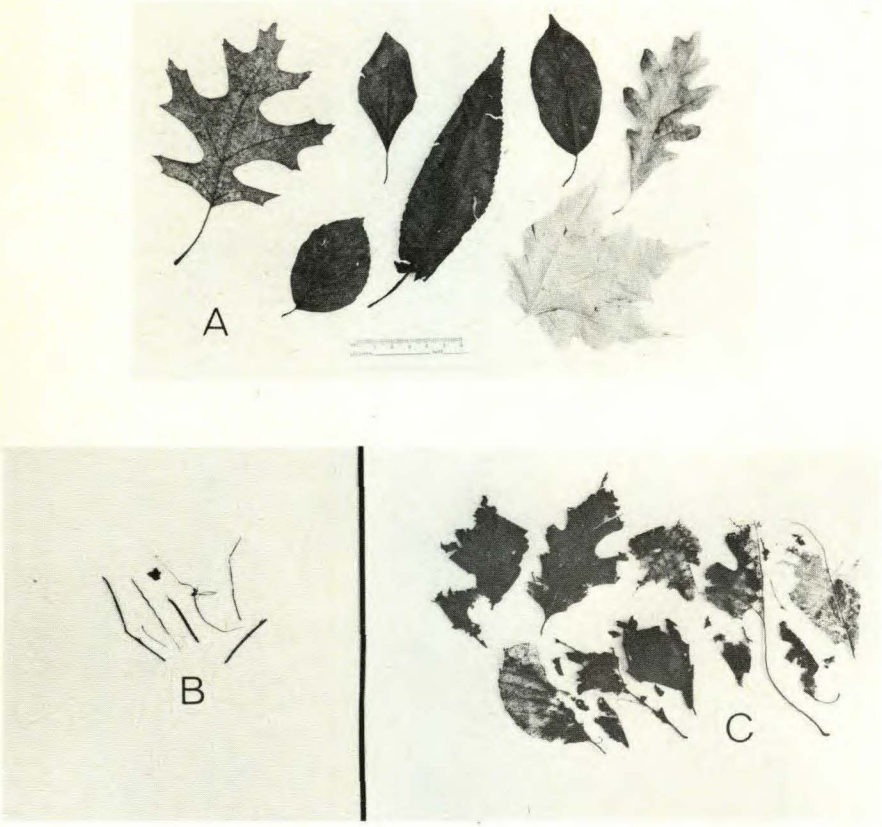

Fig. 6. Leaf processing at two sites on Augusta Creek as demonstrated with the leaf pack bioassay method. Seven species of leaves (A), with two leaves of each species, were randomized and made into leaf packs (Fig. 2). The amount of leaf pack remaining at a 1st order heterotrophic stream site (B) vs. a 3rd order autotrophic stream site (C) after 40 days during late spring-early summer is shown.

floodplain, while in the terrace and uplands progressively more leaf material remained. The faster processing rate in the point bar was attributable to greater diversity and abundance of certain leaf litter macroinvertebrates, higher soil moisture and differences in other edaphic factors (Merritt and Lawson 1978).

A typical class experiment conducted during summer in the Augusta Creek Watershed showed that leaf litter processing proceeded faster in the stream than at terrestrial sites, but faster on the floodplain than on the upland (Fig. 8A). Leaf litter in the stream attracted an invertebrate population of greater biomass than the floodplain or upland site (Fig. 8B). This was probably due to the faster leaching of dissolved organic matter from the stream litter (Petersen and Cummins 1974), followed by rapid microbial attack and subsequent macroinvertebrate colonization.

