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Nitrogen Cycling in Microcosms and Application to the Biology of the Northern Arm of the Great Salt Lake

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NITROGEN CYCLING IN MICROCOSMS AND APPLICATION TO THE BIOLOGY OF THE NORTHERN ARM OF THE GREAT SALT LAKE

by

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ABSTRACT

Nitrogen cycling was studied in the hypersaline (330 g l^{-1}) north arm of the Great Salt Lake. The lake, particularly the north arm, has a massive accumulation of organic matter from at least 10 cycles in the last 100,000 years from a freshwater to a saline lake, plus the influence of human industry and agriculture in more recent times. The north arm planktonic and attached community consists of principally, in order of biomass: bacteria of at least two genera, *Halobacterium* and *Halococcus;* two algae, *Dunaliella salina* and *D. viridis;* the brine shrimp, *Artemia salina;* and, two species of brine fly, *Ephydra gracilis* and *E. hians* and possibly one more species.

The nitrogen cycle was studied using weekly lake samples and sediment-water microcosms. Results from the lake indicated a high level of organic nitrogen (6-7 mg l^{-1}) as well as undetectable amounts of nitrate, nitrite, and nitrogen fixation. Ammonia was the only detectable inorganic nitrogen form and occurred in the lake only at times of low algal activity or high excretion rates by the invertebrates or high bacterial activity.

Microcosm studies demonstrated that ammonia, nitrate, and urea did not stimulate the bacteria directly but did so only indirectly through increased algal activity. Glutamic acid, an organic form of nitrogen, stimulated the bacteria directly. No nitrification was observed in the microcosms although nitrite was observed when the microcosms were fed nitrate (denitrification).

Based on lake and microcosm studies the algae and the bacteria appear to depend on each other for nutrients. The bacteria appear to use organic matter produced by the algae and the algae use ammonia produced by the bacteria and possibly the brine shrimp. The production of ammonia appears to be the rate limiting step although there is no shortage of other forms of nitrogen in the north arm. Based on aquarium studies, the potential for biomass production of algae and bacteria is much higher than actually observed in the north arm leading to the postulation of two additional factors controlling population; the grazing of the algae by the invertebrates with the excretion of compounds rich in nitrogen; and, the effect of a low habitat temperature and winter cold on the bacteria, reducing their metabolic activities to nearly zero. A preliminary model of the various interactions is presented and some aspects of the various organisms and their metabolism are discussed.

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INTRODUCTION

Physical and Chemical Properties of the Great Salt Lake

The Great Salt Lake in northern Utah has been a subject of interest ever since the first rumors of its existence started to circulate in the early 1700's (Handy and Hahl, 1966). Since then it has been explored, exploited for recreational and mining purposes, and lately its unusual physical, chemical, and biological characteristics have been the subjects of several scientific investigations. This hypersaline lake lies in a semiarid part of the Great Basin and is a remnant of an ancient freshwater lake, Lake Bonneville. The Great Salt Lake is 119 km long, 45 km wide, has a maximum depth of 11.5 m, and is at an elevation of 1281 m (June 1976) (Post, 1975).

The Great Salt Lake is a closed system and is the largest terminal lake in the United States (Steed and Glenne, 1972). This lake is characteristic of closed systems since it is quite sensitive to climatic changes and has a short response time (Grey and Bennett, 1972). Since it is a closed system, the only outflow or loss from the lake occurs through evaporation. Evaporation is affected by the salt concentration and surface area of the lake and also by the climatic conditions surrounding the basin. Inflows to the lake occur primarily from precipitation, surface runoff, and groundwater flows. Of these, surface runoff and precipitation account for the major portion of inflow to the lake. The Bear, Weber, and Jordan Rivers, the Salt Lake City sewage canal, the Kennecott drain, a few smaller streams, and several industrial plant effluents account for the surface water discharge into the Great Salt Lake. In addition, the three rivers previously mentioned carry 60 percent of the dissolved solids that enter the lake (Steed and Glenne, 1972; Arnow and Mundorff, 1972). In an average year, approximately 2.1 billion cubic meters of water and 3.9 million metric tons of dissolved solids are discharged into the Great Salt Lake, but the dissolved mineral inflow, although large, is insignificant compared to the 4 to 6 billion tons of dissolved solids already in the lake brine (Arnow and Mundorff, 1972; Handy and Hahl, 1966). To appreciate more fully the amount of mineral this represents, it has been calculated that the average yearly discharge of dissolved solids in the lake, if precipitated all at once, is enough to cover one square mile to a depth of 2.5 feet or to fill 70,000 standard railroad box cars.

In 1957 a rockfill railroad causeway was completed between Lakeside and Promontory Point by the Southern Pacific Company. This causeway has two 15 foot wide culverts and is supposedly permeable, but since 95 percent of the surface inflow enters the lake south of the causeway, and since the total volume of water removed from the northern portion by evaporation is larger than the annual north arm inflow, there is a net northward migration of salt water through the causeway. This results in the migration of salt, as well as organic and inorganic nutrients, to the northern arm of the Great Salt Lake (Adams, 1964; Jensen and Arnow, 1972; Post, 1975).

Before the construction of the causeway there was a free interchange of water between the northwest body and the main body of the Great Salt Lake, but when the causeway separated these two bodies of brine it severely restricted this interchange and effectively created two ecologically distinct lakes. It is primarily the salt and nutrient migration to the northwest body of the lake that accounts for this ecological difference. The lake north of the causeway is about twice as saline as the southern body and is actually at or near saturation for sodium chloride year around. This makes the northwest body of the Great Salt Lake, with a total dissolved solids concentration of 332,480 mg/l, one of the saltiest bodies of water in the world (Post, 1975). Table 1 lists the results of chemical analyses done on salt water from the northern lake basin.

The precipitated salt which covers the northern lake bottom is 99.9 percent sodium chloride. This contrasts with the sand and algal bioherms which dominate the southern basin (Handy and Hahl, 1966). Salt solubilities, however, are affected by temperature changes and this will control the type of salt that is found precipitated on the bottom. During the winter months in the northern lake basin, a jelly-like precipitate of sodium sulfate ($Na_2SO_4 \cdot 10H_2O$) replaces the more familiar sodium chloride precipitate as its lower solubility limit is reached (Handy and Hahl, 1966). The high salt concentration (Table 1), coupled with the nutrient load, produces a highly saline and highly enriched

Table 1. **Chemical composition of unfiltered brine in the northern arm of the Great Salt Lake.**

Chemical	Concentration
Chloride[a]	177,600 mg/l
Magnesium[a]	10,400 mg/l
Calcium[a]	300 mg/l
Sodium[a]	101,450 mg/l
Potassium[a]	4,140 mg/l
Sulfate[a]	21,540 mg/l
Lithium[a]	58 mg/l
Boron[a]	35 mg/l
Aluminum	4.00 mg/l
Antimony	0.01 mg/l
Arsenic	0.22 mg/l
Barium	4.25 mg/l
Beryllium	< 0.01 mg/l
Cadmium	0.17 mg/l
Chromium	0.12 mg/l
Cobalt	0.01 mg/l
Copper	0.23 mg/l
Iron	22.51 mg/l
Lead	0.53 mg/l
Manganese	1.33 mg/l
Mercury	0.01 mg/l
Molybdenum	0.05 mg/l
Nickel	0.01 mg/l
Silica	6.45 mg/l
Silver	0.03 mg/l
Thallium	0.02 mg/l
Titanium	0.05 mg/l
Tungsten	< 0.01 mg/l
Vanadium	2.35 mg/l
Zinc	0.50 mg/l
Bromide	0.15 mg/l
Fluoride	15.90 mg/l
Carbonate	270.00 mg/l
Bicarbonate	454.00 mg/l
Iodine	0.00 mg/l
Selenium	0.35 mg/l
Orthophosphate[b]	290-860 μg/l
Total phosphorus[b]	630-1260 μg/l
Nitrate[b]	0 μg/l
Nitrite[b]	0 μg/l
Ammonia[b]	0-925 μg/l
Oxygen[b]	0-1.7 mg/l
Specific gravity (field)[b]	1.200-1.230
pH	7.7
Total solids	332,480 mg/l

[a]Utah Geological Survey, June 1975.

[b]Ranges of 1974-1975 Utah State University; others from samples taken August 1974 and analyzed by Ford Laboratories, Salt Lake City, Utah.

(but unpolluted) condition in the northern portion of the lake. This extremely harsh environment results in quite a low diversity of aquatic life. In a situation like this where competition is restricted, the few forms present are able to proliferate greatly. This is seen in direct microscopic bacterial counts from the northern portion of the lake which often exceed 50 million per milliliter of water.

Biology of the Northern Arm of the Great Salt Lake

The organisms found in the northern arm of the lake clearly illustrate the low diversity of life adapted to this unique environment (Table 2). In addition to these organisms, several protozoa (three flagellates, an amoeba, and a ciliate) have been observed in laboratory maintained systems, but not directly in the lake itself. Protozoa have been reported from the southern arm of the lake prior to separation of the lake by the causeway, but have not been previously reported from the extremely saline northern arm. Of the primary producers, the red algal species is the most common planktonic form while the green alga primarily colonizes surfaces. The algae usually bloom in early summer with numbers reaching 11×10^3 per milliliter of water, the red alga being the dominant form. These algae serve as the primary food source for the brine shrimp whose populations reach a peak in August. The other arthropod found in the lake, the brine fly, spends its egg, larval, and pupal stages in the water. The larval stage grazes actively on algae and dead brine shrimp.

The halophilic bacteria living in the brine are the decomposers of the system. They contain a red carotenoid and a purplish rhodopsin-like pigment and are so numerous that they impart a distinct wine-red color to the water. These bacteria live in close association with the red and green algae. They utilize secretions from these algae as well as excretions from brine fly larvae and brine shrimp and also metabolize organic matter from dead brine shrimp, brine shrimp fecal pellets, dead algae, dead brine flies, and other dead bacteria.

Table 2. **Organisms of the northern arm of the Great Salt Lake (Post, 1975).**

Organism	
Scientific Name	Common Name
Dunaliella salina	Red alga
D. viridis	Green alga
Ephydra gracilis	Brine fly
Artemia salina	Brine shrimp
Halobacterium-Halococcus	Halophilic bacteria
Halophages	Bacterial viruses

The northern basin of the Great Salt Lake thus becomes an interesting subject for study since so little is known about the biology of hypersaline ecosystems. This terminal lake drains a huge watershed that acts as a source of human, industrial, and agricultural wastes. As population growth continues to increase and the demand for water for various uses increases, the question will become whether the Great Salt Lake is used as a water resource or as a waste sink. Due to the salt water migration described earlier, waste materials may eventually be deposited in the northwest body of the lake. Whether these materials are cycled, lost from the system, or whether the lake is acting as a nutrient sink are important questions. Answers to these questions would help to predict the effects of man's domestic, industrial, agricultural, and recreational activities on the Great Salt Lake. In addition, information on the basic biology of this unique system could lead to a better understanding of the biology of other hypersaline ecosystems in the world such as Mono Lake in California, the Dead Sea in Israel, Lake Rezaieh in Iran, Lake Assal in French Somaliland, and lagoons and some estuaries on the edges of continents.

It was the goal of the research described in this report to determine the fate of various sources of nitrogen in the northern portion of the Great Salt Lake and thereby elucidate the cycling mechanism of nitrogen as it exists in this basin.

BACKGROUND

Microcosms

Natural ecosystems

Ecosystems have been described as the basic functional unit in ecology (Odum, 1971). Whittaker (1970, p. 1) defines an ecosystem as "a community and its environment treated together as a functional system of complementary relationships, and transfer and circulation of energy and matter." Since environmental studies have recently been focusing more on the functional aspects of the environment rather than on the traditional structural descriptions, it is only logical that ecosystems, as units, be the focal point for these investigations.

Ecosystems are composed of both biotic and abiotic components which are involved in nutrient cycling, energy transfer, successional changes, and other processes. Natural ecosystems are usually characterized as being large, subject to many variables which are not static but are in a constant state of flux, and as having indefinite boundaries that gradually grade into one another (Beyers, 1964). Possessing information concerning individual functional aspects of an ecosystem is important in gaining an understanding of the entire system, but the characteristics of a natural ecosystem make obtaining such information extremely difficult. A researcher must be able to account for variables in the system and run controls in conjunction with the system being studied so that sources of error are minimized and all the factors influencing the study are taken into consideration. Unmanageable size, variables unaccounted for, and indefinite boundaries make this difficult in the field, so it is desirable to devise a system which can be worked with under controlled laboratory conditions. The microcosm approach to ecosystem analysis is such a system.

Laboratory microcosms

Abbott (1966) defines a microcosm as a miniaturized ecosystem. Microcosms, or microecosystems, can either be laboratory models or natural systems where size, variables, and boundaries do not pose problems to the investigator. Stephen Forbes (1925), in his classic paper, describes the ecology of a lake using the natural microcosm approach. Probably the first paper on the use of laboratory microcosms was given before the Chemical Society of London on March 4, 1850, by Robert Warington (Beyers, 1964). Warington's paper dealt with a balanced aquatic microecosystem which he had established in an aquarium. Since then, researchers from many disciplines have found practical applications for the microcosm approach and have utilized a myriad of experimental designs.

Advantages of microcosms

The microcosm approach to ecosystem analysis has many advantages. Among these are manageable size, defined boundaries, controlled variables, controlled complexity, replicability, reproducibility, and simulation of the mixed culture condition of the natural system (Beyers, 1964; Nixon, 1969a; Wilhm, 1970; Odum, 1971; Taub and Pearson, 1974). As was mentioned previously, analysis of ecosystems is difficult since in nature they generally are large and grade into one another. One way to avoid this problem is to isolate the ecosystem as a functional unit from the rest of the biosphere by containing it within the defined boundaries of a microcosm.

Continuously shifting variables are also characteristic of natural systems. Experimental work, however, requires control of these variables and the use of microcosms can alleviate this problem by enabling the researcher to account for, and properly manipulate, the variables.

Microcosms can also be replicated for statistical analysis. This condition is extremely rare in nature, but divergence among laboratory microcosms can be minimized by cross-seeding between the systems (Beyers, 1964). Since control of the system is possible, it is also feasible to reproduce the system being worked with at a later date. This may be important to investigators who wish to test an hypothesis on another's system.

The last point to be considered is that ecological processes are not simply composites of the biochemical characteristics of individual pure cultures, but are the results of interactions between genera, species, and strains. Microcosms, when inoculated with the desired seed, provide a method

for studying the mixed culture condition of a natural system in a manageable, controlled environment.

Criticisms of microcosms

As well as having several advantages in the study of ecosystems, the microcosm approach to ecosystem analysis also has its disadvantages. Taub and Pearson (1974) in their review of the uses of laboratory microcosms state that criticisms of this approach are based on three questions: (1) Can results from microcosm research be applied to the actual environment? (2) Is a given microcosm an adequate model of the ecosystem being simulated? and (3) Is replication possible and, if so, is it statistically valid? Beyers (1964) also recognizes the criticisms of the microcosm approach and feels that the chief criticism is in the differences that exist between the microcosms and the natural system. He believes that the investigator must assume that there are similarities between the micro and macro ecosystems if he is going to use the experimental microcosm to understand the natural macrocosm. Even though the experimental system does not exactly duplicate the natural system, it is the level of similarity between the two that is important. In other words, it is correct to assume that the study of certain processes within the microcosm will lead to a clearer understanding of the same processes in the natural ecosystem.

Justification of the microcosm approach–answers to criticisms

Gordon et al. (1969, p. 99) compared 10 characteristics of natural ecosystems to the same characteristics in laboratory microcosms to determine whether or not the microcosms would follow the expected trends. They found that data from aquatic microcosms "support the premise that a microcosm can exhibit trends and characteristics similar to those of larger natural systems" and also stated that "results of this study illustrate the usefulness of the simple, aquatic microcosm as a laboratory tool for the study of other ecosystem attributes and for examination of ecological methods and concepts."

In their study of the transfer of minerals in consumer-decomposer microcosms, Witkamp and Frank (1970, p. 473) state that "the microcosm provided a realistic medium from which results could be related to the results of other simple laboratory experiments as well as to measurements in the field." The applicability of microcosm results to the natural environment was also shown by Ferens and Beyers (1972) in their study of the effects of stress (gamma radiation) on communities within laboratory microcosms. Their study indicated that, as in natural ecosystems, stress could change the species dominants and successional patterns within microcosms. This seems to be in agreement with other investigators (Menzel et al., 1970) who made observations of the effects of stress on communities in their natural environment.

Nixon (1969a, 1969b) in both of his microcosm studies found that patterns observed in the microcosms agreed well with expected trends in the natural environment. He concluded that the use of microcosms is a feasible approach for studying ecological processes. This same conclusion is also shared by Porcella et al. (1975) who found the microcosm technique to be a very sensitive method of analyzing microbial dynamics in sediment-water systems. Their system allowed gas, water, and sediment analyses to be performed routinely and also allowed interpretation of some of the major factors controlling productivity in the ecosystem being simulated.

The applicability of results from microcosms to the natural environment and the adequacy of the microcosm as a model of the natural system are indeed two factors which must be carefully considered when utilizing this approach to ecosystem analysis. Taub and Pearson (1974) feel that results from microcosm research are more applicable to the natural environment if the researcher knows the natural ranges of the variables affecting the simulated system and then runs the microcosms under controlled conditions that fall within these ranges. The less the microcosm conditions simulate natural conditions, the more difficult it becomes to extrapolate the results.

Replicability of microcosms for valid statistical analysis is the last of the criticisms to be dealt with. Since this has been one of the major points in justifying the use of microcosms, the validity of this premise is obviously crucial. Abbott (1966, p. 268) established 18 five-gallon carboy microcosms and then monitored several parameters within them to determine the degree of divergence among the replicate microecosystems and thus the replicability of this approach. Abbott concluded that "under proper conditions, aquatic microcosms show replicability comparable to that found among replicated units in various other types of statistical trials." This shows that controlled microcosm experiments are replicable and the use of replicability as a premise for justifying this approach is a valid one. The same conclusion was reached by Beyers (1963) in his replicability studies on 12 aquatic microcosms. In addition, Taub and Pearson (1974, p. 9) state that "where investigators have sought to measure microcosm reproducibility in mixed communities, they have found satisfactory reproducibility in community properties." In other words, the research performed with microcosms has shown that the criticisms concerning their use in ecosystem analysis

can be satisfactorily answered and has also shown that the premises justifying this approach are valid.

Types and uses of microcosms

No standard microcosm design exists. In fact, a multitude of types have been designed by investigators to meet the requirements of the problems being studied. The types of microcosms employed can be separated either on the basis of the habitat being simulated or the source of the biological components within the system. Based on habitat, there can be terrestrial, terrestrial-aquatic, river, aquatic batch culture, aquatic continuous culture, and naturally occurring microcosms (Taub and Pearson, 1974). In terms of biological components there are two basic types of microcosms: those with cultures derived directly from nature and those whose cultures are built up from axenic cultures (Odum, 1971). In the latter case the microcosm is said to be gnotobiotic. Microcosms with cultures derived from nature are usually used to simulate some specific outdoor situation while the gnotobiotic approach is used to study nutritional and biochemical aspects of a single species or interactions of two or more species. In general, microcosms are most often utilized in studying transfer rates, turnover rates, bioaccumulation, biodegradation, productivity, biogeochemical cycles, toxicity, and theoretical ecology (Beyers, 1964; Taub and Pearson, 1974).

The most frequently used microcosm approach, according to Taub and Pearson (1974), is the batch culture technique. This method differs from continuous culture in that it does not employ a constant addition of nutrient media and discharge of cells and waste products. The batch culture method can be modified, however, to allow for a periodic (but not continuous) addition of media and removal of effluent. Such a semi-continuous or periodic media replacement routine was utilized by Porcella et al. (1970, 1975) in their microcosm modeling of an aquatic ecosystem.

The large variety of types of microcosms is a direct result of the specifications for experimental design required by the study itself as well as the investigator's imagination. Microecosystems have been established in test tubes, flasks, carboys, aquariums, beakers, fiber glass tanks, plastic tanks, and lucite cylinders. Open systems, closed systems, axenic cultures, mixed cultures, freshwater, salt water, aerobic environments, and anaerobic environments have all been incorporated in batch culture microcosm designs. With these designs, the batch culture method has been successfully employed in studies concerning nutrient cycles, volatile chemicals, detritus feeders, bio-accumulation of pesticides in aquatic habitats, and the degradation or modification of specific chemicals in the environment.

Cooke (1967) studied autotrophic succession in a farm pond culture by placing pond samples in beakers and measuring community metabolism, biomass, and chlorophyll concentrations. His results agree well with what is known about the metabolism and stability of immature and mature communities in other successional systems.

Succession was also studied by Gordon et al. (1969), but their emphasis was on heterotrophic succession in bacterial cultures. These cultures were maintained in flasks. The results of this study established that bacteria were functional (1) as contributors of thiamin, CO_2, and possibly nitrogen to primary producers; (2) as primary consumers of algal excretions; (3) as secondary consumers of bacterial excretions; and (4) as decomposers.

Cooper and Copeland (1973), Cooper (1973), and Ramm and Bella (1974) all used the microcosm approach to study various aspects of the natural environment. They studied hydrological conditions in estuarine areas, the role of herbivore grazing intensity as a factor affecting net primary productivity, and sulfide production in tidal flats, respectively. These investigators all reported a high degree of similarity between their models and the natural systems and concluded that their results led to a clearer understanding of the same processes in the natural environment.

Other investigators have used microcosms to study the effects of particular chemical and physical variables on natural ecosystems. Porcella et al. (1970) used cylindrical lucite microcosms to study the sediment-water exchange of phosphorus in waters where phosphorus was the limiting nutrient. They found that sediments could act as a source of phosphorus for algal growth in low phosphorus waters. The amount of phosphorus released from the sediment depended upon the amount initially present in the sediment and also upon the development of *Oscillatoria* mats which resulted in an increase in the transfer of phosphorus to the water phase.

The role of phosphorus in lake water was further studied by Mitchell (1971). He used microcosms to test the eutrophication of water by phosphates in activated sludge and found that non-phosphate detergents that were added to the sludge did not differ in eutrophying potential from phosphate detergents.

The role and fate of nitrogen, mercury, and iron in aquatic ecosystems was investigated by Porcella et al. (1975) using 16 sediment-water microcosms. These were semi-continuous systems in which nutrient, lighting, and temperature conditions were controlled and from which gas and water samples could be taken for analysis. Nitrogen and mercury

were studied at limiting concentrations while phosphorus and iron were studied at low concentrations as affected by interactions with nitrogen and mercury. This comprehensive study of materials balances in an aquatic environment found the microcosm approach to be an excellent method for studying the role and fate of various chemicals in an ecosystem.