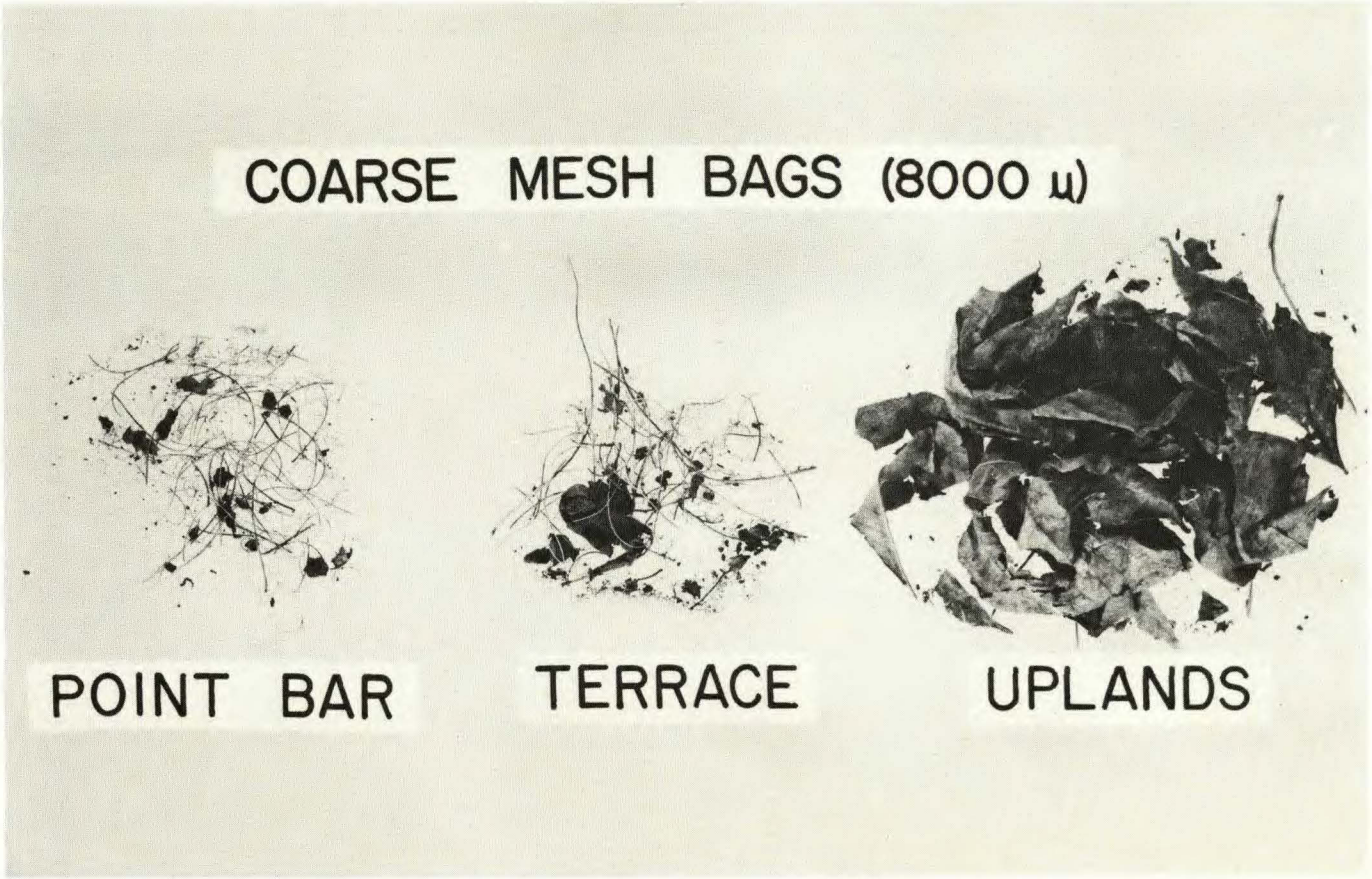

Fig. 7. Leaf processing at three elevational locations on the Augusta Creek floodplain as demonstrated with the litter bag bioassay method. Photographs were taken after 6 mo exposure.