Beyers (1962) and Allen and Brock (1968) tested the effects of temperature on heterotrophic mixed cultures in microcosms. They found that (1) the metabolism of a single organism is more dependent on temperature than is the metabolism of the entire community; (2) the more stable the community is, the more independent its metabolism will be from temperature changes; and (3) temperature alone can select for, and control, the kinds of organisms which develop.

Microcosms can also be adapted to radioactive tracer studies as shown by Whittaker (1961) and Wilhm (1970) in their tracer experiments on aquatic systems. A comparison of oligotrophic and eutrophic systems showed a rapid movement of radio phosphorus tracer out of solution in both, this movement being more rapid in the oligotrophic system.

Bioaccumulation, biodegradation, and toxicity assays are further applications of the microcosm approach to ecosystem analysis. Sanders and Chandler (1972) studied the biological magnification of polychlorinated biphenyls (PCB's) by aquatic invertebrates. The magnification of a specific PCB (Aroclor 1254) did take place quite rapidly in some organisms and resulted in high concentrations of this chemical in the tissues.

In another bioaccumulation study, Filip and Lynn (1972, p. 254) monitored mercury accumulation by the freshwater alga *Selenastrum capricornutum* to find how mercury may enter the base of the aquatic food web. The test organism was grown axenically in pyrex culture flasks. It was found that mercury uptake by live algal cells, whether in light or darkness, was not significantly higher than uptake by killed algal cells. This indicated that the mercury uptake was by passive absorption. They concluded that "algae must therefore be considered to constitute a point of possible entry of mercury into the aquatic food web at the lowest trophic level."

The degradation of DDT by microorganisms *in vitro* was investigated by Pfaender and Alexander (1972). They found that DDT can be extensively degraded by the enzymes of a single microorganism (*Hydrogenomonas* sp.) under anaerobic conditions.

The toxicity of PCB's, DDT, and dimethyl mercury have also been studied in microcosms. Morgan (1972) tested the effects of a PCB and DDT on cultures of an alga, protozoan, daphnid, ostracod, and guppy. All were found to be affected to some degree by either the PCB, DDT, or both at concentrations of 2 and 20 ppm. Kolb et al. (1973) used beakers, glass tubes, and partitioned, aquarium-like containers as microcosms to study volatilization rates, uptake kinetics, toxicities, and equilibrium concentrations of dimethyl mercury in algae, Daphnia, and fish. They found a low toxicity of dimethyl mercury to test organisms, increased volatilization with increased temperature or stirring, and an independence of concentration factors from temperature for all three test organisms.

These examples of the uses of microcosms show at least partially the multitude of applications that this approach has to ecosystem analysis. In some of these applications of the microcosm approach (Ramm and Bella, 1974; Porcella et al., 1970; and Porcella et al., 1975), the involvement of sediments in chemical and biological processes was accounted for by the inclusion of sediment in the microcosm designs. Several investigators, however, did not account for this important aspect of aquatic ecosystems and this may represent a source of error in their results.

Hypersaline Water Chemistry

Environmental significance of salt

Salts are important to man and his environment for several reasons. They are basic components of natural systems and as such may be required as essential nutrients by animal or plant life. Also, they are mined as basic raw materials and sometimes can be considered as pollutants of surface and underground waters (Martinez, 1971).

Where high concentrations of salts exist in natural waters, a very harsh environment with unique physical, chemical, and biological characteristics is established. The physical and chemical properties of such water have interesting effects on the biological components in the system as well as on chemical analyses performed on the water. Blinn (1971) reported seasonal shifts of major cations and anions in concentrated saline habitats due to selective precipitation of salts and changes in pH which were probably due to uptake of carbon dioxide by algae. The phenomenon of salt precipitation was also considered by Copeland (1967). He felt that the precipitation of salts could be very significant to any organism requiring them. With these minerals no longer in solution the organisms would be forced to either colonize the sediment surface or else face a serious survival problem.

Copeland and Jones (1965) studied community metabolism in hypersaline ecosystems and found both community metabolism and species diversity to be quite low. All photosynthesis/respiration ratios were low and it was noted that the metabolic characteristics of these communities were similar to those of polluted environments.

Copeland (1967) discusses several physical and chemical characteristics of hypersaline habitats. Included are the effects of hypersalinity on species diversity, differential ion precipitation, pH, dissolved oxygen, and temperature. He notes that an increase in the salinity of a system is usually accompanied by a decrease in species diversity. There are several factors besides osmotic stress, however, that may account for this reduction.

The theory of constant proportions says that ionic concentration varies with dilution, but the ratio of one ion to another remains the same. However, as salinity increases, differential precipitation of salts occurs depending on the concentration of the salts and their solubilities. This can further be complicated by the fact that the solubility of a given salt is different in the presence of other salts and this may lead to chemical interactions between two or more ions with the resultant separation of double or triple salts. As previously mentioned, this can affect organisms that require specific ions in the solution.

Closely related to differential ion precipitation is alkalinity. Since calcium carbonate is one of the first salts to precipitate with an increase in salinity, its removal would change the concentration of carbonate in solution as well as the carbonate-biocarbonate equilibrium. This in turn would affect the pH of the system since the pH increases with salinity until just before saturation with calcium carbonate and then decreases as carbonate precipitates.

The solubility of oxygen in water decreases with decreased pressure, increased temperature, or increased salinity. Formulas developed by previous investigators enabled calculations to be made of solubilities of oxygen in seawater relative to salinity and temperature, but only from 0-4 percent salinity. Extrapolation led to zero oxygen in water above 17 percent salinity. This is obviously incorrect since brine shrimp can live in greater salinities. Experiments showed that saturation values formed a curvilinear relationship that tended to level off at 22 percent salinity.

The specific heat of water is also influenced by salinity. An increase in salinity leads to a decrease in specific heat (< 1). Thus, air temperature fluctuations result in relatively dramatic water temperature fluctuations in hypersaline waters. This would be an added hardship to organisms existing in an already harsh environment.

Significance of salt to water chemistry

Several researchers have experienced problems when they have tried to run routine chemical analyses on hypersaline waters. Stephens (1974) reported problems with the chemical analysis of heavy brines from the Great Salt Lake. A salt error produced inconsistencies with potentiometric pH determinations and required that dilutions and standards be prepared from an artificial salt water medium.

Nixon (1969a) also reported problems with chemical analyses of hypersaline water and concluded that these were due to the large differences between ionic activities and concentrations, as well as to differential ion precipitation. Like Stephens (1974), he prepared standards from salt water, but differed in that he did not dilute his samples. No salt error was reported in these undiluted samples. However, Nixon's standard curves were constructed from salt water standards containing only sodium chloride. Stephens' standards were prepared using an artificial medium representing 99 percent of the minerals existing in the sample water. A salt error resulting from the presence of the total mineral content may have required Stephens' dilutions and represents a possible source of error unaccounted for by Nixon.

Strickland and Parsons (1972) reported that even the 3.5 percent dissolved salts in ocean water gives rise to many analytical difficulties. They add, however, that the relative constancy of an ionic environment enables one analytical method to be applied to other samples of similar ionic constancy.

The problems encountered with the chemical analysis of hypersaline water can be explained in part by ionic activities and molecular structures existing in the aqueous phase. According to Davies (1962), a salt effect on ion associations may lead to altered activity coefficients. This is because the activity coefficient of an ion complex may be altered by an asymmetrical charge distribution not accounted for in the simplified assumptions of the Debye-Hückel theory. This is in agreement with Polzer and Roberson (1967) who state that ion activity products are more useful in the prediction of soluble-salt precipitation than solubility products based on apparent concentration alone. This is because the precipitation depends primarily upon the ionic strength of the water, the nature of the solute complexes, and the concentration of the solutes. Because of the formation of complex ions and because of the difficulty in determining activity coefficients of individual species, a direct reading of ionic activity by a specific-ion

electrode would not be possible in waters with a high dissolved solids content. With a specific-ion electrode there is an assumed constancy of the liquid junction potential between the unknown and the calibrating standard which could be quite in error in complex brine systems.

The question of activity coefficients versus ionic concentrations was also taken up by Nelson (1975) who stated that activity coefficients, which are related to the total ionic strength of all ions in solution, are significantly depressed in solutions with large numbers of ions. When he diluted 16.5 grams of sodium chloride to a liter (10,000 mg/l chloride) he found that the chloride activity coefficient was depressed to 65 percent. Since the concentrations of the various ions being studied in the hypersaline water of the Great Salt Lake were quite low (< 1 mg/l) and since the ionic strength of this water was so large, it is possible that the activities of the ions being measured are depressed to a point where the amount reacting is well below the sensitivity of the test or else is not reacting with the reagents at all.

Samoilov (1965) reported that regions of different chemical structure can exist within a concentrated solution. Such a solution may have nonhomogeneous structures which, when diluted, retain a structure similar to water, but when in concentrated form resemble solid hydrates. Thus, increased concentration leads to the formation of structural inhomogeneity within the solution. For dilute solutions the solution structure is determined by water while that of concentrated solutions is determined by the crystal hydrate. A temperature-composition diagram which shows how the actual structural composition of a solution changes with variations in temperature and/or salt concentration was presented by Kaufmann (1960).

Thus, it is obvious that concentrated brine solutions pose problems not only to the organisms that must live in them, but also to the researcher who desires to quantitatively analyze them. The ionic activity coefficients, rather than concentrations and the structural peculiarities of concentrated brine solutions make this a difficult task.

Nitrogen Metabolism in Halophilic Organisms

Salt requirement

Larsen (1967) and Kushner (1968) in their reviews of halophilic bacteria point out that these organisms are not only adapted to living in concentrated brines, but actually have specific requirements for high concentrations of certain ions. These ions (K^+ and Mg^{++} in particular) are required for enzyme activity, ribosome stability, and cell envelope integrity. The absence of these essential ions results in the inactivation of enzymes, dissociation of 70S ribosomes into 31S and 52S fragments, and electrostatic repulsion within the envelope which causes cell lysis. The mechanism of salt action in preventing these from occurring is thought to be based on charge shielding and salting out phenomena. Baxter (1959) proposed that salts shield ionic charges on enzymes by providing counter ions and thereby reduce electrostatic repulsion from like charges. This allows hydrogen bonds to form giving stability to the enzyme's conformation.

The salting out of nonpolar side chains on enzymes in the presence of high concentrations of sodium chloride was reported by Lanyi and Stevenson (1970). This salting out apparently moves these side chains to the interior of the structure where they can form stable hydrophobic bonds and add to the overall stability of the enzyme.

Lieberman and Lanyi (1972) studied the effects of high salt concentrations on the action, stability, and allosteric properties of threonine deaminase in *Halobacterium cutirubrum.* Their findings on the role of salts in the activity and stability of enzymes from halophilic bacteria agreed with previous investigators. They also found subtle conformational changes in allosteric enzymes from halophilic bacteria in high salt concentrations and postulated that the salt requirement for the stability of many halophilic enzymes may have some common structural origin.

Stevenson (1966) reported a specific requirement for sodium chloride (4-5 M) for the uptake of L-glutamate by the halophilic bacterium, *Halobacterium salinarium.* When sodium chloride was replaced by isomolar concentrations of potassium chloride, sodium acetate, or potassium acetate, only negligible uptake of the amino acid occurred. This study also showed that since, in the presence of sodium chloride, glutamate in cells is in a free state and at a concentration of at least 50 times that in the medium there is also a specific requirement of sodium chloride in the function of a preformed mechanism of active transport.

A requirement for high concentrations of sodium chloride for growth and photosynthetic glycerol production in the unicellular halophilic alga, *Dunaliella parva,* was reported by Ben-Amotz and Mordhay (1972). This study also showed, however, that isomolar concentrations of glucose and glycine could replace sodium chloride for oxygen evolution. This function in the alga, unlike other functions which had a specific requirement for sodium chloride, had a general high isomolarity requirement.

Organic nitrogen

The metabolism of organic sources of nitrogen by halophilic organisms has been studied by a number of researchers interested in such things as the biochemistry of unique organisms, the spoilage of salt-cured materials, and the culturing of halophilic organisms. Shah and DeSa (1965) used gelatin-salt agar, milk-salt agar, and fish muscle protein-salt agar media to study proteolytic activity in 217 cultures of halotolerant and halophilic bacteria. Proteolysis was exhibited by 88 of the cultures: 26 hydrolyzed gelatin only, 31 hydrolyzed gelatin and casein, and 31 hydrolyzed all three proteins. Obligate halophiles did not seem capable of attacking fish muscle protein although some hydrolyzed casein and gelatin. Most fish muscle protein hydrolyzers were marine halotolerants.

Norberg and Hofsten (1969) stated that extremely halophilic bacteria would be expected to be proteolytic since they occur so often in protein rich salt brines. These investigators found that some strains of *Halobacterium* form a true extracellular proteinase and reported that proteinase activities were higher in the presence of sodium chloride than potassium chloride and were also dependent upon divalent metal ions. The assumption that there must be an enzymatic complex responsible for amino acid metabolism in some halophilic bacteria was further supported by Gutiérrez and González (1972) and Kamekura and Onishi (1974) in their studies on proteolytic activity in these organisms. They reported the presence of proteolytic activity in both extreme and moderate halophiles.

The culturing of halophilic organisms can be a difficult task due to their complex biochemical requirements. Several investigators, however, have developed artificial media for this purpose. Dundas et al. (1963) composed a chemically defined medium for *Halobacterium salinarium* which also supported the growth of several other halophilic bacteria. Growth rates in this synthetic medium, however, were not as great as in a more complex medium. They found that doubling the concentration of any single amino acid did not affect growth rates, but the omission of them led to an inability of the medium to support growth or else led to poor growth. This established that amino acids are nutritionally important to several halophiles.

Onishi et al. (1965) developed a synthetic medium for halophilic bacteria that produced just as good growth as a more complex medium for several species. Addition of L-glutamine or DL-asparagine caused a marked stimulation of growth. Glutamine produced the best yields and, at higher concentrations, stimulated early growth. Four amino acids (arginine, leucine, lysine, and valine) were found to be essential, six were stimulatory, and eight were inhibitory or without effect. The absence of 18 amino acids resulted in no growth. This supported the findings of Dundas et al. (1963) that amino acids are important to several halophilic species.

Onishi et al. (1965) also tested the stimulatory effect of amine versus ammonium nitrogen on halophilic species. The stimulatory effect of ammonium chloride was greater than asparagine and it was found that ammonium chloride could be substituted for asparagine or glutamine and is preferable in simpler media.

Dundas (1965) was able to grow *Halobacterium salinarium* on a synthetic medium containing free amino acids. Like Onishi et al. (1965) he found that addition of glutamine to the medium stimulated growth, but also found that six amino acids (lysine, arginine, valine, methionine, isoleucine, and leucine) rather than four were essential. It must be noted, however, that these two sets of investigators were working with two different species of *Halobacterium* which may have had differing nutritional requirements.

Gochnauer and Kushner (1969) contend that the synthetic media developed for halophiles by Onishi et al. (1965) and others were limiting to growth because of low potassium concentrations. As was previously mentioned, extreme halophiles require high concentrations of potassium for ribosome stability and for protein synthesis, so limiting amounts of this ion may indeed retard growth. Previous studies also suggested that vitamins do not stimulate the growth of extreme halophiles when added to amino acid media, but Gochnauer and Kushner found that when potassium is not growth limiting, vitamins, as well as carbohydrates, stimulate growth.

Biochemical properties of 49 strains of extreme halophiles were examined by Gibbons (1957) in 15, 20, 25, and 30 percent salt. Some of the biochemical reactions, such as gelatin and casein hydrolysis, varied with salt concentration while others occurred only at specific salinities. All of the organisms tested were negative for urease. This is surprising since urea can be a significant source of nitrogen in aquatic environments (Carpenter et al., 1972).

Crane (1974) in his characterization of extremely halophilic isolates from the Great Salt Lake reported 40 percent of them to be positive for urease and 76 percent positive for gelatin hydrolysis. It is interesting to note that Crane's lake isolates were apparently able to produce their own amino acids since a specific test medium that contained only the appropriate inorganic nitrogen source, glucose, agar, and salt solution was able to support growth. The

halophilic cultures used by previous investigators which required specific amino acids for growth may have lost the ability to synthesize them by being cultured for generations in complex media.

Inorganic nitrogen

The metabolism of inorganic sources of nitrogen by halophilic organisms has been an interesting and profitable area of research for those engaged in studying these unique organisms.

Campbell et al. (1961) showed that ammonium is required for the growth of certain halotolerant organisms and if not supplied is obtained by the deamination of glutamic or other amino acids. Research by Onishi and Gibbons (1965) has shown ammonium to have a stimulating effect on the extreme halophile, *Halobacterium cutirubrum.* They found that the stimulating effect of ammonia is not due to a specific requirement for ammonia itself, but rather is related to the utilization of amino acids. When glutamic acid was omitted from the basal medium and the threonine concentration was reduced, ammonium inhibited growth, but when only threonine was omitted, growth was stimulated markedly by addition of ammonium chloride. Ammonium apparently stimulates amino acid metabolism in this organism.

The effect of cations on the stability of a halophilic nitrate reductase from a Great Salt Lake isolate, *Halobacterium* sp., was studied by Marquez and Brodie (1973). They found that increased salt concentrations raised the temperature at which maximum nitrate reductase activity occurred. Also, nitrate reductase activity was increased up to a point in a sodium chloride solution with the addition of magnesium chloride, but decreased after the sodium chloride concentration exceeded this level. Protection against heat inactivation was greatest at 4.27 M and 5.31 M sodium chloride. These investigators concluded that at higher salt concentrations the conformation of the enzyme is better suited for optimal activity in the presence of monovalent rather than divalent ions.

Bienfang (1975) investigated interactions between nitrate and ammonium kinetics in a nitrogen limited population of the marine phytoplankter, *Dunaliella tertiolecta.* The simultaneous assimilation of nitrate and ammonia by the phytoplankter indicated the absence of ammonium suppression of nitrate uptake under these limiting conditions. When available together, ammonium is assimilated preferentially to nitrate, but nitrate storage inside the cells appears to continue while cell synthesis continues preferentially on ammonium.

Orthophosphate and inorganic nitrogen are the usual nutrients of primary interest in algal growth, especially with regard to limiting factors. From chemical analyses and bioassays, Porcella and Holman (1972) concluded that inorganic nitrogen is the limiting factor in primary production in the southern end of the Great Salt Lake where the salinity is approximately one-half that found in the northern basin. They postulate that release of ammonia by bacterial decomposition of organic nitrogen in the sediments cycles nitrogen in this system. This release of ammonia would be significant since there is little input of inorganic nitrogen to the lake.

Crane (1974) in his characterization of bacterial isolates from the north arm of the Great Salt Lake reported that 64 percent of the isolates were able to use ammonia and 71 percent were able to use nitrate as their sole source of nitrogen. Of those isolates able to reduce nitrate to nitrite, several were capable of reducing the nitrite and four of these have been shown to produce nitrogen gas. Gibbons (1957) also showed nitrate reduction by extreme halophiles. Some of his strains reduced nitrate at several salt concentrations while others varied in their response to different salinities. Nitrogen fixation tests run on Great Salt Lake isolates in laboratories at Utah State University have all been negative.

It can be seen from these examples that inorganic as well as organic sources of nitrogen are involved in the metabolism of halophilic organisms. Both forms must be accounted for if a complete understanding of the metabolism of a hypersaline community is to be reached.

Previous Hypersaline Ecosystem Studies

Studies on saline systems

Life which exists in the extremely harsh conditions of hypersaline habitats has attracted the interest of physiologists and ecologists who wish to study the unique organisms adapted to these habitats and the interactions that these organisms have with their unique environment. Saline systems are also studied because they lend themselves so well to comprehensive ecosystem analysis (Walker, 1973). Because of their low species diversity and relatively simple trophic structure, the harsh environments found within saline systems make the study of ecosystem structure and function considerably easier than similar studies in more complex systems. A simple natural system lends itself quite well to the holistic concept of ecosystem analysis since a less complex system has fewer variables to be accounted for and minimizes the difficulties encountered in studying complex ecosystems. Habitats such as these are found

around the world and include lagoons, salt marshes, salt evaporating ponds, and brine lakes.

Carpelan (1957) studied the hydrobiology of commercial salt ponds near San Francisco Bay. He measured several physical and chemical properties and described the flora and fauna in six of these ponds. Salinities ranged from that of brackish bay water to about four times that of sea water. He described a general decrease in diversity with increasing salinity and a succession of dominant types with changing salinity. A similar study was carried out by Hedgpeth (1967) on the Laguna Madre, a hypersaline estuary in Texas. The effects of salinity and temperature on species diversity, oxygen production, photosynthesis, respiration, and biomass were determined and corresponded well with Carpelan's findings.

Mason (1967) studied the physical, chemical, and biological aspects of Mono Lake in California. This lake has a salinity about twice that of sea water. Mason noted that due to the salinity, simple extrapolation of what is known of sea and freshwater ecosystems was not possible. This paper represents a comprehensive limnological investigation of a saline ecosystem.

Brissou et al. (1974) studied Lake Assal in French Somaliland, a hypersaline lake with a salinity of 40 percent. This study was mainly descriptive of the bacteria found in the lake with little or no attention given to the ecology of the system. Of the 164 bacterial strains isolated, none were strict anaerobes and most (90 percent) were not extreme halophiles, but were halotolerant forms. Gelatin was hydrolyzed by 54 percent of the strains, 71 percent denitrified, and 25 percent had a urease. The preponderance of halotolerant bacteria was of the genus *Bacillus.*

Copeland and Jones (1965) and Copeland (1967) examined environmental characteristics and community metabolism in hypersaline lagoons. Copeland's findings were previously reviewed under the subject of Hypersaline Water Chemistry in this report. The earlier study by Copeland and Jones found low metabolism, low diversity, and low photosynthesis/respiration ratios to be characteristics of these systems. They noted that diversity and metabolism in a hypersaline habitat resembles that found in a polluted environment. This is a result of the selection that occurs under the strenuous conditions imposed on the biotic components by both types of systems.

Nixon (1969a) studied characteristics of hypersaline ecosystems both *in situ* and in laboratory microcosms. He found natural hypersaline systems to be low in biological diversity, low in nutrients and apparent photosynthesis, and high in dissolved organic matter (dissolved organic carbon often exceeded 75 mg/l). Chlorophyll values were low and animal biomass did not exceed 0.4 gm dry weight/m^2. The biotic component of these systems consisted primarily of algae, brine shrimp, ciliates, and abundant planktonic bacteria. Nixon's laboratory microcosms were seeded with one alga and six bacterial isolates from natural systems. Slow rates of recycling developed in these systems and unusual diurnal patterns of oxygen and carbon dioxide were observed. A light-accelerated respiration just after illumination resulted in an increase in carbon dioxide and a decrease in oxygen when just the opposite was expected. It was postulated that photorespiration or photoheterotrophy accounted for this.

Nixon also tested the effects of the fertilization of hypersaline ecosystems in his microcosms. After treatment, chlorophyll *a* in the benthic algal mats increased over four times and apparent benthic photosynthesis increased over six times. The protein composition of the algae tripled. Chlorophyll *a* in the plankton increased over 500 times after treatment and apparent plankton photosynthesis increased over 48 times.

Nissenbaum (1975) studied the microbiology and biogeochemistry of the Dead Sea. This is a closed, hypersaline ecosystem with a total dissolved solids content of 322,600 mg/l. The dominant cation in the Dead Sea is magnesium and the dominant anion is chloride. Biological diversity was found to be quite low with halophilic bacteria and the green halophilic alga, *Dunaliella viridis,* being the major components. Besides the obligate halophiles (rods and cocci), halotolerant and haloresistant bacteria were also isolated from the lake. Sulfate reducers and a unique obligate magnesiophile bacterium were isolated from the lake's sediments. The halophilic bacteria of the system *(Halobacterium* sp. primarily) and the green alga, *Dunaliella viridis,* both adjust to this harsh environment in different ways. The algae adjust to high salinity by developing a mechanism for the exclusion of salts from the intracellular fluid and using glycerol for osmotic regulation. The bacteria adapted by adjusting their internal inorganic ionic strength, but not composition, to that of the medium.

Studies on the Great Salt Lake

There have also been a few studies conducted on the hypersaline Great Salt Lake in northern Utah. Stephens (1974) studied planktonic and benthic energy flow systems and annual productivity in the southern basin of the Great Salt Lake. Planktonic energy flow was primarily contained in primary production by algae, grazing of algae by brine shrimp, and decomposition of organic matter by bacteria. The benthic energy flow system consisted of primary production by blue-green algae, grazing by brine fly

larvae, grazing of brine fly larvae by birds, and bacterial decomposition of organic matter.

The mean annual phytoplankton productivity of the southern basin was estimated by Stephens to be 85 gm C m^{-2} yr^{-1}. The spring bloom of the principal phytoplankter, *Dunaliella* sp., was found to be limited during April by self-shading and during the remainder of the year by the availability of nitrogen. Grazing by the brine shrimp, *Artemia salina,* also acts to reduce the phytoplankton population during late summer.

Effects of polluted waters from the Salt Lake City sewage canal on the biota of the southern basin of the Great Salt Lake were studied by McDonald and Gaufin (1965). They found the area of the lake where the effluent is dumped to be virtually a biological desert. The polluted water evidently inhibits the indigenous inhabitants of the lake and the hypersaline lake water seems to inhibit the organisms present in the sewage.

The northern basin of the Great Salt Lake (north of the railroad causeway) and the organisms living there have been studied by Crane (1974) in his characterization of bacterial isolates from this portion of the lake and by Post (1975) in his report on the biology of the north arm of the lake. The results of these two investigations have been reviewed in previous sections of this paper.

MATERIALS AND METHODS FOR MICROCOSM STUDIES

Experimental Design

The design of the experimental microcosm for this study of the fate of various nitrogen sources in simulations of the Great Salt Lake is diagrammed in Figure 1. Six experimental microcosms were studied under two conditions of illumination (Figure 2) and four different nitrogen sources. This study simulated mid-summer conditions on the north arm of the Great Salt Lake. These conditions were determined by previous field studies.

The two lighting conditions employed in this study were: illuminated microcosms and dark microcosms. The three dark microcosms were contained in an unlighted box and served as reference points and comparisons to the lighted columns to provide conditions where algal growth did not occur. Since the illuminated microcosms were populated almost exclusively with halophilic algae and bacteria, a condition which closely resembles that found in the study area, the absence of algae in the dark columns made possible an evaluation of the influences that the algal populations and particular sources of nitrogen had on the bacteria. It also permitted an evaluation of the response of the algae to the different nitrogen sources. The three remaining microcosms were illuminated from above by a horizontal bank of four 97 percent spectrum-corrected-to-sunlight Optima 50 fluorescent bulbs (Duro Test Corporation). The average illumination of these columns was 3×10^3 Lux. The horizontal arrangement of bulbs was designed to duplicate the direction of light penetration under natural conditions and also provided a diurnal cycle of illumination for the microcosms. This light-dark cycle was controlled by a timer switch and consisted of 16 hours of light and 8 hours of darkness. These lighting conditions were maintained as constantly as possible for the duration of the study.

The experimental microcosms were constructed of clear lucite columns 75 cm high and 15.5 cm in diameter (Figure 2). These units closely resembled those used by Porcella et al. (1970, 1975). They were completely closed systems with sealed tops and had gas traps for the collection of gases evolved within the microcosms. The gas traps consisted of tygon tubing, 50 ml volumetric burettes fitted with gas sampling septa, and leveling bulbs. This system contained a 2.5 percent H_2SO_4 solution plus methyl red for color (Porcella et al., 1975). Stirring was not employed in the microcosms for this study. Except for a narrow vertical strip which was taped, the bottom 15 cm of the microcosms were painted black inside and out to prevent exposure of the sediments to light. The taped strip acted as a window through which the sediments could be examined periodically for the presence of layering, colors, and gas bubbles. The columns were also provided with inlet and outlet ports for the exchange of media. These ports were fitted with rubber tubing and were closed with pinch clamps.

The microcosms were initially set up on October 7, 1974, after water and sediment samples had

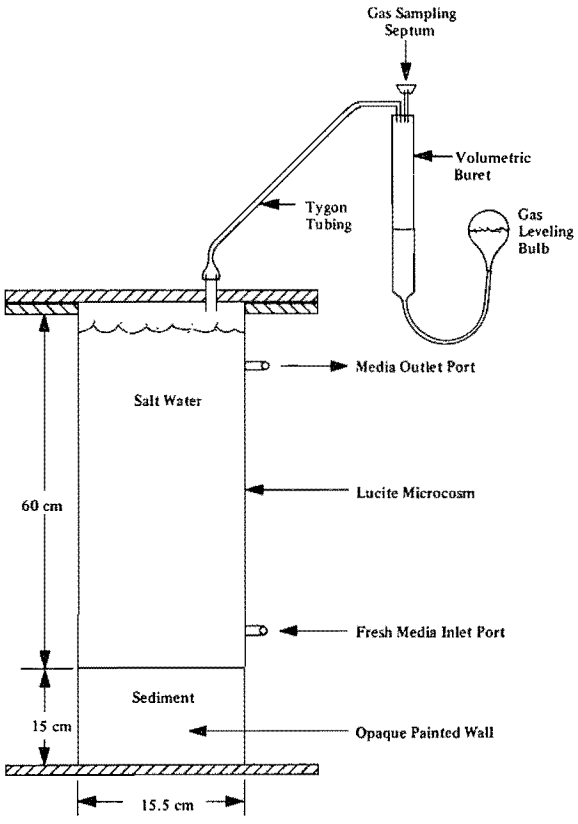


Figure 1. Design of the experimental microcosm (section).

been collected from the northern arm of the Great Salt Lake at Rozel Point (Figure 3). These samples were collected the first week in October and provided the initial seed for the microcosms. The sediment was thoroughly mixed and was then added to each of the six microcosms to a depth of 15 cm (2.25 liters). Approximately 9 liters of mixed lake water were then added to each microcosm to within 3 cm of the top seal. The microcosms were then left to sit for a week so that the sediment and water phases would approach equilibrium before the initial analysis of water samples was undertaken. These procedures resulted in the establishment of six sediment-water microcosm simulations of the northern portion of the Great Salt Lake which were closed to the atmosphere and were capable of undergoing fluid exchange and gas trapping and sampling. The study was terminated with a final chemical and biological analysis on July 2, 1975.

The nitrogen sources utilized in this study were nitrate, ammonia, urea, and L-glutamic acid. They were added to the microcosms to a final concentration of 1 mg/l N. This level of nitrogen was arbitrarily chosen to represent the eutrophic, but unpolluted,

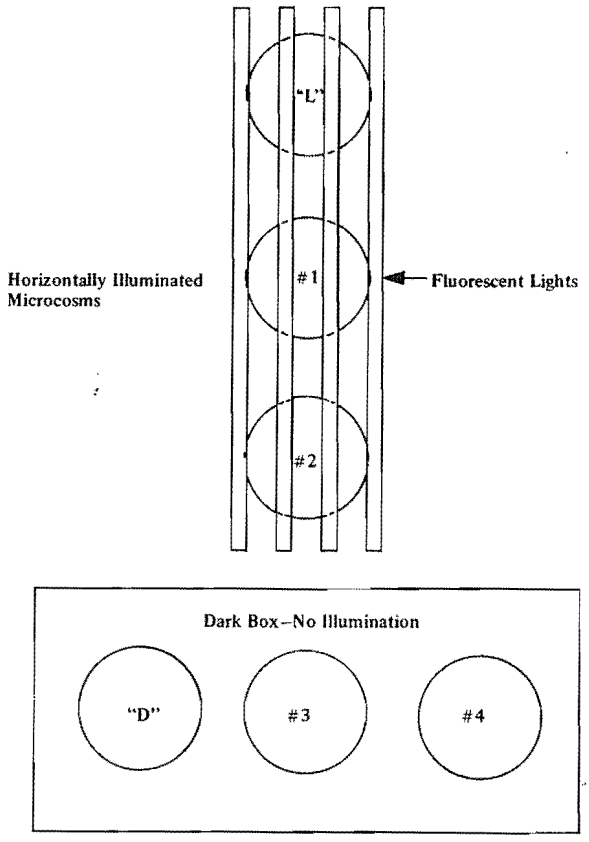


Figure 2. Arrangement of the experimental microcosms (top view).

condition existing in the northern portion of the Great Salt Lake.

The microcosms were run in three light-dark pairs so that the effects of each source of nitrogen could be observed under both lighting conditions. Nitrate was initially fed to all columns. When chemical analysis showed that the microcosms had come to steady state on nitrate, microcosm No. 2 light and microcosm No. 4 dark were switched to ammonia. Microcosms No. 1 light and No. 3 dark remained on nitrate throughout the study to serve as comparisons. This made it possible to determine whether the initial steady state attained in all six columns would remain stable or whether it would undergo further ecological succession when continuously fed a single source of nitrogen.

When the periodic chemical analysis again revealed that a steady state condition had been reached, microcosms No. 2 light and No. 4 dark were switched from ammonia to urea while microcosms "L" light, "D" dark, No. 1 light, and No. 3 dark remained on nitrate. Microcosms "L" light and "D" dark were subsequently changed from nitrate to L-glutamic acid to show the effects of switching from an inorganic to an organic source of nitrogen.

Temperature within the microcosms was closely controlled at 26 C ± 2 C throughout the study. This was accomplished by using heaters during the winter and fans during the summer in an environmental room consisting of two 6 mil plastic walls with a 4 inch dead air space between them. This environmental room was contained within a larger room at the Utah Water Research Laboratory.

Microcosm Media Exchange Procedures

Artificial medium

After the six microcosms had equilibrated for one week, they underwent a schedule of twice weekly medium exchanges (Wednesday-Saturday) and weekly chemical and biological analyses for the remainder of the study. The medium utilized in these exchanges was an artificial salt water solution representing the composition of Great Salt Lake water (Table 3). The composition of this artificial medium was formulated from previous work done on the Great Salt Lake by Handy and Hahl (1966), Porcella and Holman (1972), Stephens (1974), and a chemical analysis by Ford Chemical Laboratory, Inc. (1974).

Each week, 12 liters of a solution of the macro elements were made up from reagent grade chemicals in deionized water. The water displacement by these salts had previously been determined to be 167 ml/l. This allowed the preparation of large volumes of this solution prior to its use. This solution of macro

elements was then placed in a refrigerator and cooled to approximately 10 degrees below room temperature. This cooling prevented the mixing of fresh medium with the medium being removed. Previous studies with dyes (Porcella et al., 1975) have shown that cooling of the fresh medium introduces a temporary thermal gradient near the bottom which is sufficient to allow the removal of displaced effluent from the top port of the microcosm without the inclusion of fresh medium.

Concentrated stock solutions of the micro elements and nitrogen sources were added to the solution of macro elements just prior to the medium exchange.

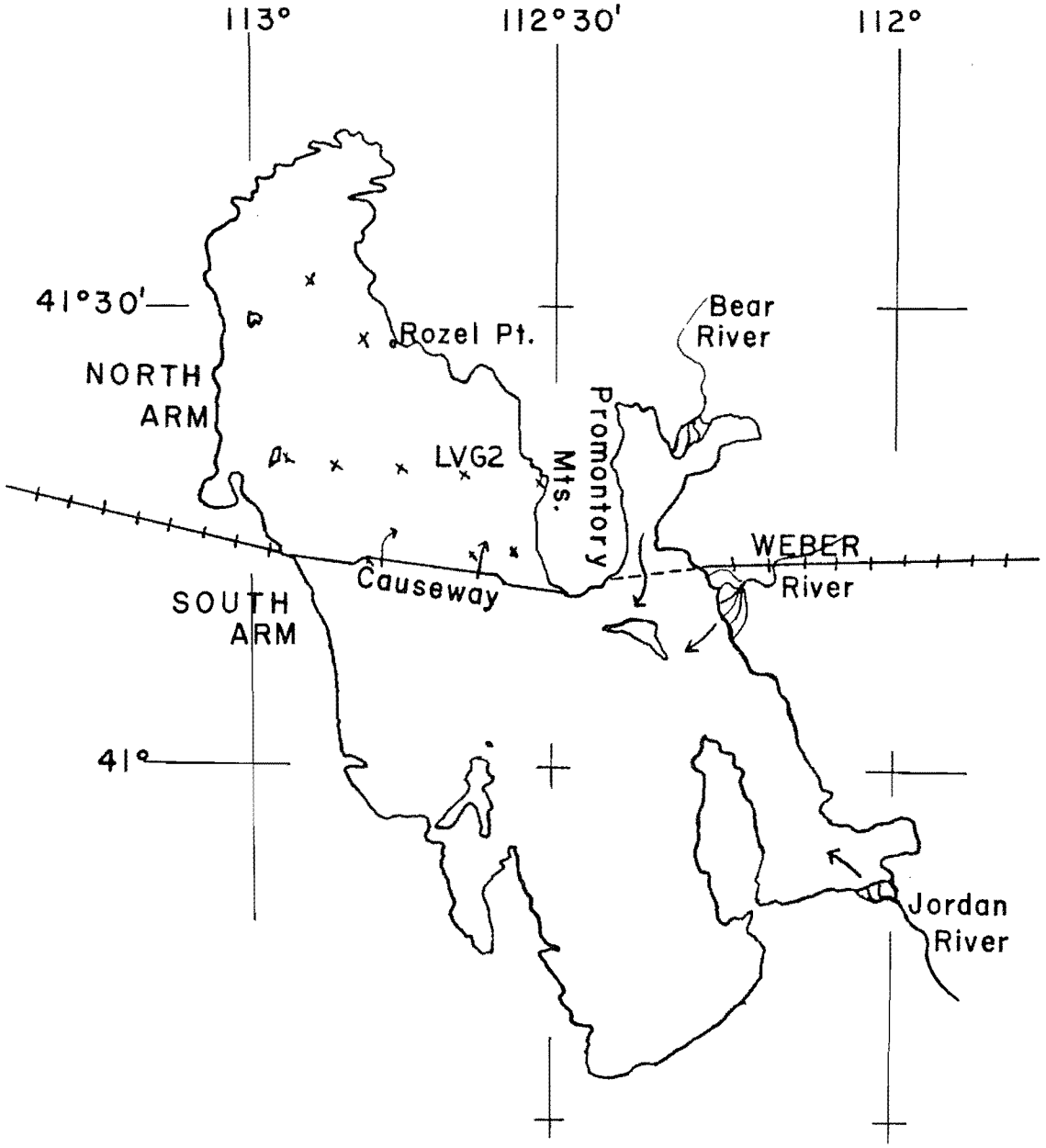


Figure 3. Map of the Great Salt Lake. Scale approximate, 9 km cm^{-1}. Stations marked x sampled vertically four times a year with the cooperation of the Utah Geological and Mineral Survey. Only station LVG2 is identified. LVG1 is east of LVG2 and westward LVG3, LVG4, and LVG5. The north most station is NML, the next south is RD2, in the southeast LVH. Station marked o at Rozel Point is a year around shallow site accessible without a boat and sampled weekly from May to October, then once or twice a month as weather permitted.

Exchange routine

Each time the medium was exchanged, the barometric pressure (mm Hg) and the temperature of the environmental room were recorded. Then the liquid levels in the buret and leveling bulb gas traps were leveled and these levels (read on the burets) were recorded. Additions of helium to the gas phases or the removal of gas produced within the microcosms were sometimes necessary to properly level the gas traps. These removals or additions were also recorded. When the gas traps had been leveled, the tygon tubes leading to the microcosms from the gas traps were clamped shut to prevent the suction of leveling fluid (acid) into the microcosms during the medium exchange.

A pre-determined amount of the cooled macro element solution (1980 ml) was then placed in each of three plastic containers (one for each pair of microcosms). To this volume, 2 ml of each of the micro element stock solutions were added to make up a final volume of 2 liters of artificial medium per pair of microcosms. Each of the three containers then had the appropriate source of nitrogen and concentration of salts for its pair of microcosms.

A syphon was established between the media reservoirs and the microcosms by placing one end of a length of tygon tubing into the medium, filling it with the medium by suction, and then connecting it to the inlet port near the bottom of the microcosm. Another piece of tygon tubing was connected to the outlet port at the top of the microcosm and drained into a 1 liter graduated cylinder. The pinch clamps on the inlet and outlet ports were released simultaneously and replacement of effluent media by fresh media was effected. In this manner approximately 10 percent of the volume of each microcosm (900 ml) was turned over on each exchange day. This corresponded to a mean residence time of 32 days. The simultaneous release of the pinch clamps on the inlet and outlet ports prevented any drastic pressure differences from occurring within the microcosms during the exchange. Any large pressure differences could have resulted in acid being sucked into the microcosms. After pinch clamps had been replaced on the inlet and outlet ports, the clamp on the tube from each microcosm to its gas trap was carefully removed and the acid was adjusted to its previous level. This was accomplished by either opening the upper port to raise the acid level in the buret or by opening the lower port to lower this level.

The volume and temperature of each effluent sample was recorded and on days scheduled for chemical analysis the samples were saved in stoppered glass bottles.

Table 3. Chemical composition of artificial Great Salt Lake medium.

Chemical	Concentration
Macro Elements	
NaCl	255 g/l
KCl	15 g/l
$MgSO_4 \cdot 7H_2O$	71 g/l
$CaCl_2 \cdot 2H_2O$	1.3 g/l
$MgCl_2 \cdot 6H_2O$	36.6 g/l
$NaHCO_3$	1.5 g/l
LiCl	0.4 g/l
H_3BO_3	0.3 g/l
Micro Elements	
$MnCl_2$	264 μg/l
$ZnCl_2$	33 μg/l
$CoCl_2$	0.78 μg/l
$CuCl_2$	0.01 μg/l
$Na_2MoO_4 \cdot 2H_2O$	7.26 μg/l
$FeCl_3 \cdot 6H_2O$	96 μg/l
$Na_2EDTA \cdot 2H_2O$	300 μg/l
H_3PO_4 (85%)	1 mg/l
NaF	13 mg/l

Chemical Analyses

Water

Eight standard chemical analyses were performed once a week on effluent samples from each of the six microcosms. These included determinations of dissolved oxygen, pH, alkalinity, nitrate, nitrite, ammonia, orthophosphate, and total phosphorus. These analyses were also performed on weekly water samples from the sampling site at Rozel Point on the northern arm of the Great Salt Lake. It should be noted that due to the hypersalinity of all these samples, standard methods would not provide acceptable results when performed on unmodified samples. Possible explanations for this salt effect were reviewed in the section on Hypersaline Water Chemistry. When analyses were first run on untreated samples, it was found that results were not reproducible. There were significant and unexplainable differences between split standards where there should have been no differences. Accurate standard curves could not be obtained, and there was even a great deal of week-to-week variation in the differences observed within any one test.

In general, it was found that a 1:10 dilution of the sample, followed by filtering, was necessary before the tests could be made. Also, standards and blanks that were made up in filtered artificial salt

water media that matched the mineral composition and concentration of the samples were run in duplicate with every analysis performed. Thus, a set of standard curves for each analysis was generated each week and in this way the error resulting from week-to-week variations in the saline standards and blanks was minimized. These procedures made it possible to utilize accepted standard methods for all analyses. A flow chart of the chemical analysis procedures is presented in Figure 4.