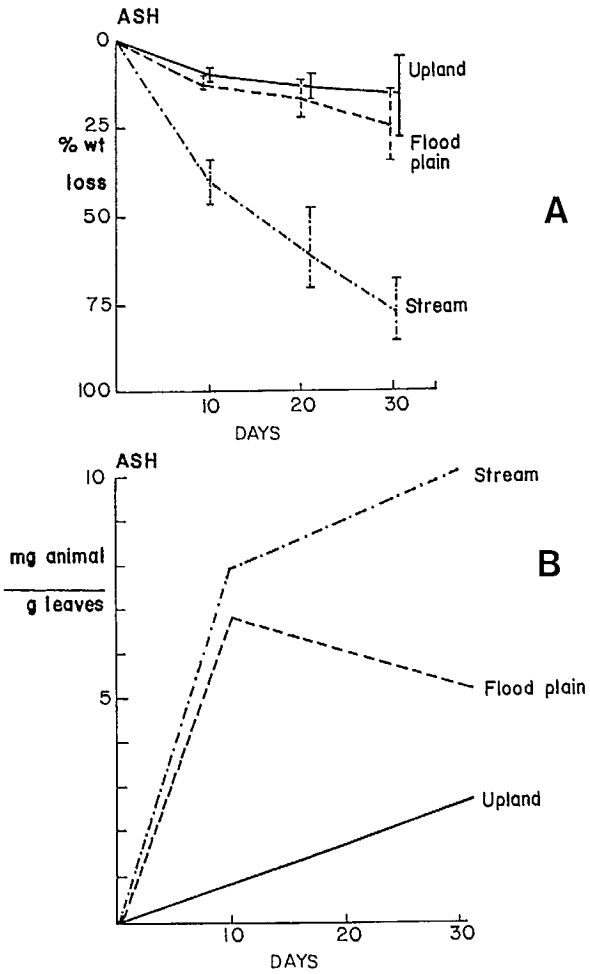

Fig. 8. A class experiment involving leaf litter processing in three different areas of the Augusta Creek Watershed during summer.

FUNCTIONAL GROUPS

To evaluate community level processes associated with the use of various organic energy sources in watersheds, it has proven useful to categorize aquatic and semi-aquatic invertebrates on the basis of functional role (Cummins 1973, 1975; Merritt and Cummins 1978). The relative abundance (numbers and/or biomass) of functional groups can be used to indicate the importance of different organic resources in a watershed. A general function--shredders, collectors (filtering or gathering), scrapers, predators--can be ascribed, at various taxonomic levels, depending on the group. The procedure proposed is to identify invertebrates to

a taxon sufficient to characterize function at a high level of probability, focusing on *mechanisms* of obtaining food rather than *what* is eaten. In its simplest form, the information is expressed as a ratio of various groups (e.g. shredders to collectors). This concentrates identification to maximize the ecological information obtained concerning utilization of organic resources in streams. Some taxa (e.g. Megaloptera and Odonata) are categorized functionally at the ordinal level while others (e.g. Chironomidae) may be characterized reliably only at the specific level. In some, various age classes may function differently (Coffman et al. 1971). Again it *must* be stressed that the procedure focuses more on the mechanisms of obtaining the food source and less on what is eaten.

An attempt has been made recently by Merritt and Cummins (1978) to assign the families and genera of North American aquatic insects to functional groups. Workers in benthic ecology should add to, revise, and correct these initial assignments to enable more rapid and accurate characterization.

The leaf pack and invertebrate functional feeding group bioassay techniques have proven to be useful methods for demonstrating and evaluating community processes (e.g. Cummins 1977). For example, by measuring the processing rates of leaf or needle CPOM (i.e. the conversion to dissolved organic matter (DOM), fine particulate organic matter (FPOM), CO_2 and animal biomass), field classes can compare this community level process and the relative importance of terrestrially derived organic matter at different stream and/or floodplain sites. The role of invertebrates in detrital processing can be evaluated by enumerating shredder populations in leaf packs or tethered leaves and by observing the effects of various exclosures (cages or bags). By comparing shredder densities to the abundance of other functional groups (e.g. scrapers), different sites can be contrasted on the basis of the relative importance of the utilization of coarse and fine detritus (in transport or in the sediments) and periphyton.

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Merritt, R.W., and K.W. Cummins (ed.) 1978. An introduction to the aquatic insects of North America. Kendall/Hunt Publishing Co., Dubuque, IA. 441 p.