For each of the tests, acid washed glassware was rinsed prior to use with deionized-distilled water. This water was also used in all dilutions and in making up the saline standards and blanks (except in the ammonia determination). The colorimetric determin-

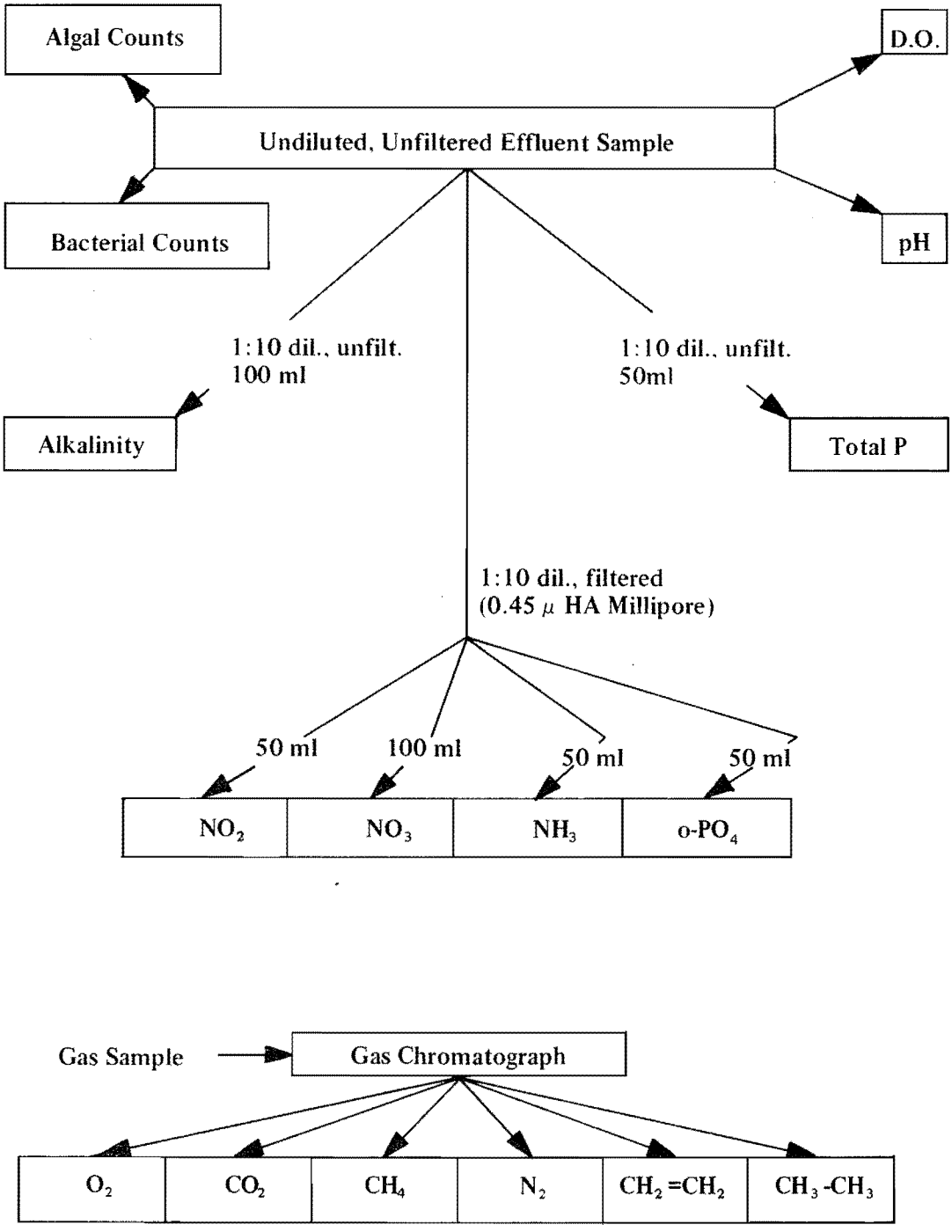


Figure 4. Flow chart of chemical and biological analyses.

ations were all made in a Bausch and Lomb Spectronic 70 using either 1 cm or 5 cm cuvettes.

The pH of undiluted samples was measured on a Beckman Zeromatic II double electrode pH meter. Standard pH solutions were made up with deionized-distilled water. It was found that a salt effect on the pH of hypersaline water causes an increase in pH with increasing dilution. A graph of the relationship between pH and dilution showed that this increase levels off between dilutions of 1:8 and 1:16. A comparison was also made between the pH of a brine sample using a meter standardized with a solution made from brine water and using a meter standardized with a deionized-distilled water solution. The brine water standardized meter gave a pH of 9.50 for a saline sample whereas the same meter, when standardized with a solution made up with deionized-distilled water, gave a pH of 7.33. Due to these salt effects and for the sake of having a reproducible standard method, it was decided that pH's would be measured in undiluted samples on a double electrode pH meter standardized with solutions made up from deionized-distilled water.

The azide modification of the Winkler method was used to determine dissolved oxygen concentrations (Standard Methods, 1971). Undiluted, unfiltered samples were used for this test. A white, flaky precipitate remained after addition of the H_2SO_4 reagent, but went into solution if allowed to sit for a few hours before being titrated.

Alkalinity was determined potentiometrically APHA, 1971) on unfiltered samples that were diluted 1:10. Because of the depression of the pH by the salt effect, the samples were titrated to a pH 4.5 endpoint.

Reactive nitrite was measured in diluted and filtered samples by the diazotization method of Strickland and Parsons (1968). This method consistently produced good standard curves. A standard addition of 7 μg/l of nitrite was added to the microcosm (and lake) samples to bring the reactive amount within the range of the standards. This was then subtracted in the final calculations.

Reactive nitrate was measured on diluted and filtered samples by the cadmium reduction method of Strickland and Parsons (1968). This method proved to be somewhat inconsistent so samples were run through only one cadmium column which was used specifically for salt water samples. This reduced the error so that acceptable standard curves were produced. Previous nitrate tests had been run on several cadmium columns which were also used for other water samples. This introduced an added source of error since there may have been interfering compounds left in the columns from previous samples and each column had to be standardized for the samples that passed through it. However, the use of one specific cadmium column for all salt water samples reduced the error so that acceptable standard curves were produced.

Ammonia concentrations were determined with the indophenol method of Solórzano (1969). Diluted, filtered samples and standards and blanks were all made up with ammonia-free distilled water and, after the addition of reagents, all samples were tightly stoppered during color development. Even with these precautions, however, this method produced the most frustrating results. Split standards which should have been identical often varied greatly. This variation often differed from week-to-week, and reagent blanks were quite high. Thus, the error involved in drawing the line of best fit on the standard curves was somewhat greater for this method than for the others.

Reactive orthophosphate concentrations were measured on diluted and filtered samples with the ascorbic acid technique of Strickland and Parsons (1968). This method gave good results. The microcosm (and lake) samples were spiked with 100 μg/l of orthophosphate to bring the reactive amount within the range of the standards. This standard addition was then subtracted in the final calculations.

Total phosphorus was determined in diluted, unfiltered samples with the persulfate oxidation hydrolysis technique (APHA, 1971). This method, along with the one for reactive nitrite, gave the most consistent results of all the analyses employed. No standard addition was needed in any of the samples from the microcosms or the Great Salt Lake for total phosphorus determination.

Because of machine variability, the 1:10 dilution of samples, the accuracy of drawing the line of best fit on the standard curves each week, and the seemingly inherent error that was involved in the colorimetric determinations carried out on the brine system studied, the lower limit of sensitivity for all the colorimetric analyses was set at 50 μg/l. This limit was set only after experience with the analyses had shown that there were practical limits imposed on these methods by the various sources of error.

The tests most affected by the 50 μg/l limit were the ones for nitrate, nitrite, and ammonia. Quite low concentrations of these species were often encountered during the study and care had to be taken in analyzing the results. Peaks and general trends on the graphs of results often appeared to be significant, but may in fact have been below the sensitivity limit of 50 μg/l. Where this occurred, the results were interpreted as being zero and the peaks were ignored. For the rest of the parameters the 50 μg/l limit determined the significance of peaks on the

graphs and also determined whether or not real differences existed between corresponding curves.

Gas

Gas samples were collected from the microcosms in 2.5 ml disposable syringes. Gas could be removed by these syringes through self-sealing septa located at the tops of the burets in the gas trap systems (Figure 2). The syringes were then inserted into a labeled rubber stopper to prevent gas leakage and were taken to the gas chromatograph to be analyzed.

A Hewlett Packard 5750 research gas chromatograph equipped with a gas sampling valve delivering 0.5 cc of gas to the column was utilized in the gas analyses. Instrument specifications were as follows:

Instrument–Hewlett Packard 5750 Thermal Conductivity and Flame Detector
Columns–6 ft. x 1/8 in. o.d. stainless steel; 60-80 molecular sieve 5A (O_2, N_2, CH_4)
6 ft. x 1/8 in. o.d. stainless steel; 100-120 Porapak S (CO_2, CH_2=CH_2)
Carrier Gas–Helium
Flow Rates–35 ml/min. Helium
55 psi G tank pressure
Temperature–Column 100-110 C
Detector 265 C
Injector Port 110 C

Known standards of oxygen, nitrogen, carbon dioxide, methane, ethane, and ethylene were analyzed along with the gas samples from the microcosms. The mole fraction for each gas was then determined by calculating the area under each peak with the triangulation method.

Biological Analyses

Algal and bacterial counts

On those media exchange days also scheduled for chemical analysis, a representative subsample was taken of each microcosm's effluent for algal and bacterial direct counts. Algae were counted in 132 fields on a Neubauer hemacytometer using a magnification of 200 X. Ten fields of bacteria were counted using a Whipple eyepiece disk and hemacytometer using a magnification of 500 X. All results were reported as organisms/ml.

Final biomass determination

At the close of the study, biomass calculations were made to determine total planktonic algae and bacteria per column, total attached algae and bacteria per column, total biomass per column, and the weights of individual algal and bacterial cells.

Planktonic numbers were determined for effluent samples taken from each of the microcosms by using the procedures outlined in the preceding section. A microcosm was then unsealed and carefully drained so that the algal mat on the sediment surface would not be disturbed. Exactly one liter of a 25.5 percent sodium chloride solution was then carefully added to the drained microcosm and a rubber spatula was used to scrape the attached population from the walls of the microcosm into this solution. The algal mat was disrupted and this turbid solution was drained from the microcosm into a large beaker where it was stirred to break up the clumps of algae and bacteria. Subsamples of this turbid, homogeneous mixture were taken for direct algal and bacterial counts, total nitrogen determinations, and for ash-free dry weight determinations. The microcosms were unsealed and handled one at a time in this manner to prevent the inside walls, with their attached populations of algae and bacteria, from drying.

The weights of individual algal and bacterial cells were calculated from the average volumes of these cells and the densities of their internal environments. For bacteria this internal density was determined from the density of the surrounding environment since extreme halophiles have been shown to adjust their internal ionic strength to that of the medium. Algal density was based on the density of glycerol which these algae produce to prevent osmotic stress in environments of high ionic strength. The weights of individual algal and bacterial cells were determined to be 6.8×10^{-10} g/cell and 2.94×10^{-12} g/cell, respectively. The total biomass per column was calculated by multiplying the total algal and bacterial counts from each column by their respective weights (g/cell). Biomass results were reported as g/column.

The total nitrogen in each sample was measured on a Coleman model 29 nitrogen analyzer. The ash-free dry weight was determined by first placing 20 milliliters of the aufwuchs mixture in a preweighed evaporation dish. This was evaporated at 103 C for 1 hour, cooled and weighed. The result was the weight of salts plus algae and bacteria. This residue was then ashed at 550 C for 1 hour, cooled, and weighed again. This result was subtracted from the evaporation weight and the difference represented the ash-free dry weight of the organisms in each sample.

It is common that during evaporation procedures unstable, volatile salts such as ammonium bicarbonate are completely volatilized. However, these salts are normally in such small amounts in relation to the amount of total solids that their influence is usually ignored. Also, calcium carbonate is decomposed only at temperatures in excess of 825 C so normal ashing procedures would not adversely

affect its stability. These assumptions remain valid for the salt water solution that was evaporated and ashed in the ash-free dry weight determination. The salt water solution was made up from deionized-distilled water and sodium chloride so there was very little ammonium or carbonate present in the samples. Also, the ashing temperature was not sufficient to cause decomposition of calcium carbonate. Thus, the loss in weight was interpreted in terms of organic matter and any volatile loss of salts was considered negligible.

After each microcosm had been unsealed, drained, scraped down and drained again, a sediment core was examined for the presence of layering, colors, and H_2S (by odor).

LAKE STUDIES

Sampling

Samples were collected from Rozel Point weekly during the period from the end of May to the end of October and monthly thereafter as weather and roads permitted. One sample period in August 1975, one in October 1975, and one in February 1976 were devoted to one or more sample stations scattered over the northern arm of the lake at 1.5 meter depth intervals. Samples at Rozel Point were collected at about noon and on the lake trips between 10 a.m. and 2 p.m.

Samples consisted of one sterile bottle for algae and bacterial counts, triplicate dissolved oxygen samples, and one large sample for chemical analysis. Other samples were collected as needed.

Chemical Analyses

All chemical analyses were performed as described earlier.

Biological Analyses

Direct microscopic counts of bacteria and algae were performed as described earlier. Viable bacterial counts were made by plating serial dilutions on the surface of Halorich agar plates, spreading with a sterile bent glass rod and incubating in plastic bags (Falcon, mylar) at 37 C for six weeks. Counts were made at 6 X magnification with stereomicroscope.

Halorich medium was composed as follows (all figures are in grams made up to a final volume of 1 liter): NaCl, 220; KCl, 5; $MgSO_4 \cdot 7H_2O$, 10; $CaCl_2 \cdot 2H_2O$, 0.2; casein hydrolysate (Nutritional Biochemical Co.), 5; yeast extract, 1; KNO_3, 1; Na_2 citrate, 3; agar, 10; pH 7.2 - 7.4.

RESULTS AND DISCUSSION OF MICROCOSM STUDIES

Nitrate Nitrogen

The results obtained from microcosms No. 1 light and No. 3 dark will be used to trace the fate of nitrate in the experimental systems and to determine the effects that nitrate had, as the sole source of nitrogen, on the biological components. These two microcosms remained on nitrate at a concentration of 1 mg N/l throughout the entire experimental period and, in addition to tracing the fate of nitrate, were also used to determine whether the initial steady state attained in the systems would remain stable or whether it would undergo further ecological succession.

At the start of the study, the aqueous phases of both the light and dark microcosms were pink due to the large population of pigmented planktonic bacteria. The sediment surfaces were devoid of any noticeable pigmented growth and had a clean, grey, sandy appearance. The pink color of the water began to fade about two weeks after the initial nitrate feeding began and both the light and dark microcosms were colorless after six weeks. By the seventh week specks of green growth had appeared on the walls of the illuminated microcosm and there was a tinge of green color on the sediment surface. At the end of the eighth week the water phase of the illuminated microcosm was distinctly green and the sediment surface was becoming covered with a mat of green growth. The sediment surface eventually became completely covered with the green growth and the green color of the water became more intense. This condition remained stable for the remainder of the study. Once the dark microcosm had become colorless, it remained colorless for the remainder of the study and the sediment surface remained free of any pigmented growth. This succession and eventual stabilization of color in microcosm No. 1 light and No. 3 dark was observed generally in all four remaining microcosms during the period they were on nitrate. This trend, along with the following discussion, supports the view of Beyers (1963), Abbott (1966), and others that microcosms are replicable and can produce valid results when the amount of variation between replicate systems is minimal.

The color changes in microcosms No. 1 light and No. 3 dark can be understood by examining the algal and bacterial curves for these systems (Figures 5, 6, 11, 12). Initially, the concentration of bacteria in each microcosm was quite high (80 x 10^6/ml), but these numbers began to decrease rapidly as the microcosms continued to be fed nitrate. Eventually, the bacterial concentrations stabilized at levels much lower than the initial ones (approximately 20 x 10^6/ml in the lighted microcosm and 4 x 10^6/ml in the dark microcosm). This decrease in the numbers of pigmented bacteria accounts for the fading of color and subsequent colorless condition of the microcosms after approximately six weeks of nitrate input. The algal curves (Figures 5, 11) indicate the reasons for the green coloration that developed in the illuminated microcosm and the reasons for the continuation of the colorless condition in the dark microcosm. The appearance of algae in the direct counts from the dark columns was due to a carryover that occurred during the sampling procedure. At first, the lighted columns were sampled before the dark columns and one graduated cylinder was used per pair to collect the effluent. Reversing the order of sample collection resulted in no carryover of algae and algal direct counts fell to zero in the dark microcosms. It is felt that all algae observed in counts from the dark columns should be interpreted as carryover and it is for this reason that these algal numbers were not plotted.

The green color in the lighted microcosm was due to the chlorophyll pigmentation of the principal phytoplankter that developed in the system, *Dunaliella viridis*. The phytoplankton concentration continued to increase during the course of the study, reaching its maximum level near the end of the study period. This gradual increase suggests an ability of the algae to utilize nitrate as a source of nitrogen. The development of the algal population can also be seen in the dissolved oxygen curve (Figure 5) whose peaks correspond with the peaks in algal numbers. Also, it can be seen in Figure 6 that the bacterial population in illuminated microcosm No. 1 peaked shortly after the algal population in this microcosm had peaked. The bacteria were probably responding to the increase in dissolved oxygen and organic matter that resulted from the increase in algal numbers. After the bacteria had peaked and were declining, the carbon dioxide concentration in this microcosm was at an elevated level and the algae responded to this favor-

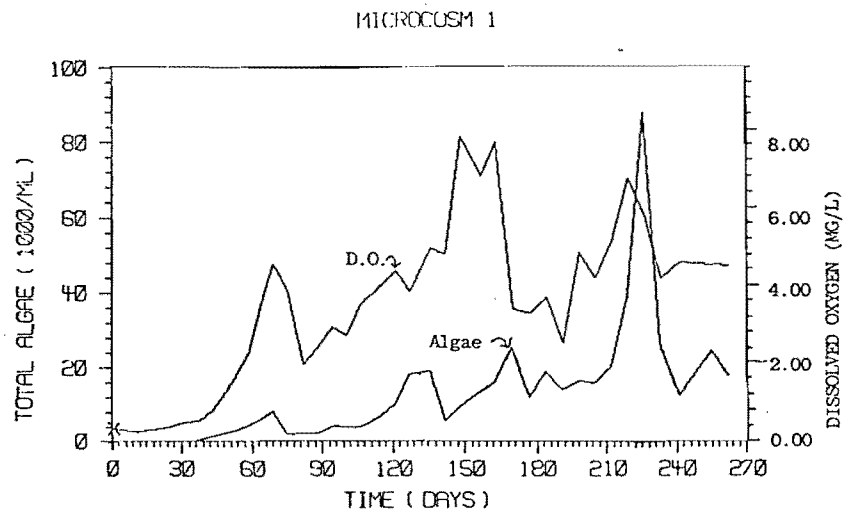


Figure 5. Total algae (+) and dissolved oxygen (x), microcosm #1, light.

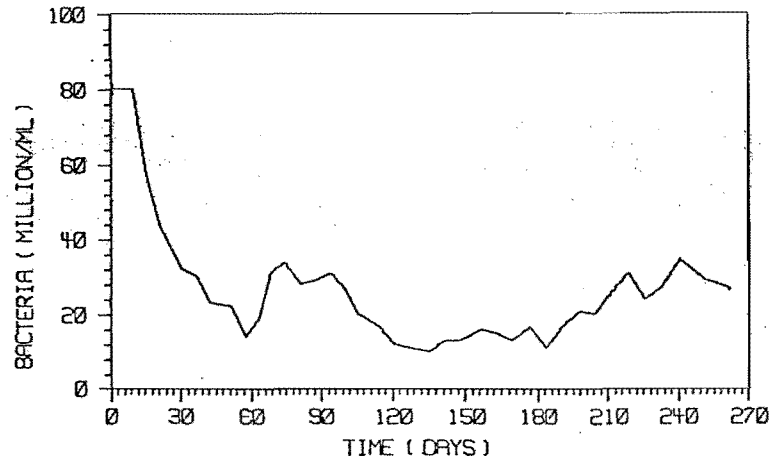


Figure 6. Direct bacterial count, microcosm #1, light.

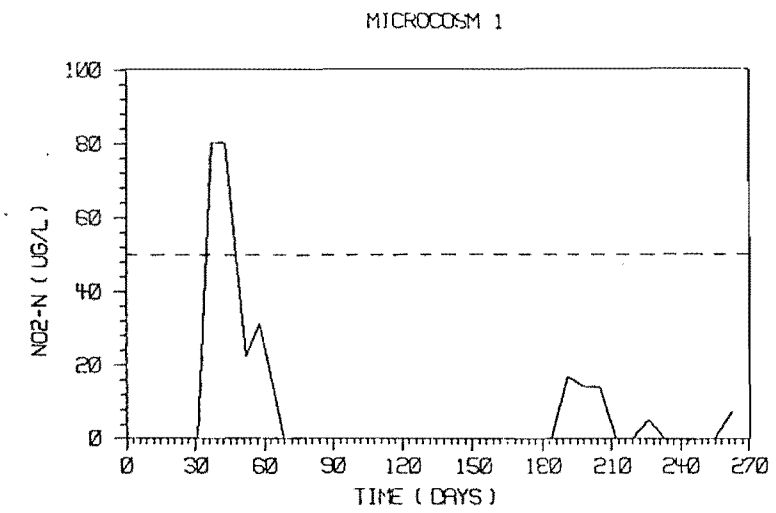


Figure 7. Nitrite nitrogen, microcosm #1, light. Dashed line indicates lower sensitivity limit of test.

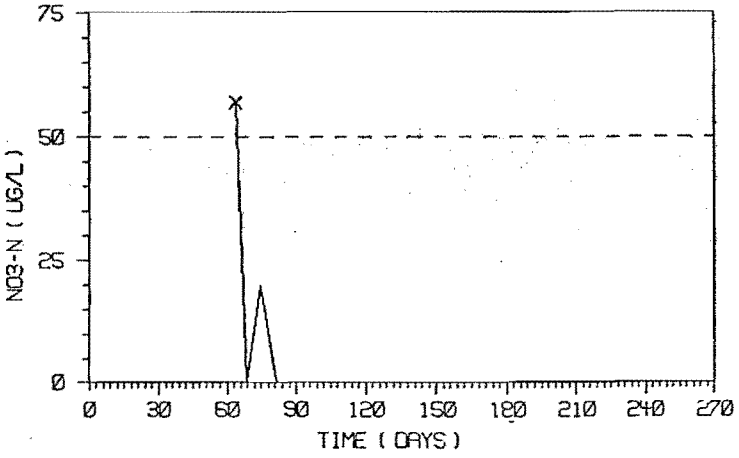


Figure 8. Nitrate nitrogen, microcosm #1, light. Dashed line indicates lower sensitivity limit of test.

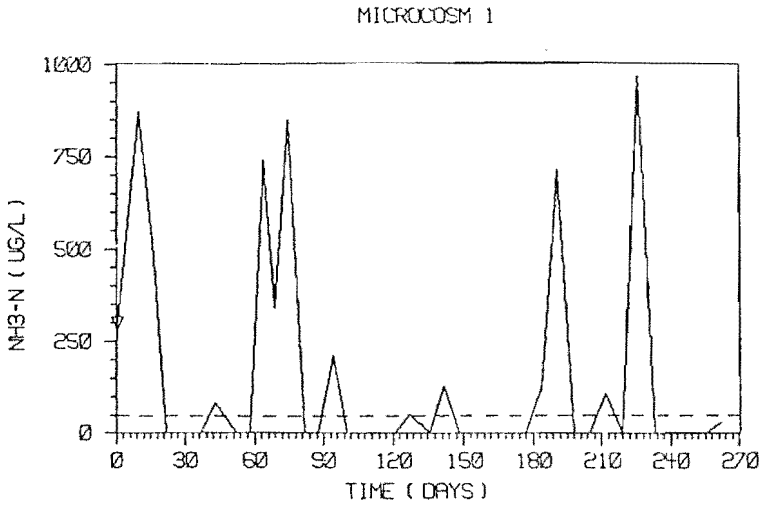


Figure 9. Ammonia nitrogen, microcosm #1, light. Dashed line indicates lower sensitivity limit of test.

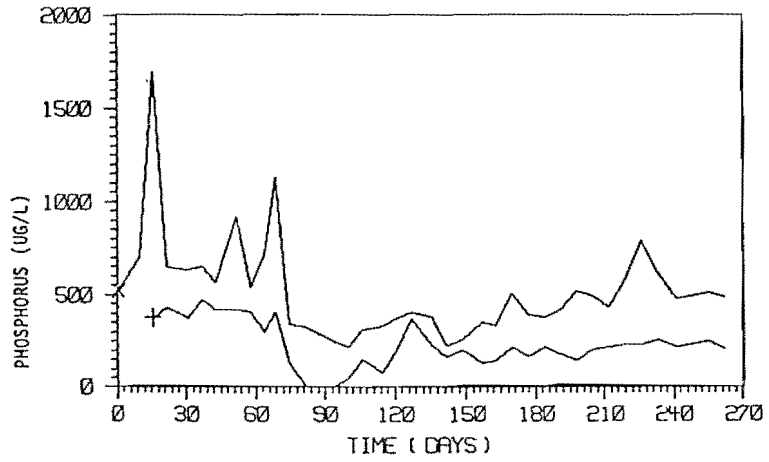


Figure 10. Total phosphorus (x) and orthophosphate (+), microcosm #1, light.

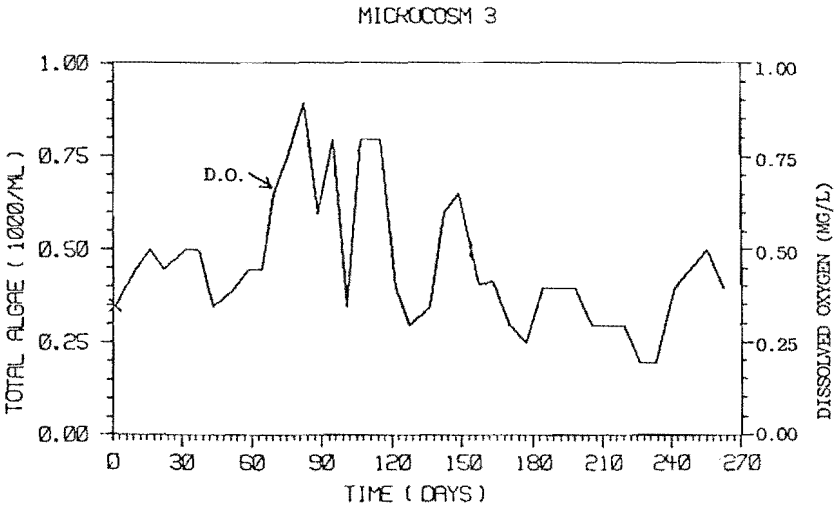


Figure 11. Total algae (+) and dissolved oxygen (x), microcosm #3, dark.

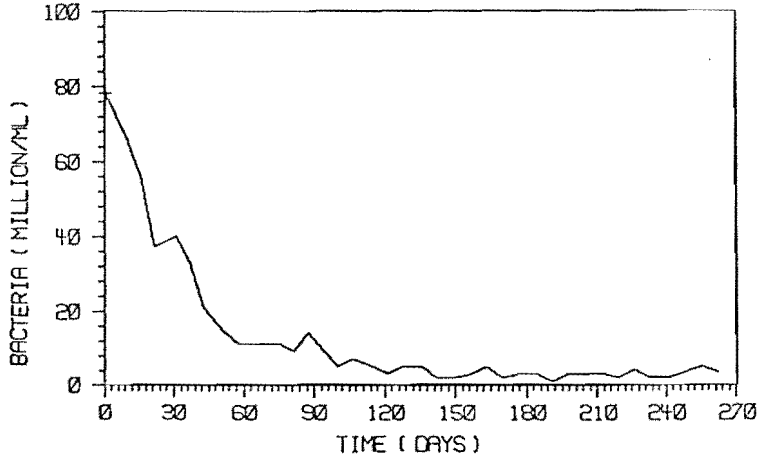


Figure 12. Direct bacterial count, microcosm #3, dark.

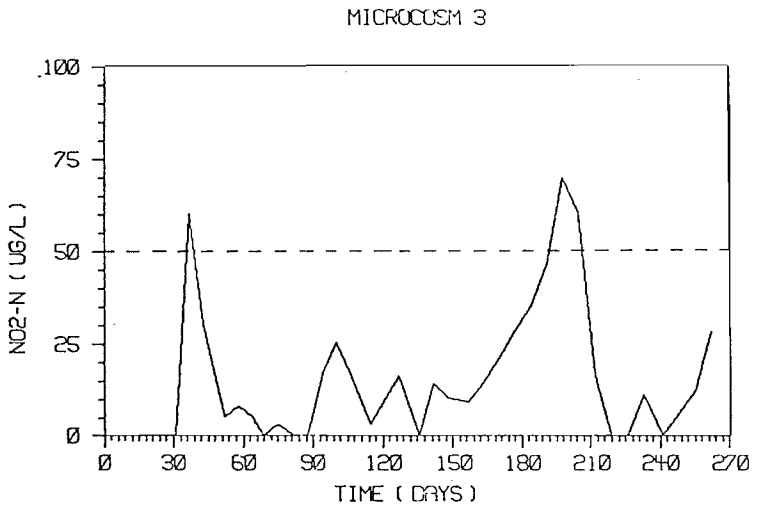


Figure 13. Nitrite nitrogen, microcosm #3, dark. Dashed line indicates lower sensitivity limit of test.

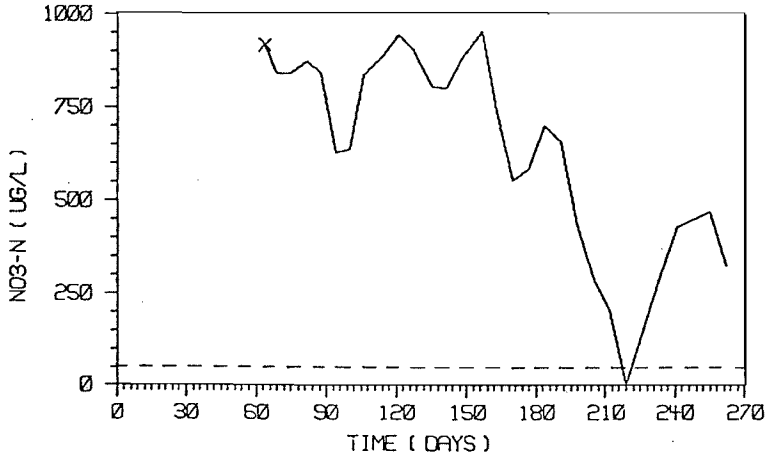


Figure 14. Nitrate nitrogen, microcosm #3, dark. Dashed line indicates lower sensitivity limit of test.

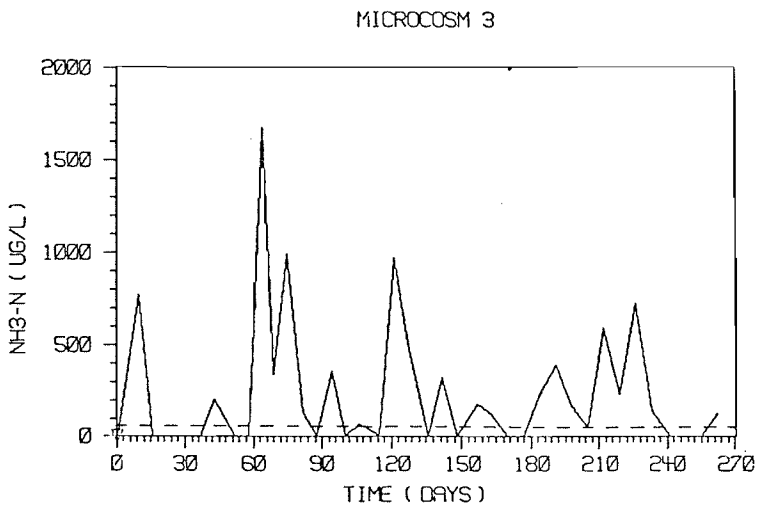


Figure 15. Ammonia nitrogen, microcosm #3, dark. Dashed line indicates lower sensitivity limit of test.

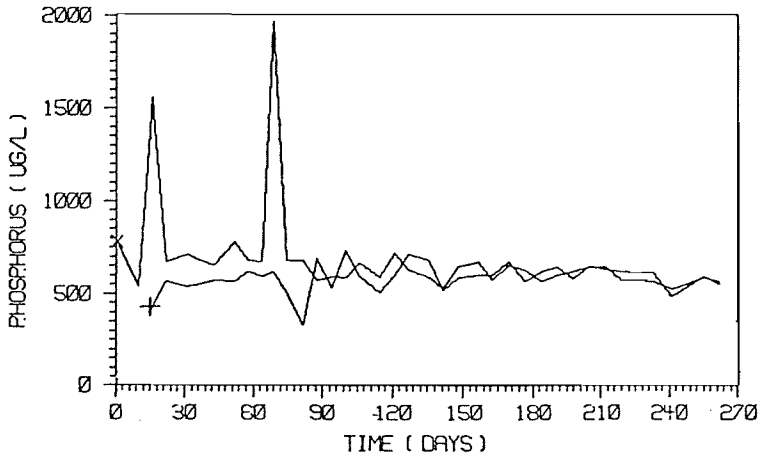


Figure 16. Total phosphorus (x) and orthophosphate (+), microcosm #3, dark.

able growth condition by once again increasing in number. During the time that the illuminated microcosm was undergoing this photosynthetic-heterotrophic successional pattern, the dark column remained colorless since there was no energy source for the photosynthetic algae. The dissolved oxygen curve for the column (Figure 11) remained correspondingly low. While the dissolved oxygen remained at an elevated level in the illuminated microcosm (No. 1) because of photosynthesis (Figure 5), the low level of dissolved oxygen in the dark microcosm (Figure 11) was probably maintained by addition that occurred with the twice weekly medium exchanges. This low level of oxygen allowed for the development of a limited bacterial population which was probably dependent upon the release of organic matter from the sediment for growth nutrients.

An interesting comparison can be made between the bacterial curves for microcosms No. 1 light and No. 3 dark (Figures 6, 12) which shows the effect that an algal population had on the bacteria in this system. Although the bacterial concentrations in both microcosms decreased rather quickly, the bacterial concentration in the illuminated microcosm stabilized at a higher level than the bacterial concentration in the dark microcosm (approximately 20 x 10^6/ml compared to 4 x 10^6/ml). The only significant difference between these two systems was the development of the algal population in the illuminated microcosm. The presence of this algal population sustained a larger number of bacteria than was found in the system without algae. This was most likely due to the higher concentrations of organic matter and dissolved oxygen in the illuminated microcosm which had resulted from the growth of the algae. While nitrate alone was not stimulating to bacterial growth, it did stimulate the development of an algal population in the illuminated microcosm, which in turn had a stimulating effect on the bacteria in the system.

Nitrite production in microcosms No. 1 light and No. 3 dark (Figures 8, 13) was quite sporadic and of limited amount, but it evidently did take place. It occurred most often and in the greatest quantities in the dark microcosm (Figure 13). This was a result of the low oxygen tension existing in this system which created reducing conditions favorable to bacterial nitrite production from the nitrate feed. Crane (1974) reported that several bacterial isolates from the northern arm of the Great Salt Lake were capable of nitrite production or denitrification. Given suitable natural conditions, it was entirely possible that nitrite production from nitrate might occur, as indeed it appeared to do in the dark microcosm (No. 3).

The fate of nitrate in microcosms No. 1 light and No. 3 dark is shown in Figures 8 and 14. The graph starts at day 64 since that was the first analysis for which the nitrate test had been perfected. The dramatic decrease in the concentration of nitrate in illuminated microcosm No. 1 (Figure 8) corresponds closely with the development of the first two peaks of the algal population in that system (Figure 5). The nitrate concentration quickly went to zero and was not detected again even though it was continuously being fed to the system. The algae evidently removed it as fast as it was added. The nitrate concentration in dark microcosm No. 3 (Figure 14) remained at a high level for most of the study indicating that there was very little utilization of it by the bacteria in the system. This agrees with the bacterial curve in Figure 12 which shows no stimulation of the bacteria by nitrate. The sharp decrease in the nitrate concentration in column No. 3 (Figure 14) is probably due to the failure of the cadmium reduction column. It had been used many times and was not reducing all the nitrate in the samples. The subsequent rise in the nitrate concentration in microcosm No. 3 occurred after a new cadmium reduction column had been prepared.

Ammonia production in microcosms No. 1 light and No. 3 dark (Figures 9, 15) was quite sporadic and may have been due in part to analytical difficulties. The greatest ammonia concentrations in illuminated microcosm No. 1 occurred at roughly the same points in time as the increases in the bacterial population and at, or slightly after, algal increases. Mineralization of organic matter by the bacteria in the system would account for this trend. It should be noted that the levels of ammonia in this lighted microcosm could have been determined not only by the mineralization of organic matter by bacteria, but also by the uptake of ammonia by the algae which had developed in this system. Peaks in the ammonia concentration in dark microcosm No. 3 occurred more frequently and at higher levels than those in lighted microcosm No. 1, but this pattern only seemed to be related to general trends in the dissolved oxygen measured in this system. Ammonia concentrations decreased as the dissolved oxygen levels increased and then rose as the dissolved oxygen concentrations decreased. Uptake of ammonia under suitably aerobic conditions and excretion of ammonia under reducing conditions is a well known occurrence in aquatic habitats (Ruttner, 1963). Since bacteria made up the only metabolically active population in microcosm No. 3 dark, the peaks in ammonia concentration were probably due to the metabolism of existing organic matter, which included bacterial cells, and subsequent excretion of ammonia by the bacteria under reducing conditions. The decreases in the ammonia concentration were probably due to uptake of ammonia by these bacteria under the more aerobic situations or during periods of less active metabolism.

Trends in total phosphorus and orthophosphate were also measured with respect to the source of nitrogen in all six microcosms. A comparison of these

two parameters in microcosms No. 1 light and No. 3 dark shows that at equilibrium there was a slightly higher level of total phosphorus in the dark microcosm (Figure 16) than in the illuminated one (Figure 10) and that total phosphorus in the dark column was predominantly in the form of soluble orthophosphate. The difference between the equilibrium levels of total phosphorus in these two systems is probably due to the algal population that developed in the illuminated microcosm, most of which was attached to the walls and sediment surface and unavailable for total phosphorus analysis. There was also a greater bacterial population present in the illuminated microcosm than in the dark one. All this resulted in a much larger metabolically active community in illuminated microcosm No. 1, a large part of which was attached. More phosphate was thus removed from solution in this system than was removed from solution in dark microcosm No. 3 with its rather small concentration of bacteria and little or no attached growth. The equilibrium level of total phosphorus measured in the illuminated microcosm (No. 1) was lower than that in the dark microcosm (No. 3) due to uptake by attached organisms.

The steady increase in total phosphorus in lighted microcosm No. 1 seems to be correlated with a similar increase in planktonic bacteria in this system while the stable bacterial population in microcosm No. 3 dark is reflected in the relatively stable phosphorus curves for that system.

The relationship between ortho and total phosphorus in these two microcosms was also quite different. In the dark microcosm with its stable, low concentration of bacteria, most of the total phosphorus was in the form of orthophosphate while soluble phosphorus represented a smaller part of the total in the illuminated system. After the bacterial population in the dark system had come to steady state and had stabilized, the biological uptake of phosphorus and the exchange of phosphorus with the sediment must have come to equilibrium. Since there was a constant addition of soluble phosphate to the system, the equilibrium established between uptake by bacteria and exchange with the sediments led to a condition in which total phosphorus determined in the water phase was represented by soluble phosphate. The larger number of bacteria and the presence of algae in the illuminated microcosm resulted in a greater uptake of soluble phosphorus in this system and therefore a greater disparity between the levels of ortho and total phosphorus.

Trends in the total phosphorus levels in these systems were more sensitive as indicators of trends in bacterial and algal populations than were trends in orthophosphate. A comparison of algal, bacterial, and total phosphorus curves from illuminated microcosm No. 1 (Figures 5, 6, 10) shows how the response of total phosphorus in this system paralleled trends in algal and bacterial populations while the level of orthophosphate remained at a more or less constant level. This also seems to be true of conditions in the northern arm of the Great Salt Lake. Here orthophosphate remains quite constant while total phosphorus responds to fluctuations in algal and bacterial numbers. This is in agreement with Stumm and Stumm-Zollinger (1972) who point out that the total reserve of phosphorus in a body of water is a pertinent gross parameter because it gives the ultimate capacity for biomass synthesis while the dissolved phosphate gives little indication of phosphate availability and therefore is not a very sensitive parameter.

All six microcosms in this study were initially fed nitrate as the sole source of nitrogen. Parameters measured in nitrate fed microcosms No. 2, No. 4, "L," and "D" all followed the same trends just described for microcosms No. 1 and No. 3.

Ammonia Nitrogen

The fate of ammonia nitrogen and its effects upon the biological components of the microcosms will be elucidated from graphs of parameters measured in microcosms No. 2 light and No. 4 dark. These two systems had previously been on a nitrate nitrogen source, but were switched to ammonia when chemical analysis revealed that a steady state had been reached on nitrate.

Microcosm No. 2 was green both in the water phase and on the sediment surface due to the green algal population that had developed on the previous nitrate nitrogen source. After approximately three weeks on ammonia, this green color seemed to fade slightly, but not enough to make any large difference in the gross appearance of the microcosm. Dark microcosm No. 4 remained as colorless on ammonia as it had been on nitrate.

The persistence of the green algal population in illuminated microcosm No. 2 and the continuation of the colorless condition in dark microcosm No. 4 after they were switched to an ammonia nitrogen source can be understood by studying the algal and bacterial graphs for this pair of microcosms (Figures 17, 18, 23, 24). The arrows on these graphs indicate those points in time when the nitrogen source for a particular microcosm was changed. Thus, the intervals between the arrows on Figures 17 through 28 represent the amount of time spent by each microcosm on a particular source of nitrogen. The nitrogen source used during each interval is indicated at the top of the graph. This labeling system is also used on Figures 29 through 40 for microcosms "L" and "D."

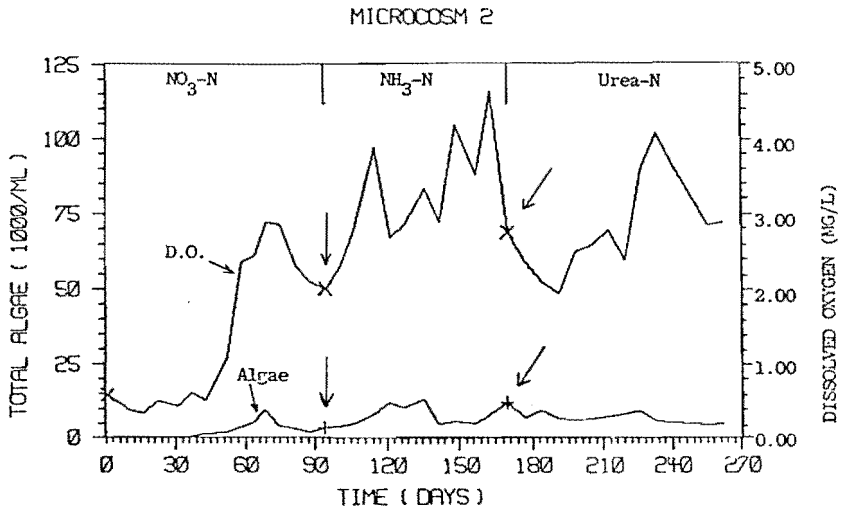


Figure 17. Total algae (+) and dissolved oxygen (x), microcosm #2, light. Arrows indicate input of new nitrogen source.

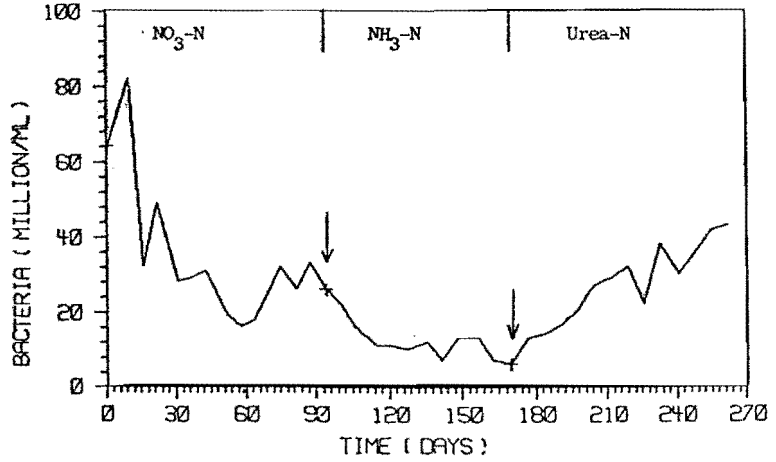


Figure 18. Direct bacterial count, microcosm #2, light. Arrows indicate input of new nitrogen source.