Merritt, R.W., and D.L. Lawson. 1979. Leaf litter processing in floodplain and stream communities, p. 93 -105. *In* R.R. Johnson and J.F. McCormick (ed.). Strategies for protection and management of floodplain wetlands and other riparian ecosystems. Proceedings of a symposium held Dec. 11 - 13, 1978, Callaway Gardens, GA. Gen. Tech. Rep. WO-12, Forest Serv., U.S.D.A., Washington, D.C.

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Witkamp, M., and J. Olson. 1963. Breakdown of confined and nonconfined oak litter. Oikos 14: 138 -147.

RÉSUMÉS

Morse, J.C. 1979. Techniques used in teaching aquatic insect taxonomy in North American colleges and universities, p. 3 - 13. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

L'enseignement de la façon d'identifier les insectes aquatiques est un élément fondamental des cours d'entomologie aquatique. Pour évaluer jusqu'où on insistait sur cet élément et découvrir comment se faisait la formation taxonomique dans les universités et collèges nord-américains, on a envoyé un questionnaire à des chargés de cours sélectionnés. On a ainsi découvert que les professeurs d'entomologie aquatique utilisaient une grande variété de méthodes didactiques efficaces dont plusieurs témoignent de la débrouillardise et de la sensibilité de leurs auteurs. Cette enquête a également permis d'établir une liste des chargés de cours d'entomologie aquatique en Amérique du nord et d'obtenir un exposé sommaire des cours. La dernière enquête a démontré qu'au moins 79 chargés de cours donnent des cours sur les insectes aquatiques dans 71 universités et collèges d'Amérique du nord à plus de 820 étudiants par an.

Green, R.H. 1979. Simulation of ecological data for hypothesis-testing and evaluation of sampling designs, p. 15 - 20. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On peut utiliser des données simulées pour évaluer les plans échantillonnage, déterminer la rigueur des méthodes statistiques et vérifier les hypothèses. Cette méthode devrait être incorporée aux cours de méthodologie quantitative en écologie benthique. La simulation utilise des éléments aux caractéristiques connues, contrairement aux éléments obtenus par échantillonnage réel en milieu naturel. Les expériences faites avec des données simulées ont pour but de: (1) déterminer quels sont les résultats si l'on utilise des unités d'échantillonnage de dimensions différentes pour échantillonner les organismes qui sont distribués dans des espaces différents, et (2) vérifier l'Ho qui veut qu'il n'y ait pas de changement dans la quantité d'une espèce indicatrice dans une zone déterminée quand les éléments ne respectent pas les hypothèses des expériences normales. Il est aussi possible de simuler la gradation dans le taux décroissant des biocénoses complètes comprenant de nombreuses espèces, qu'on échantillonne pour évaluer les méthodes à variantes multiples.

Kaushik, N.K., J.B. Robinson, and L. Chatarpaul. 1979. Role of certain benthic microorganisms and invertebrates in nitrogen transformations in stream sediments, p. 21 - 30. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On décrit une expérience de classe simple servant à illustrer quelques aspects des transformations que subit l'azote contenue dans les sédiments d'un cours d'eau et le rôle que jouent les micro-organismes et les vers oligochètes dans ce processus. On introduit dans des éprouvettes de plexiglas des sédiments de cours d'eau et on les incube pendant des périodes variables. Les changements qui s'opèrent dans les concentrations de nitrate indiquent que la nitrification ou la dénitrification est en cours. Ces processus sont accélérés par la présence

des vers oligochètes. On peut également étudier les effets que produisent d'autres facteurs comme la température, la teneur en oxygène de l'eau, la quantité d'énergie (carbone) que contient le sédiment, et la profondeur du sédiment.