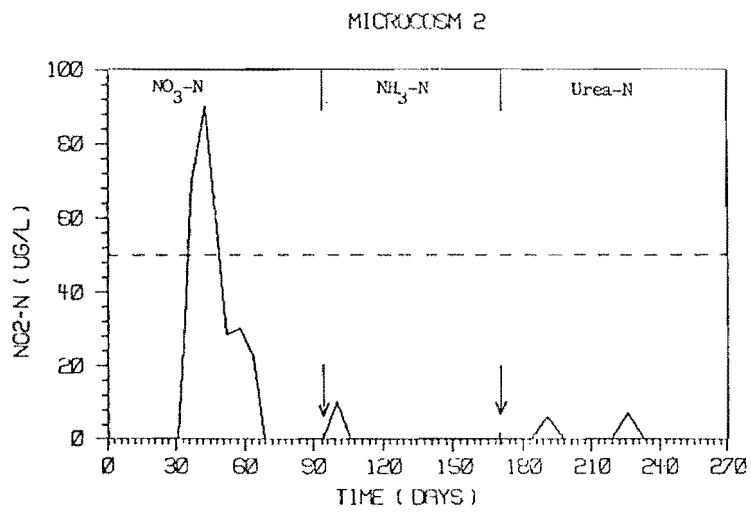


Figure 19. Nitrite nitrogen, microcosm #2, light. Arrows indicate input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

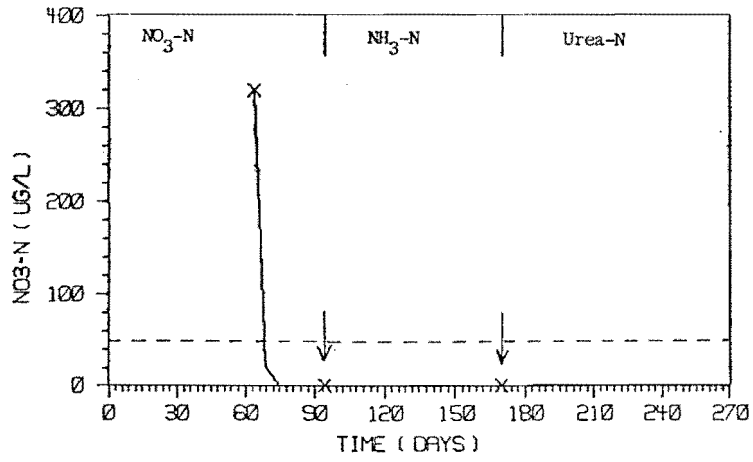


Figure 20. Nitrate nitrogen, microcosm #2, light. Arrows indicate input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

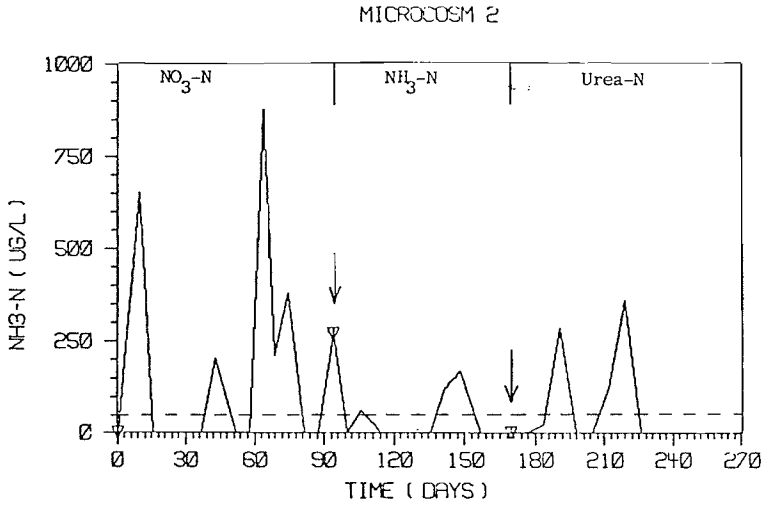


Figure 21. Ammonia nitrogen, microcosm #2, light. Arrows indicate input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

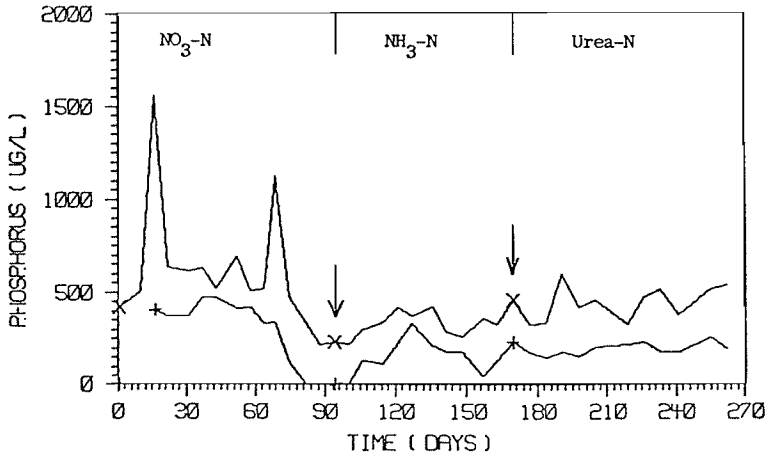


Figure 22. Total phosphorus (x) and orthophosphate (+), microcosm #2, light. Arrows indicate input of new nitrogen source.

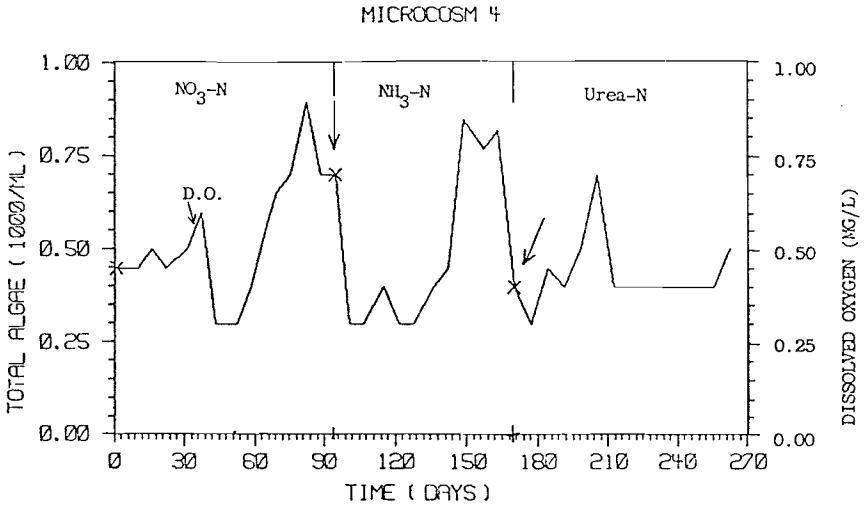


Figure 23. Total algae (+) and dissolved oxygen (x), microcosm #4, dark. Arrows indicate input of new nitrogen source.

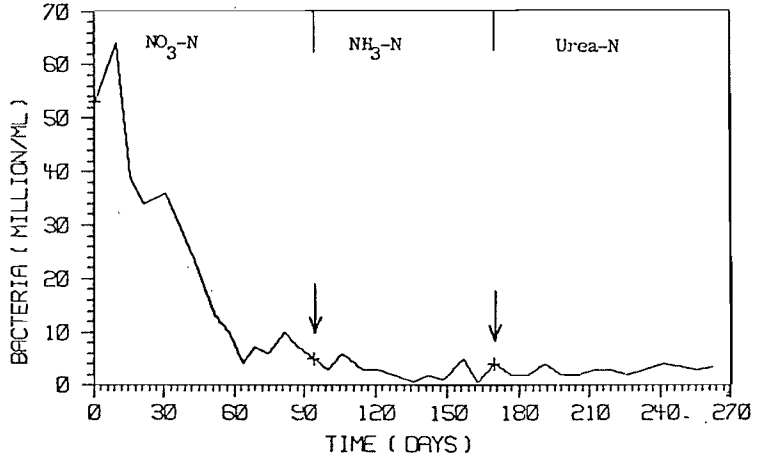


Figure 24. Direct bacterial count, microcosm #4, dark. Arrows indicate input of new nitrogen source.

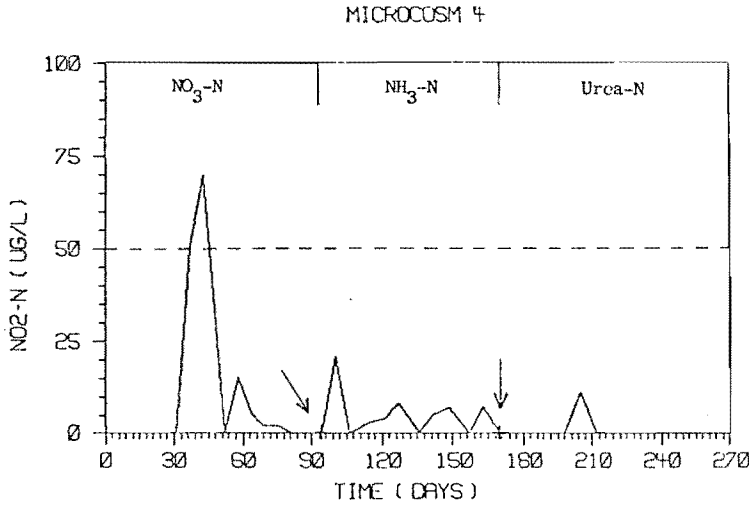


Figure 25. Nitrite nitrogen, microcosm #4, dark. Arrows indicate input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

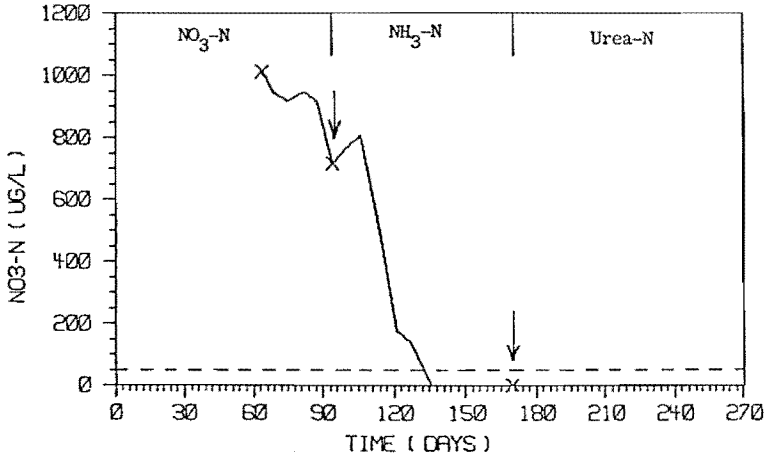


Figure 26. Nitrate nitrogen, microcosm #4, dark. Arrows indicate input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

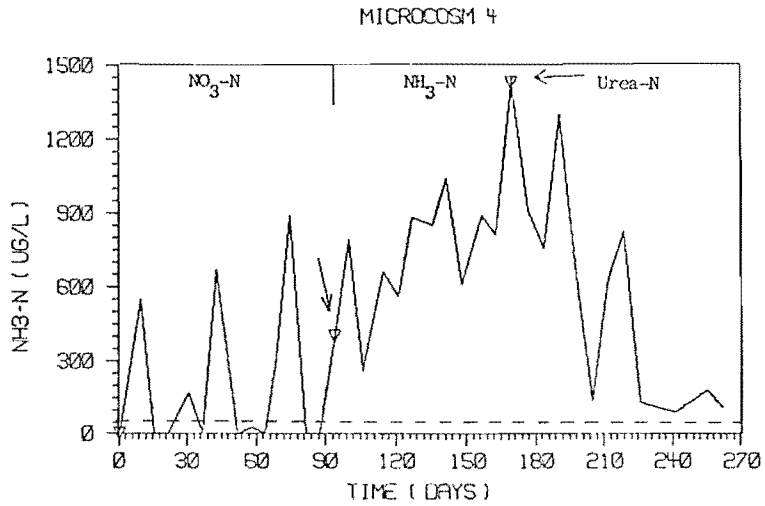


Figure 27. Ammonia nitrogen, microcosm #4, dark. Arrows indicate input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

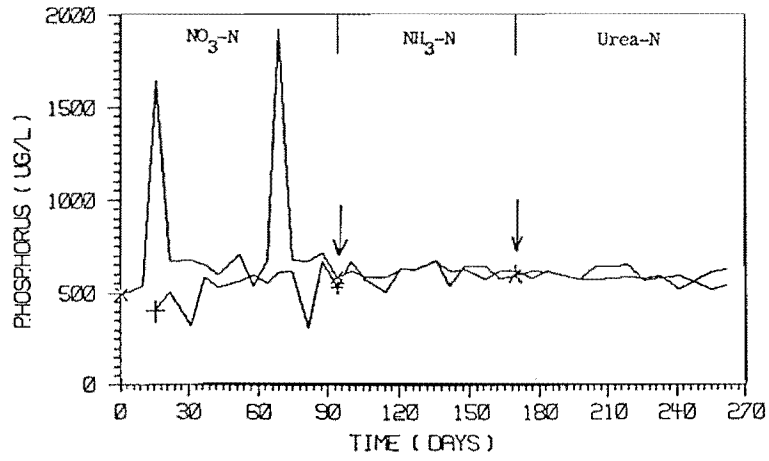


Figure 28. Total phosphorus (x) and orthophosphate (+), microcosm #4, dark. Arrows indicate input of new nitrogen source.

The algal concentration in illuminated microcosm No. 2 (Figure 17) did not vary greatly during the period that ammonia was the sole nitrogen source but judging from the dissolved oxygen curve, the algae remained metabolically active. Comparing this algal curve (Figure 17) to the one in Figure 5, one can see that for the same time interval a greater concentration of algae was produced on the nitrate nitrogen source than was produced on the ammonia nitrogen source. This seems to indicate that the more oxidized form of nitrogen is more stimulating to the growth of the major phytoplankter of the microcosm system, the green alga, than is the reduced form. This would seem to be energetically unfeasible, but may explain why a red alga is more numerous than the green alga during algal blooms in the northern arm of the Great Salt Lake. It can be seen in Appendices B and C that the green alga is stimulated by nitrate, but show little response to ammonia, while the red alga is stimulated by ammonia and not by nitrate. The number of red alga in lighted microcosm No. 2 increased over 14 times while on ammonia and green algal numbers under the same conditions increased only a little over three times. Since chemical analyses of water from the northern arm of the Great Salt Lake have shown ammonia to be the only significant form of inorganic nitrogen present in that system and since other work indicates the presence of nitrate and nitrite in the south arm water (Porcella and Holman, 1972), it is quite possible that the ammonia selects for the red alga whereas nitrate selects for the green form. This might account for the observation that the green form predominates as the phytoplankter in the southern arm of the lake and the red form in the northern arm.

Dissolved oxygen concentrations in lighted microcosm No. 2 (Figure 17) show the expected responses to fluctuations in the algal population. The slight increases in dissolved oxygen during the period on ammonia can be correlated to the slight increases in algal numbers during this time. This contrasts with the low level of dissolved oxygen present in dark microcosm No. 4 (Figure 23). As in microcosm No. 3 dark (Figure 11) the presence of even a low level of oxygen in this system is probably due to an addition which occurred with the twice weekly medium exchanges.

Bacterial numbers in illuminated microcosm No. 2 (Figure 18) decreased continuously while on ammonia. The only increase in the bacterial population was a minor one which occurred shortly after a small peak in the algal population in this microcosm. The increased level of organic matter and dissolved oxygen due to the activity of the algal population probably created conditions favorable to the bacteria. This slight increase in the bacterial population was subsequently followed by a drop in the dissolved oxygen concentration as seen in Figure 17. The bacterial population in dark microcosm No. 4 (Figure 24) followed the same general trend while on ammonia that the bacterial population in dark microcosm No. 3 had followed while on nitrate. A constant decrease in bacterial numbers was followed by the subsequent stabilization of the population at a low concentration. A comparison of the bacterial curves in Figures 18 and 24 indicates that the algal population which persisted in illuminated microcosm No. 2 was responsible for the stabilization of its associated bacterial population at a higher concentration than that found in the dark microcosm (No. 4). Increased levels of organic nutrients and dissolved oxygen in the lighted microcosm must have accounted for this. Apparently, ammonia itself is not stimulating to bacterial growth which must be associated with a more favorable nitrogen source if bacterial growth is to be enhanced. Onishi and Gibbons (1965) found that ammonium chloride had a stimulatory effect on the growth of *Halobacterium cutirubrum,* but also noted that the ammonia itself was not specifically required. Its stimulatory effect was related to the utilization of amino acids. This could also be occurring in lighted microcosm No. 2 where a high concentration of bacteria was associated with an algal population using the ammonia source of nitrogen. Bacteria without the added organic nitrogen source supplied by algae (dark microcosm No. 4) were not stimulated by ammonia alone.

Nitrite production in microcosms No. 2 light and No. 4 dark (Figures 19, 25) was absent. The peaks that do show up are well below the lower sensitivity limit of 50 μg/l that was chosen for this study. They could also represent some conversion of previously added nitrate to nitrite by bacteria (Figures 25, 26).

One would also expect nitrate, if not being produced in the system, to steadily decrease in concentration as the twice weekly medium exchanges continued without added nitrate. With an average effluent volume of 950 ml being removed on each exchange day it would then theoretically take 9.4 exchanges (32 days) to completely replace the nitrate in the system (assuming that the input of fresh medium and displacement of microcosm fluid followed a plug-flow removal). Figures 20 and 26 show that while this is essentially what happened during the time that ammonia was being added as the sole nitrogen source, it did take a little longer to completely replace the nitrate than was calculated indicating that the displacement of medium was not strictly by plug-flow movement. It is quite apparent, though, that nitrate was not being produced in these systems. In fact, as in lighted microcosm No. 1 (Figure 8), the nitrate concentration in lighted microcosm No. 2 was at zero by day 75 in the study and was not detected again. The algae were evidently using it up faster than it was being added. It is apparent from the nitrite and

nitrate results obtained from Microcosms No. 2 light and No. 4 dark that, even while on ammonia, no nitrification took place in these systems.

Figure 27 shows an increasingly high level of ammonia in dark microcosm No. 4. Since the bacteria in this system were at quite low concentrations while on ammonia and since there was no algal population present to provide favorable growth nutrients for the bacteria, the high level of ammonia must have been due more to the twice weekly input of ammonia than to its production by bacteria. There was very little use of ammonia by the organisms present so it simply accumulated in the microcosm.

The highest ammonia levels that occurred in lighted microcosm No. 2 (Figure 21) corresponded with the slight increases in bacterial numbers that occurred in this system while on ammonia. A comparison of the ammonia curves in Figures 21 and 27 shows that even with a continuous input of ammonia to these systems there was no steady accumulation of it in lighted microcosm No. 2 as there was in dark microcosm No. 4. Since the results from dark microcosm No. 4 show that the bacteria were not utilizing the added ammonia to any large extent (Figures 24, 27), the major phytoplankter in the illuminated microcosm (No. 2), *Dunaliella viridis*, must have subsisted on ammonia nitrogen well enough to maintain a fairly stable population which in turn provided favorable conditions for the bacteria. The bacteria probably mineralized organic nitrogen produced by the algae to form ammonia and it was this ammonia that is seen to correspond with bacterial population trends.

The previous observations concerning orthophosphate and total phosphorus in microcosms No. 1 light and No. 3 dark are essentially identical to those for microcosms No. 2 light and No. 4 dark. In fact, superimposing the phosphorus curves for the illuminated or the dark microcosms of these two sets shows that not only the week-to-week trends were the same, but also that the actual concentrations of ortho and total phosphorus were identical in the separate microcosms. Although trends in total phosphorus were closely related to trends in algal, bacterial, and ammonia data, it appears that ortho and total phosphorus concentrations did not differ whether the source of nitrogen was ammonia or nitrate. This probably results from both inorganic nitrogen sources having the potential for maintaining essentially the same gross amounts of biomass in these hypersaline systems.

Urea Nitrogen

Urea was the last nitrogen source employed in microcosms No. 2 light and No. 4 dark. Application of urea began when chemical analysis had shown that the microcosms had come to equilibrium on ammonia. The green color of the illuminated microcosm (No. 2) that was characteristic of this system while on ammonia persisted for approximately three weeks after the initial application of urea. Microcosm No. 2 then began to clear up a little and a pink tinge developed in the light-green plankton. A pink coloration also became associated with the previously all-green algal mat on the sediment surface. This condition continued for the duration of the study while the system was on urea. During this time, the plankton and sediment surface of dark microcosm No. 4 remained as free from any visible growth as it had while on the nitrate and ammonia nitrogen sources. Figures 17, 18, 23, and 24 suggest the reasons for these trends in the appearance of the microcosms.

The planktonic algal population in lighted microcosm No. 2 (Figure 17) declined for about three weeks after urea was substituted for ammonia, then decreased slightly again and remained stable for five weeks, and subsequently decreased slightly once more and remained stable till the end of the study. These slight decreases in algal numbers may account for the slight clearing that occurred during the period of urea input. An apparent discrepancy exists for this microcosm between the dissolved oxygen curve, which increases during this period, and the algal concentration which remains low and stable (Figure 17). However, increased algal photosynthesis without a concomitant increase in biomass could be responsible for the apparent discrepancy. The bacterial curves (Figures 18, 24) are closely related to this subject.

The concentration of bacteria in lighted microcosm No. 2 increased over five times while on urea and was still increasing at the end of the study (Figure 18). This would account for the pink color that developed in this urea-fed system. In addition, an examination of the algal and bacterial curves in Figures 17 and 18 might lead one to think that while urea was not stimulatory to the algae, it was to the bacteria. This, however, was not the case. Figure 24 shows that bacteria not associated with an algal population (dark microcosm No. 4) were not stimulated by urea, but stabilized at a much lower concentration than in the illuminated microcosm. This, along with the algal and dissolved oxygen curves, suggests that it was the algal population, rather than a direct effect by the urea, that was responsible for the increase in bacterial numbers.

The increase in dissolved oxygen without an increase in algal numbers indicates that the metabolism of the existing algal population increased when the algae were fed urea. This increased algal metabolism, with its resultant increase in production of organic matter, accounts for the increase in

bacteria. The stimulation of algal metabolism without an increase in algal numbers also suggests that some element, such as carbon or a trace element, may have been limiting to the algae in this system. In the presence of algae, bacteria were stimulated to a higher degree by a urea source of nitrogen than they were by either nitrate or ammonia nitrogen, again suggesting that much of the urea was transformed to a form more readily used by the bacteria than either nitrate or ammonia.

Nitrite production did not occur in microcosms No. 2 light or No. 4 dark during the time they were fed urea. Nitrate was also absent from these systems (Figures 19, 20, 25, 26). This, along with the conclusion drawn from previous results (while these two microcosms were on ammonia), indicates that no nitrification took place in these systems during the study.

Ammonia production in illuminated microcosm No. 2 seemed to be quite irregular (Figure 21). The ammonia concentration in this microcosm was quite low immediately before and after the initial urea input, so algal metabolism of urea with the excretion of excess ammonia, as well as the mineralization of organic matter by bacteria, might have accounted for the peaks that occurred in the ammonia concentration.

The ammonia concentration in dark microcosm No. 4 (Figure 27) was quite high prior to the initial input of urea. This level, due mainly to the previous input and small bacterial utilization of ammonia, steadily decreased to a lower concentration. The peaks are probably due to mineralization of urea by bacteria and the decrease in ammonia is probably a result of dilution. Since no analyses for urea were performed, it is difficult to determine whether or not it accumulated in the system. But since the level of ammonia and the concentration of bacteria in microcosm No. 4 did not increase and in fact decreased, it is apparent that urea was not utilized to any significant extent and probably did accumulate.

The trends and concentrations in ortho and total phosphorus while microcosms No. 2 light and No. 4 dark were on urea (Figures 22, 28) are virtually identical to those that occurred in microcosms No. 1 light and No. 3 dark during the same time period while the latter were on nitrate. Total phosphorus in lighted microcosm No. 2 increased in response to the increasing bacterial population, but orthophosphate remained stable. The total phosphorus in the dark column was in the orthophosphate form and remained at the same concentration that the total phosphorus in dark microcosm No. 3 was at for this period of the study. Thus, the concentration of ortho and total phosphorus remained unchanged regardless of whether the nitrogen source was nitrate or urea. As before, only total phosphorus responded as a sensitive indicator of biological activity in the microcosms.

Glutamic Acid Nitrogen

Results from microcosms "L" light and "D" dark were used to determine the fate of glutamic acid nitrogen in the experimental systems and the effects that it had on the biological components. Prior to the initial input of glutamic acid, these two microcosms had been receiving nitrate as the sole nitrogen source. The parameter responses to nitrate nitrogen that were measured in these systems were identical to those discussed earlier for microcosms No. 1 light and No. 3 dark.

Before the initial addition of glutamic acid, microcosm "L" had come to equilibrium on nitrate and had developed a green growth in the plankton, on the microcosm walls, and on the sediment surface. Within two weeks after being on glutamic acid, lighted microcosm "L" began to clear up and by the third week the green color was much lighter. By the end of the fourth week the green color was all but gone and a pink color dominated the microcosm. The pink tinge became darker and stabilized by the sixth week. The sediment surface took on a dark and then a black appearance. Like the other dark microcosms, microcosm "D" had cleared up by the sixth week of the study and remained colorless for the duration of the experiment. No color changes were observed in this microcosm while it was on glutamic acid. The reasons for this will be discussed in a following section.

The succession of color changes that did occur can be understood after examining Figures 29, 30, 35, and 36. The algal population in illuminated microcosm "L" began to decrease soon after the initial input of glutamic acid and continued this rather steady decrease through the remainder of the study. The algal population exhibited a minor peak once toward the end of the study which is reflected in the concomitant slight increase in dissolved oxygen which also occurred at this point. This dissolved oxygen curve closely followed the trends in the algal population. The disappearance of the green color that was observed in illuminated microcosm "L" is accounted for by the quite dramatic decrease in algal numbers in this system. The algal population dropped from approximately 10×10^3/ml to about 3×10^3/ml. No algal population developed in dark microcosm "D" and the dissolved oxygen concentration was correspondingly low.

As bacterial numbers increased in response to the glutamic acid nitrogen source, algal numbers began to decrease greatly. The immediate response by the bacteria to glutamic acid was seen in the pink color that quickly developed in the microcosm.

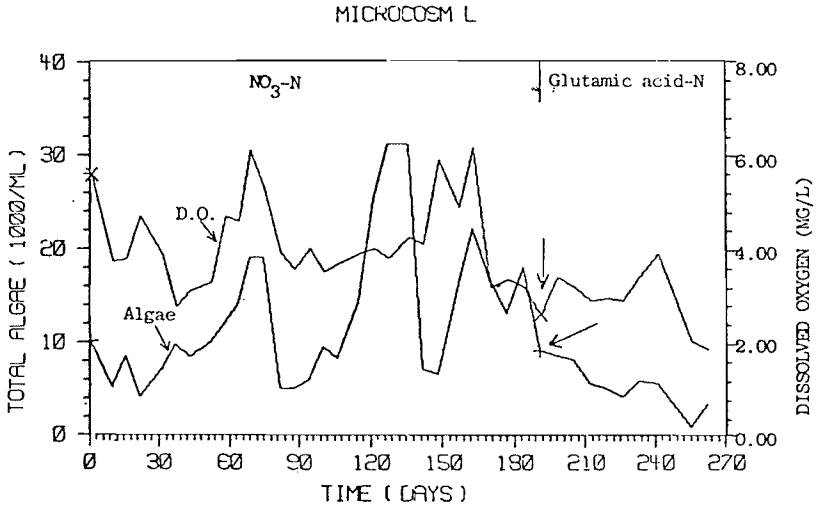


Figure 29. Total algae (+) and dissolved oxygen (x), microcosm L, light. Arrow indicates input of new nitrogen source.

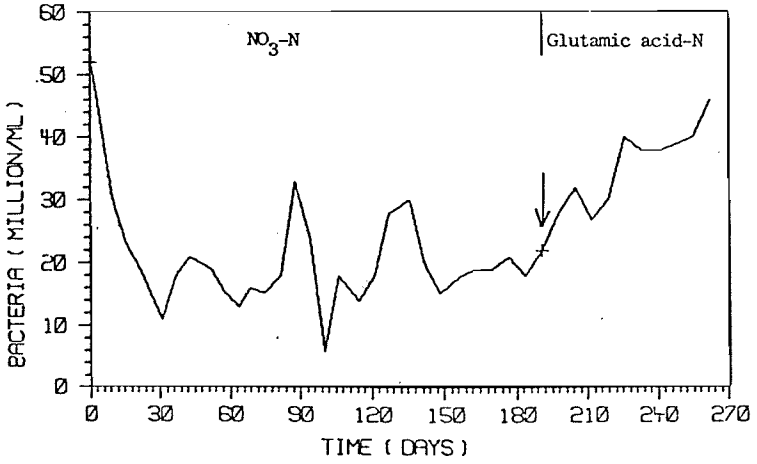


Figure 30. Direct bacterial count, microcosm L, light. Arrow indicates input of new nitrogen source.

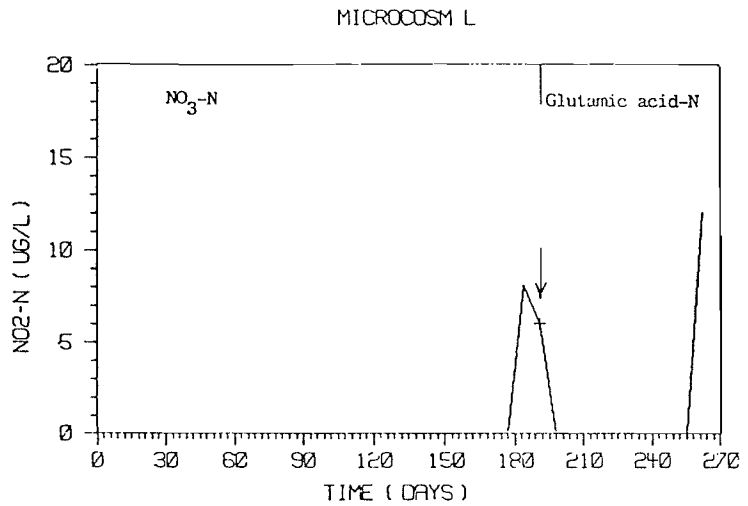


Figure 31. Nitrite nitrogen, microcosm L, light. Arrow indicates input of new nitrogen source.

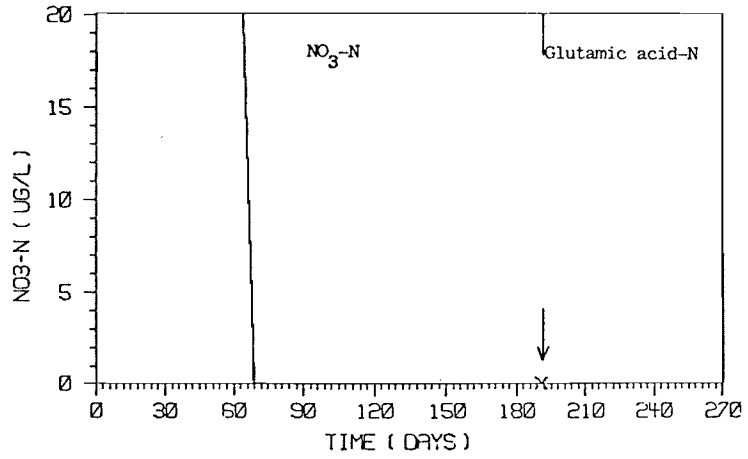


Figure 32. Nitrate nitrogen, microcosm L, light. Arrow indicates input of new nitrogen source.

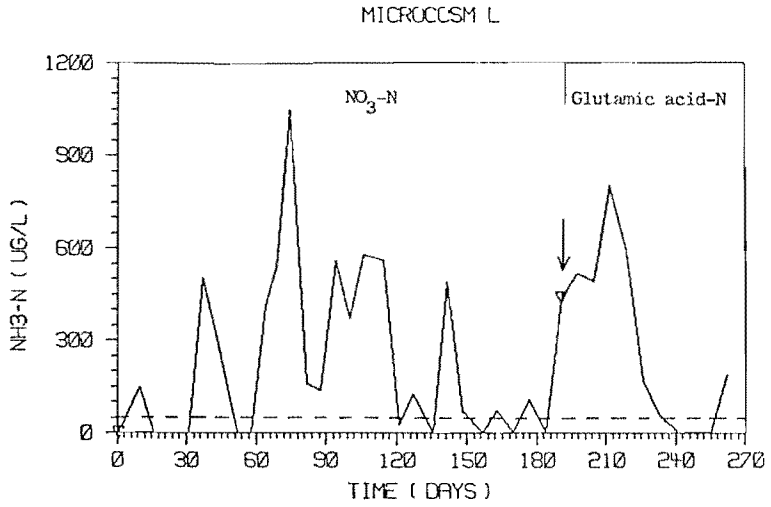


Figure 33. Ammonia nitrogen, microcosm L, light. Arrow indicates input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

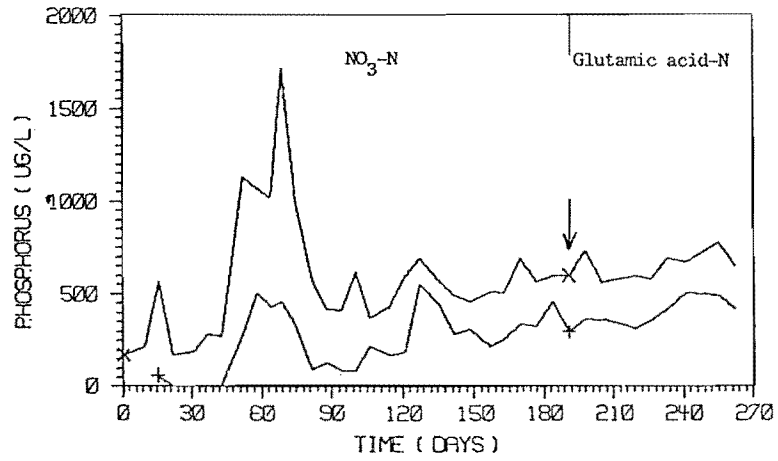


Figure 34. Total phosphorus (x) and orthophosphate (+), microcosm L, light. Arrow indicates input of new nitrogen source.

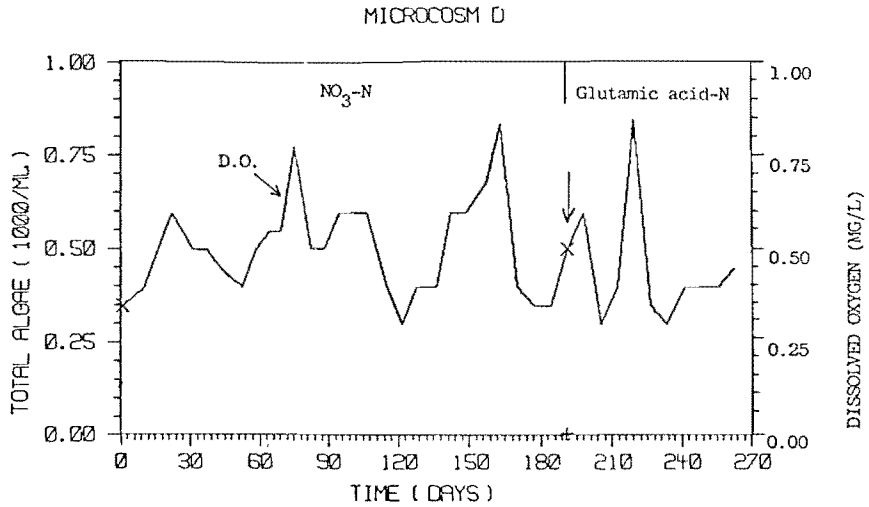


Figure 35. Total algae (+) and dissolved oxygen (x), microcosm D, dark. Arrows indicate input of new nitrogen source.

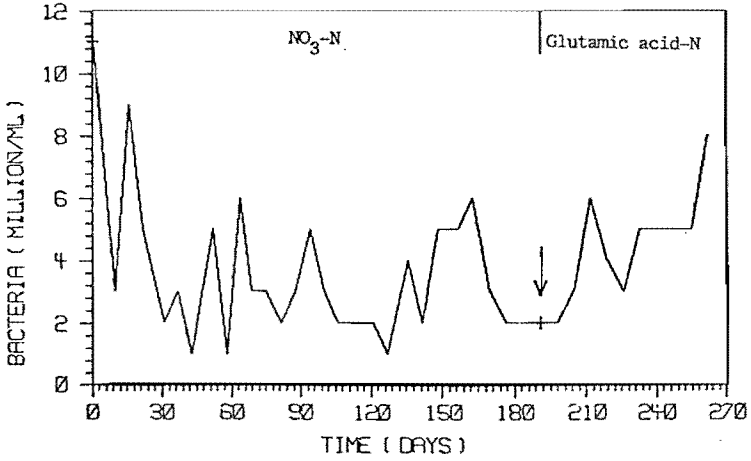


Figure 36. Direct bacterial count, microcosm D, dark. Arrow indicates input of new nitrogen source.

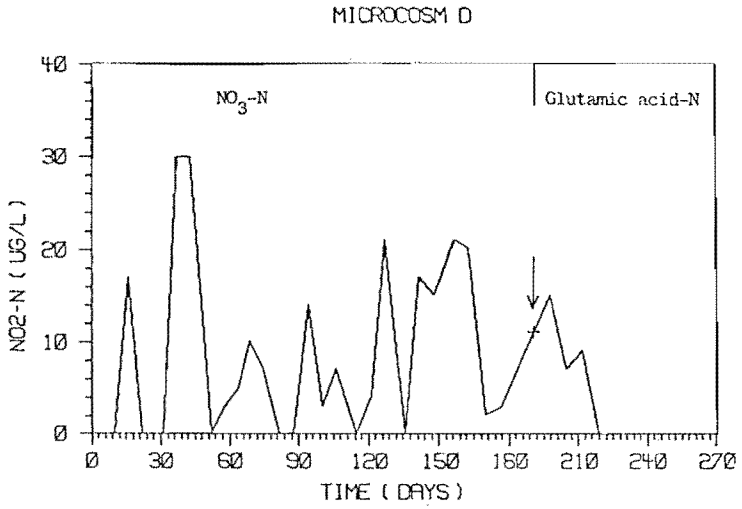


Figure 37. Nitrite nitrogen, microcosm D, dark. Arrow indicates input of new nitrogen source.

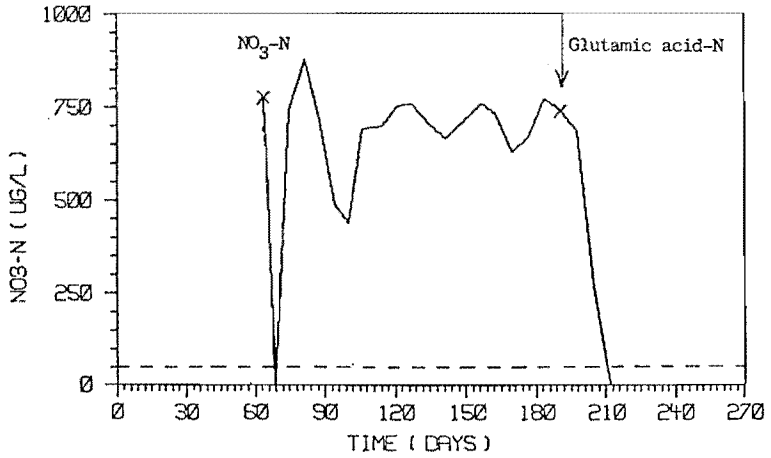


Figure 38. Nitrate nitrogen, microcosm D, dark. Arrow indicates input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

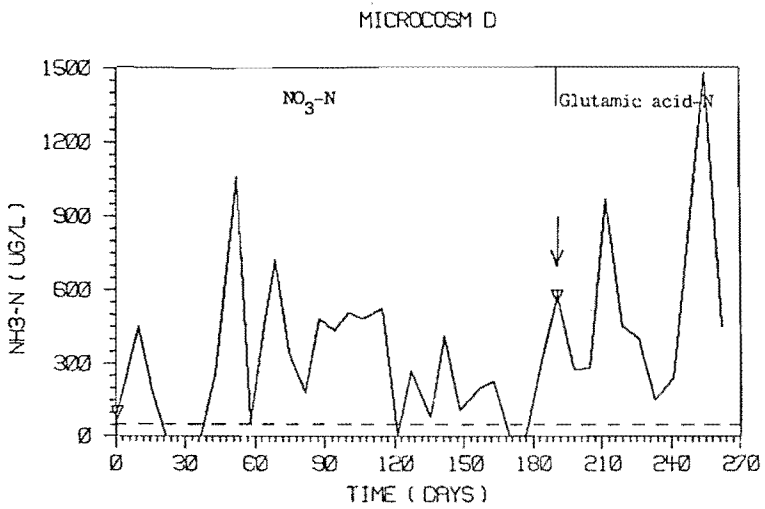


Figure 39. Ammonia nitrogen, microcosm D, dark. Arrow indicates input of new nitrogen source. Dashed line indicates lower sensitivity limit of test.

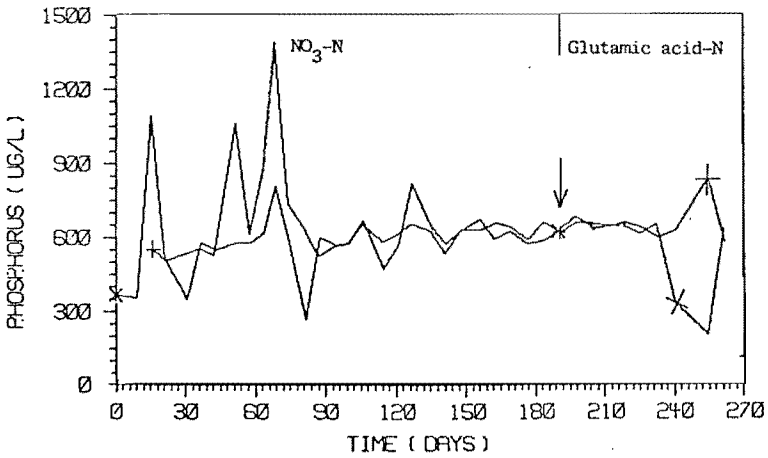


Figure 40. Total phosphorus (x) and orthophosphate (+), microcosm D, dark. Arrow indicates input of new nitrogen source.

Increased levels of carbon dioxide and ammonia from increased bacterial metabolism in lighted microcosm "L" probably are responsible for the peak in algae that occurred at day 233 shortly after the second peak in bacterial numbers (Figures 29, 30). The bacterial population stabilized for approximately two weeks while the dissolved oxygen concentration paralleled the increasing algal population. The levels of oxygen and algae then abruptly fell off as the bacteria once again began to increase.

A comparison of bacterial numbers in the illuminated and dark microcosms (Figures 30, 36) shows that even without the presence of an algal population the bacteria of the system were greatly stimulated by the glutamic acid nitrogen source. With nitrate, ammonia, and urea the bacteria were stimulated only indirectly by way of stimulation of an existing algal population. In contrast, glutamic acid is apparently able to directly stimulate bacterial metabolism. If the algae were able to utilize glutamic acid they were probably outcompeted by the bacteria for this organic source of nitrogen. In any case, bacteria were selected as the dominant biological component by this nitrogen source. Dark microcosm No. 4, however, did not develop a pink color as microcosm "L" did. The reason for this is seen in the bacterial concentrations that developed in these two systems. From the bacterial curve in Figure 30 (after day 216) it is apparent that a concentration of approximately 30-40 x 10^6 bacteria/ml is required to impart a pink color to the water. The maximum concentration of bacteria that developed in dark microcosm "D" is only one-fourth that required for visible color. Therefore, the low concentration of bacteria is the factor responsible for the lack of color in the dark microcosm ("D"). Even though the bacterial population was directly stimulated by the glutamic acid and responded with an immediate and dramatic increase in numbers, the concentration did not increase to the point of coloring the water pink. Development of a larger population of bacteria was probably limited by the low concentration of dissolved oxygen (Figure 35). Although there was a favorable nutrient source available (glutamic acid) the obligately aerobic halophiles did not have the oxygen present that they require for growth. In contrast, the dissolved oxygen concentration in the illuminated microcosm (Figure 29) was at an elevated level due to the algal population that had developed on the previous nitrate nitrogen source. The presence of this oxygen and the favorable nutrient source led to the development of the large concentration of bacteria in the illuminated microcosm ("L") which colored the water pink.

Nitrite production did not take place in microcosms "L" light or "D" dark. Nitrate was not detected in these systems either. In microcosm "D" a high level of nitrate had been present prior to the initial glutamic acid input. Within three weeks after nitrate input had ceased, the level of nitrate dropped to zero and was not detected again. This drop in nitrate was probably due to dilution by the continuing media exchange and some was possibly due to denitrification occurring in the low oxygen tension that existed in this microcosm. Only a few isolated bacterial strains from the northern Great Salt Lake have the ability to reduce nitrate and not all of these are able to produce nitrogen gas. Since there were no significant increases in the concentrations of nitrite or ammonia during the drop in nitrate levels, denitrification probably accounts for very little, if any, of this drop.

Ammonia production occurred in both microcosms "L" light and "D" dark, the highest levels being reached in the latter system (Figures 33, 39). Ammonia peaked in microcosm "L" light as the bacterial population increased. This was probably due to mineralization of the organic source of nitrogen and subsequent excretion of excess ammonia. The ammonia level then dropped as the algal population peaked and rose again as the bacterial population began to increase. The decreasing ammonia can be accounted for by the uptake of it under aerobic conditions and its subsequent production can be explained by increased mineralization of organic matter by bacteria and the resulting decrease in dissolved oxygen.