Szczytko, S.W., and K.W. Stewart. 1979. Stonefly drumming as a model classroom study of aquatic insect behavior, p. 31 - 37. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

L'étude du tambourinement de la plécoptère peut être utilisée dans la salle de cours pour intéresser directement les étudiants aux insectes vivants. On fournit des instructions sur la façon d'obtenir le matériel vivant, d'observer le tambourinement et d'enregistrer les signaux. Voici les expériences qu'on peut faire en utilisant les combinaisons de nymphes de plécoptère vivantes, d'enregistrements sur ruban et de relevés d'oscillographe: (1) illustrer le phénomène du tambourinement; (2) identifier l'espèce ou la spécificité d'après le comportement et les dialectes possibles; (3) étudier les effets de la température; et (4) examiner la capacité des perles à identifier leurs signaux parmi les sons étrangers. On donne des exemples d'autres questions intéressantes à étudier. L'enregistrement cinématographique du tambourinement des plécoptères permettrait aux étudiants d'examiner plus à fond ce comportement intéressant, et on prévoit le filmer dans l'avenir.

Berg. C.O. 1979. An improved technique for projecting images of living aquatic insects, p. 39 - 46. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On décrit deux sortes de petits aquariums qui peuvent être introduits dans un diapos-projecteur de 2 po sur 2 po pour projeter des images d'insectes vivants et d'autres invertébrés d'eau douce. On décrit également un bâti de miroirs qui permet de rectifier les images des aquariums. On peut utiliser l'appareil dans la salle de cours, par exemple, pour démontrer les mouvements respiratoires, locomotifs et alimentaires des espèces aussi bien que les différences morphologiques qui existent entre elles.

Anderson, N.H. 1979. Bringing live insects into the classroom, p. 47 - 49. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On décrit quatre démonstrations ou expériences de laboratoire servant à initier les étudiants aux activités des insectes: (1) le fonctionnement des branchies physiques, en utilisant des corixidés ou des notonectidés; (2) les propriétés physiques de la pellicule de surface de l'eau, en utilisant les coléoptères staphylinidés *Dianous* et *Stenus*; (3) la mue, en utilisant une importante culture en laboratoire de tipules ou de libellules par exemple, et (4) le comportement et l'alimentation du limnéphilidé larvaire *Clistoronia magnifica* (Banks).

White, D.S. 1979. Utilization of study streams, p. 51 - 55. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On décrit l'expédition annuelle d'entomologie aquatique de l'University of Michigan dans la région de la Pigeon River une innovation apportée par Justin Leonard en 1965. Les données accumulées au cours des voyages précédents (c'est-à-dire les catégories des formes vivantes qui ont été constamment réunies par les classes au cours des 13 années précédentes) servent à préparer les étudiants au voyage et à faire des comparaisons d'une année à l'autre. On visite des sites d'étude semblables plus près d'Ann Arbor pour initier les étudiants avant le voyage à la Pigeon River. La communication énonce les principes directeurs selon lesquels on choisit les ruisseaux pouvant servir à des expéditions d'entomologie aquatique en milieu naturel.

Mackay, R.J. 1979. Winter field work, p. 57 - 62. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On y décrit les observations hivernales en milieu naturel à l'intention des étudiants qui se spécialisent en biologie d'eau douce et en entomologie aquatique, à l'University of Toronto. Les exercices consistent à observer la biocénose hivernale (croissance, alimentation, transformation des feuilles tombées à l'automne, migrations à l'intérieur d'un ruisseau), et à élaborer des dossiers. On y discute d'échantillonnage efficace et de problèmes logistiques comme la détermination des ruisseaux qui conviennent, le nombre optimal d'étudiants à amener, les vêtements d'hiver appropriés et la nourriture.

Hart, D.D. 1979. Association analysis, species interactions, and the structure of benthic invertebrate communities, p. 63 - 71. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

L'interaction entre les espèces joue un rôle dans la structure des biocénoses. On obtient une évaluation élémentaire de l'importance de cette interaction par la quantification des types d'association des espèces. Pour décrire les types de co-occurence relatifs aux associations d'espèces, on expose ici les méthodes qui utilisent à la fois les données se rapportant à la présence ou l'absence et les données relatives à la densité de la population. On fait cas aussi de l'analyse statistique de ces donnés modèles. Des associations significatives peuvent découler soit de l'interaction écologique active soit de l'hétérogénéité de l'habitat. On aura souvent besoins de méthodes expérimentales additionnelles pour établir la différence entre ces explications biologiques possibles. Prises ensemble, ces approches descriptives et expérimentales peuvent fournir une méthode analytique solide pour déterminer le rôle que joue l'interaction écologique dans la formation des sociétés benthiques d'invertébrés.

McGinniss, M.J., and W.J. Trush. 1979. Islands in the stream: an analysis of the species-area patterns of stream rocks, p. 73 - 79. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

La relation région-espèces, une partie de la théorie de la biogéographie insulaire, peut s'étudier en milieu naturel et en laboratoire. Cette étude aide l'étudiant à associer certaines connaissances écologiques à un phénomène observé. On décrit la marche à suivre pour choisir le ruisseau et les roches, enlever les

organismes des roches, évaluer le nombre d'espèces, évaluer la surface des roches et tracer les courbes région-espèces. Une légende simplifiée des classes auxquelles appartiennent les principaux groupes de jeunes insectes aquatiques est incluse. On examine également les causes inhérentes de variation (c'est-à-dire la texture variable de la surface des roches, l'exposition à différentes influences physiques et biologiques dans le ruisseau, les variations de l'échantillonnage). On propose des façons de développer davantage les exercices.

Gottfried, J., and V.H. Resh. 1979. Developing modules for field exercises in aquatic entomology, p. 81 - 93. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

Un cours d'instruction programmé (appelé "module") pour l'étude en milieu naturel a été mis au point par l'University of California, Berkeley. Il permet aux étudiants de faire des expériences réelles en milieu naturel avant d'aborder la théorie dispensée dans les salles de cours. Le module a été conçu pour initier les étudiants de premier cycle en entomologie aquatique à l'échantillonnage benthique, à la diversité des espèces et à l'utilisation des invertébrés benthiques comme indicateurs de la qualité de l'eau. Le module fournit la documentation de base, définit les tâches, pose des questions et permet de vérifier immédiatement les réponses grâce à un corrigé. L'exposé énumère les avantages des modules. On pourrait composer d'autres modules sur des sujets choisis en biologie aquatique et organiser un programme d'échange de modules.

Stewart, K.W. 1979. A module for advanced field exercises in aquatic entomology, p. 95 - 100. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On décrit un module sur l'"Ecology of benthic organisms" pour étudiants de deuxième cycle. Le module leur permet de faire sous surveillance des expériences en milieu naturel et en laboratoire sur l'échantillonnage du micro-habitat en eau courante (tas de feuilles), sur le recensement et la diversité des insectes aquatiques, et d'étudier le cycle trophique ou vital. La structure du module stimule l'innovation, la confiance en soi et la coopération entre les étudiants pour solutionner les problèmes.

Merritt, R.W., K.W. Cummins, and J.R. Barnes. 1979. Demonstration of stream watershed community processes with some simple bioassay techniques, p. 101 - 113. *In* V.H. Resh and D.M. Rosenberg (ed.) Innovative teaching in aquatic entomology. Can. Spec. Publ. Fish. Aquat. Sci. 43.

On expose deux techniques simples de bio-essai pouvant servir à illustrer le développement de biocénoses dans le bassin versant d'un cours d'eau. On peut étudier la transformation des feuilles mortes en utilisant les tas expérimentaux de feuilles dans les ruisseaux, et les sacs à ordures dans les plaines d'inondation et autres sites terrestres. Ces techniques permettent aussi d'étudier des groupes d'invertébrés à des fins déterminées ou la classification des biocénoses, basée sur les adaptations morphologiques et étologiques dans la recherche de la nourriture. L'analyse de la transformation des feuilles mortes et celle du groupe d'invertébrés chargé de l'alimentation se sont révélées des outils précieux pour les classes qui étudient en milieu naturel la fonction et la structure des biocénoses de ruisseau et de celles de plaine d'inondation.