Peaks in ammonia production in microcosm "D" (Figure 39) coincided with peaks in the growing bacterial population (Figure 36). The absence of algae accounts for the close relationship between the ammonia and bacterial curves for this microcosm.

Ortho and total phosphorus trends in microcosms "L" light and "D" dark while they were on glutamic acid were identical to the trends observed in the other microcosms under different nitrogen source conditions (Figures 34, 40). Total phosphorus was in the form of orthophosphate in the dark column and was a sensitive indicator of biological activity in the illuminated column. Unlike previous results, however, orthophosphate in the illuminated system followed the trends in total phosphorus instead of remaining stable. Both forms increased gradually in the two systems as the bacterial populations increased. The response of the orthophosphate concentration to biological activity could be a result of the extremely high level of metabolic activity that was present in the systems on glutamic acid. This increased activity might have caused an increase in the rate of exchange of phosphorus with the sediment so that the orthophosphate level never had a chance to reach an equilibrium level, but was constantly in a state of flux as the exchange rate varied. Final biomass calculations for each of the microcosms (see section on Final Biomass Determination) showed that the use of the

glutamic acid nitrogen source resulted in a larger amount of biomass in both illuminated and dark microcosms than did the other nitrogen sources. This supports the suggestion that there was a higher level of biological activity in the microcosms on glutamic acid that might have led to increased phosphorus exchange rates.

Near the end of the study the total phosphorus concentration in dark microcosm "D" (Figure 40) quickly dropped far below its equilibrium level and then rose just as quickly back to this level. At the same time, the orthophosphate concentration increased and then decreased back to the equilibrium level. This occurred while bacterial numbers were increasing steadily in response to the glutamic acid nitrogen source. Since orthophosphate levels can never be greater than total phosphorus levels, the results are obviously in error. An analytical error involving switched samples or bad reagents could have accounted for this.

Gas Analyses

The gas phases of all six microcosms were analyzed for oxygen, nitrogen, carbon dioxide, methane, ethane, and ethylene three times during the course of the study (Table 4). The last gas analysis revealed the presence of an unknown hydrocarbon in all six systems so a propane standard was run along with the other standards (Table 4). The first analysis took place while microcosms "L" light, "D" dark, No. 1 light, and No. 3 dark were on nitrate and 27 days after microcosms No. 2 light and No. 4 dark had been switched from ammonia to urea. The second gas analysis took place after microcosm pairs No. 1 light and No. 3 dark, No. 2 light and No. 4 dark, and "L" light and "D" dark had come to equilibrium on nitrate, urea, and glutamic acid, respectively. The third gas analysis was performed just prior to the final biomass determination. The nitrogen sources for the three pairs of microcosms at this time were identical to those being used at the time of the second gas analysis. The second and third sets of data are the most useful since they were obtained when the microcosms were at steady state on their respective nitrogen sources. Gas data were not obtained while microcosms No. 2 light and No. 4 dark were on ammonia.

Oxygen concentrations were at elevated levels in the illuminated microcosms. The highest levels were recorded during the first analysis while illuminated columns "L" and No. 1 were on nitrate. These columns had actively increasing algal populations that accounted for the increase in oxygen. The oxygen level in lighted microcosm No. 1 decreased as the study continued, but not as much as in lighted microcosm "L." This decrease in oxygen in No. 1 could have been due to the metabolism of the bacterial population that was associated with the algal population. As oxygen and organic nutrients became more plentiful, the bacteria were favored and utilized an increasing percentage of the oxygen being produced by the algae. The presence of algal metabolism is seen not only in the elevated oxygen levels, but also in the depressed carbon dioxide levels in the illuminated microcosms.

The oxygen concentration in lighted microcosm "L" reached a lower level than that in lighted microcosm No. 1 since the glutamic acid in column "L" selected for bacteria as the dominant biological component. Bacterial respiration accounts for the decrease in oxygen. This also accounts for the decrease in oxygen in lighted microcosm No. 2 which was on urea during the second and third gas analyses. The respiration of the large and increasing bacterial population that was associated with the algal population during this time utilized a large amount of the oxygen being produced by the algae.

Table 4. Concentrations of the major (%) and minor (mg/l)[a] gas phase components.

	"L"			"D"			#1			#3			#2			#4		
	1[b]	2	3	1	2	3	1	2	3	1	2	3	1	2	3	1	2	3
O_2 %	79.2	56.4	45.4	15.5	13.3	13.9	78.8	67.6	53.9	17.4	16.5	18.8	75.7	65.6	44.9	19.7	20.0	19.8
N_2 %	18.7	41.6	52.6	82.3	84.3	84.5	20.1	31.1	45.0	81.2	79.9	72.8	22.3	32.4	52.2	79.4	77.2	78.1
CO_2 %	1.5	1.2	0.8	2.2	2.4	1.6	0.7	0.9	0.4	1.4	3.6	8.4[d]	1.2	1.2	0.9	0.9	2.8	2.1
CH_4 %	0.6	0.8	1.2	T[c]	T	T	0.4	0.4	0.7	T	T	T	0.8	0.8	2.0	T	T	T
CH_3-CH_3 (mg/l)	0.20	0.30	1.0	0.12	0.12	0.40	0.10	0.20	1.0	0.07	0.01	0.20	0.20	0.30	1.0	0.03	0.01	0.20
CH_2=CH_2 (mg/l)	0.60	0.60	2.0	0.10	0.10	1.0	0.50	0.50	2.0	0.05	0.01	1.0	0.60	0.60	2.0	0.02	0.01	1.0
CH_3-CH_2-CH_3 (mg/l)	-	-	2.0	-	-	1.0	-	-	2.0	-	-	1.0	-	-	2.0	-	-	1.0

[a] See text concerning unknown hydrocarbon fraction.

[b] Samples taken on 4-27-75 (1), 7-13-75 (2), 8-25-75 (3).

[c] Trace.

[d] An acid spill elevated the CO_2 level in this microcosm and the figure cited was determined by difference.

The oxygen levels in the dark microcosms remained rather stable during the study. The presence of oxygen was probably maintained due to oxygen being added with fresh medium and after repeated additions this level came to steady state. The bacterial populations in the dark microcosms remained at quite low levels during the study. If this community was not using up the introduced oxygen faster than it was being added, then the presence of oxygen in the gas phase of the dark microcosms can be accounted for.

Production of methane also occurred in the microcosms. It is worth noting that methane production only occurred in significant amounts in the illuminated microcosms. Since methane production is normally thought of as being an anerobic process, this might seem somewhat unusual. However, it must be remembered that the oxygen tensions in the illuminated microcosms were quite low. This, together with a significant amount of biomass production and bacterial metabolism, could lead to conditions favoring methane production since this process depends upon the accumulation of organic matter in sediments. In fresh water and in moderately saline conditions (upper salinity tolerance limit is unknown) populations of methane oxidizing bacteria which can remove methane from solution before it can enter the atmosphere are known to exist. However, this biochemical potential has not been demonstrated in bacterial isolates from the hypersaline northern end of the Great Salt Lake.

In contrast to the illuminated microcosms, the dark microcosms with their low level of biological activity exhibited almost no methane production. So in spite of the low oxygen levels in these dark microcosms, there was not enough organic matter to sustain methane production.

The data from Table 4 show an apparent increase in nitrogen gas in the illuminated microcosms during the study. However, this table indicates gas composition by percent. The apparent increases in the percent of nitrogen in the gas phases is actually a reflection of the decreases in the percentages of oxygen that took place in these systems. A decrease in the percentage of oxygen in these systems shows up as in increase of the same magnitude in the percentages of nitrogen and carbon dioxide.

Ethane, ethylene, and propane were also identified and measured in the microcosm gas phase. The production of each of these compounds was greater in the illuminated systems than in the dark systems. This indicates that, as with methane, the production of these hydrocarbons was greatest in those systems supporting the most biological activity.

By comparison with a known standard, propane was tentatively identified as being part of the unknown hydrocarbon fraction. However, two unidentified hydrocarbon peaks, other than the propane peak, were also present. This unknown fraction is quite significant since it accounts for 20-60 percent of the hydrocarbons in the microcosm gas phase. This is about equal to the amount of methane produced. The source of this unknown hydrocarbon fraction may have been phosphatidyl glycerophosphate lipid, the dominant lipid present in red halophilic bacteria. Kaplan and Baedecker (1970) found biochemical remains of these organisms in the reducing sediments of the Dead Sea. Natural breakdown products of the phosphatidyl glycerophosphate lipid from halophilic bacteria proved to be identical to certain organic residues isolated from Dead Sea sediments. Thus, dihydrophytol, phytanic acid, and/or phytane produced as a by-product of halophilic lipid degradation may account for the unknown hydrocarbon fraction in the microcosm gas phase. Other possibilities include isopentene or isopentane which may be involved in the synthesis or breakdown of the phytanol moiety of the bacterial lipids.

Final Biomass Determination

At the conclusion of the study a biomass determination was performed on each of the six microcosms. Direct microscopic counts were made to determine the total planktonic and attached populations of bacteria and algae, calculations of the densities of attached populations were made, and the total biomass of each microcosm was determined. The weights of individual algal and bacterial cells were calculated from the densities and volumes of these organisms (see MATERIALS AND METHODS—Final Biomass Determination). These weights were then used in the determination of total biomass. The results of this final biomass determination are found in Tables 5, 6, 7, and 8. It should be noted that, at the time of this final biomass determination, only nitrate, urea, and glutamic acid were being added to the columns. Ammonia inputs had been discontinued when urea inputs were initiated. However, it can be assumed on the basis of previous discussion that results with ammonia nitrogen paralleled results observed on nitrate nitrogen, the direct counts on ammonia being lower.

Tables 5 and 6 illustrate that in the illuminated microcosms 80 percent of the total bacteria in the systems were attached while in the dark microcosms 90 percent of the total bacteria were attached. In addition, 98-99 percent of the algae in the illuminated microcosms colonized surfaces. These figures, along with the population densities/cm^2 (Table 8), indicate that the attached populations of organisms accounted for most of the biomass and biological activity in the microcosms.

Results of the total biomass determinations are listed in Table 7. These figures represent the biomass that existed in each of the microcosms at steady state on a particular source of nitrogen. A comparison of the biomass produced on nitrate, urea, and glutamic acid indicates that glutamic acid accounted for the greatest production of biomass with nitrate and urea producing less. In fact, urea accounted for less biomass than did nitrate. These results correspond with observations made in previous discussion. Glutamic acid seemed to be responsible for the greatest resurgence of biological activity in the microcosms. This response was seen mainly in the stimulation of the bacterial population, a response that changed the illuminated system's color from green to pink. As was noted, this apparently resulted from a selection for the bacteria over the green alga by the glutamic acid.

The second highest level of biomass was produced in microcosms on nitrate nitrogen. The illuminated microcosm of this pair was green due to the algal population that had developed. The gradual development and stabilization of this population, when compared to the quick response of bacteria to glutamic acid, probably accounts for the smaller biomass production that resulted on nitrate. Also, biological activity in the microcosms on glutamic acid was due to bacterial metabolism while that in the illuminated microcosms on nitrate was due primarily to algal metabolism.

Urea produced even less biomass than nitrate. It was pointed out earlier that bacteria in this system only seemed to respond to urea when in the presence of algae, but even the algae in this system did not respond well to urea. Algal metabolism was seen to increase while algal numbers remained low. This increase in algal metabolism was followed by an increase in the bacterial population, but this increase in bacteria was limited by the fact that the algal population itself was being limited. So in spite of increased algal metabolism and a concomitant increase in bacteria, the amount of biomass produced was limited by the fact that bacteria were not stimulated directly by urea and algae were limited by some other factor.

Hydrogen sulfide was detected (by odor) in the sediment cores taken from all six microcosms. Illuminated microcosms "L" and No. 1 had the strongest indication of H_2S while microcosms No. 2 light and Nos. 3 and 4 dark had only slight amounts in comparison. This suggests that the sediments of all six microcosms were anaerobic. Colored layers within

Table 5. Planktonic organisms.

Column	Last N Source	Algae/ml		Bact./ml
		Red	Green	(x 10^6)
"L" light	Glut.	0	10,000	52
"D" dark	Acid	0	0	8
#1 light	NO_3	1,000	11,000	33
#3 dark		0	0	3
#2 light	Urea	740	2,300	34
#4 dark		0	0	7

Table 6. Attached organisms.

Column	Last N Source	Algae/ml		Bact./ml
		Red	Green	(x 10^6)
"L" light	Glut.	0	17,000	200
"D" dark	Acid	0	0	69
#1 light	NO_3	1,400	570,000	160
#3 dark		0	0	52
#2 light	Urea	2,900	180,000	160
#4 dark		0	0	53

Table 7. Algal, bacterial, and total biomass in the microcosms.

Column	Last N Source	Algae (g)	Bacteria (g)	Total (g)
"L" light	Glut.	0.07	1.96	2.03
"D" dark	Acid	0.00	0.41	0.41
#1 light	NO_3	0.46	1.34	1.80
#3 dark		0.00	0.24	0.24
#2 light	Urea	0.14	1.37	1.51
#4 dark		0.00	0.34	0.34

Table 8. Algae and bacteria per cm^2.

Column	Last N Source	Algae (x 10^3)	Bacteria (x 10^6)
"L" light	Glut.	6.29	74.33
"D" dark	Acid	0.00	25.52
#1 light	NO_3	212.80	59.54
#3 dark		0.00	19.23
#2 light	Urea	67.64	60.28
#4 dark		0.00	19.75

the sediments were observed only in illuminated microcosm No. 2 and dark microcosm No. 4. These had been on a urea source of nitrogen for approximately three months. The layers in microcosm No. 2 were slightly below the sediment surface. They were cream colored with green and pink tinges in them and occurred in patches. Dark microcosm No. 4 had a single, seemingly continuous, black layer beginning about 2 mm below the light grey sediment surface. This black layer was about 2 mm thick. Below this black layer the sediment was light grey. The light colored patches in the sediment from microcosm No. 2 were probably aerobic since the green tinge was most likely due to algae. The H_2S detected in this microcosm's sediment core was probably produced in anaerobic zones either between or under the lighter patches of growth. The black layer in dark microcosm No. 4 was probably a reduced, anaerobic zone responsible for the H_2S detected in this column's sediment core.

RESULTS AND DISCUSSION OF LAKE STUDIES

The Biota of the Lake

Nissenbaum (1975) reports that only bacteria and a chlorophyton alga, *Dunaliella viridis,* are found in the Dead Sea (a body of water with a chemical load similar to the Great Salt Lake) with no higher forms (e.g. invertebrate grazers) present. The Great Salt Lake on the other hand has several algae, a number of bacteria, several protozoa, a brine shrimp and two or three species of brine fly in the north arm. The south arm has the same organisms plus a wider variety of bacteria, algae, and protozoa.

The Algae

The primary producers of the lake are algae of the genus *Dunaliella.* In the south arm, as in the Dead Sea, *D. viridis,* a small bright green highly motile flagellate, is considered the principal planktonic alga reaching levels of 200,000 ml^{-1} in late spring (Stephens and Gillespie, 1972, 1976).

Two other small unidentified flagellated green pigmented algae occur in significant numbers in the south arm but have not been observed in the north. Algae belonging to the cyanophyta grow in reef-like bioherms or biostromes in the south arm. These consist of calcium carbonate precipitated around cells of *Oscillatoria* sp. and *Coccochloris elebans* (Aphanocapse halophytica (Brock, 1976)) to form dome-like structures (Carozzi, 1962). The bioherms also occur in the north arm but may or may not be active due to the high salinity. They are at present under about 3 meters of water and unavailable for study. In two years of sampling the north arm, only one water sample has shown cyanophyte filaments tentatively identified as *Nodularia* sp. These probably washed in from the south end where they are also observed abundantly in the plankton, or from springs along the margin of the lake. There is no evidence of any cyanophyta in the aufwuchs or benthos around the north arm margin although *C. elabans* (Kaoetal, 1973) has been isolated from water of comparable solids content. Brock (1976) reported recovering this organism from the crust of a low salinity (10 percent solids) spring emerging from what appears to be an old bioherm at Rozel Point. Whether this organism occurs generally in the north arm deserves further work.

In the north arm, *D. viridis* appears at low levels in the plankton but is generally most common on the underside of wood, rocks, or debris along the shallow margin (15 cm or less deep) where they are out of the direct sunlight and where the temperature of the water is fairly high. Over the two year period of sampling only once did the planktonic *D. viridis* population exceed 300 ml^{-1}. In about half of the samples no *D. viridis* cells were found. Instead, the north arm is populated by a rather large (16 μm diameter) red pigmented alga with two flagella, which is sluggishly motile. This alga was found exclusively in the plankton and only rarely in the shallow habitat of the green form. The red alga was never absent from the plankton and generally ranged from 200 to 1000 ml^{-1} with peak blooms of 3-10,000 ml^{-1}. The red pigmented alga does not bloom uniformly in the lake but in huge patches.

The red colored alga has been compared morphologically with *Dunaliella salina* strains 1644 and 200 supplied by the Indiana University Algae Collection. The Great Salt Lake alga is considerably larger and forms much more carotenoid pigment than the Indiana strains. Chlorophyll pigment is completely masked by the abundant orange-red carotenoid pigment. The organism does not appear to have a cell wall although an occasional cell will be observed to have a layer of rectangular plate-like bacteria covering the surface. These can be observed to slough off leaving a bald patch behind. The cell is under extreme osmotic tension. When a drop of distilled water is placed next to a drop of lake water on a microscope slide, it causes the algal cells to suddenly explode leaving only small reddish particles and debris. Merely touching cells growing on an agar surface with an inoculating needle causes them to lyse. There is no evidence of a cell wall in the debris. Until further work can be done, this is tentatively considered to be a strain of *D. salina.* A similar organism has been described (Nixon, 1969) from salterns.

Ben Amotz and Avron (1973) with *D. parva* and Borowitzka and Brown (1974) with *D. viridis* have demonstrated that these organisms form high intracellular concentrations of glycerol which apparently maintains osmotic equilibrium and prevents entry of salt into the cell. By extension, *D. viridis* and *D. salina* in the Great Salt Lake would also

form considerable glycerol. With recent isolation of an axenic strain of *D. salina,* this assumption will be tested.

Based on the microcosm studies *D. salina* appears to be stimulated by ammonia in preference to nitrate while *D. viridis* appears just the opposite, also suggested by Stephens and Gillespie (1976). These observations are now being corroborated by axenic culture studies.

One further observation is the effect of low temperature on the algae. When the temperature of the lake drops to 0 C or below, it appears to have little effect on motility of either alga and some are continuously present and active (motile) in the lake throughout the winter months. Round cyst-like cells of the *D. salina* increase in numbers during the cold season especially on the lake bottom but motile forms are always present. These round forms are presumed to be some kind of a dormant state. A single experiment with north arm water showed continued activity of both algae for three weeks at -6 C in the dark with no loss in numbers. At -18 C all activity ceased in two days. Whether the algae can reproduce at sub-zero temperatures has not been determined.

The Bacteria

The bacteria of the north arm are the most conspicuous members of the community. These occur in such vast numbers that the water becomes a rosé wine color as a result of their carotenoid pigments. This color has often been attributed to the red colored alga which does not usually occur in sufficient numbers except in localized "bloom" patches to impart the deep wine color of the lake. The entire north arm is red being most visible from the air but very apparent from the shore. Visitors have often referred to the north arm as the Red Sea, literally. A Skylab color composite photograph (Post, 1975) shows the remarkable color of the north arm in contrast to the more usual blue-green color of the south.

Two methods of determining bacterial numbers were used, a direct microscopic count and a surface plate method for viable bacteria. Direct counts were made using a calibrated ocular, a hemacytometer and a phase microscope at 500X. Most cells are fairly large and obviously bacteria. If anything the counts are underestimates since very small rod and coccoid organisms are easily overlooked.

Direct counts taken vertically in the water column at sample sites over the whole north arm (Figure 3) indicate a fairly uniform distribution of bacteria throughout the lake and over the entire year (Figure 41 and Appendix C). An increase in numbers was observed during algal blooms and near the sediment surface. Bacterial numbers varied between 40 x 10^6 and 100 x 10^6 ml^{-1} except near the sediment surface and after algal blooms when counts ranged from 100 x 10^6 to 240 x 10^6 ml^{-1}. These counts are more than enough to account for the red color of the lake water. Direct counts on the south arm show less than 10^6 bacteria per ml. Kaplan (1970a) reports direct microscopic counts of the Dead Sea between 8.9 x 10^6 ml^{-1} in November and 2.3 x 10^6 ml^{-1} in March and decreasing with depth. Since he makes no comment about color, it is presumed that this level is insufficient to produce a red colored water.

Viable plate counts indicated a sizable population of living bacteria in the north arm ranging from a high of 5.5 x 10^6 ml^{-1} in summer to a low of 5.4 x 10^4 ml^{-1} in winter. As with most aquatic studies there is a sizable difference between direct and viable counting methods. The viable method is 10 to 20 times smaller than the direct method which is considered good for such comparisons (Collins, 1963). The medium used could probably be improved and the counts increased. However, increasing the salt to more closely approximate the lake lowers the bacterial count. Addition of agar to the high salt medium or allowing the medium to dry too long, lowers the aqueous activity below that of the lake itself and presumably inhibits growth due to too little free water.

The decline of viable bacteria during the coldest time of the year suggests that extremely cold temperatures have an adverse effect on the bacteria. This has been observed during two winters and is now being followed up in the laboratory. Attempts to grow lake bacteria at temperatures below 10 C have been unsuccessful.

The optimum growth temperature of most of the lake isolates studies has proven to be around 45 C with extremes of 50 C and 37 C. Preference for high temperature is seen in the lake where massive bacterial growth occurs on wood, tar balls (especially), and other organic debris along the lake margin where the water temperature is near 40 C in the spring and summer. The surface of these objects is covered with a heavy visible red slime.

Viable bacteria are found in uniform numbers horizontally and vertically over the entire lake and even in the bottom hypolimnion where oxygen is absent. Recent investigations (Danon and Stoeckenius, 1974) have demonstrated that the halobacteria have a rhodopsin-protein in the cell membrane capable of generating ATP in the absence of oxygen and in the presence of light. Light levels were determined in the deepest part of the lake on February 11, 1976. The Secchi disk depth was 1

meter and there are 10 lumens at 8 meters. It is presumed that some light does reach the bottom since a few motile algae were found there.

Since counts were available for the various organisms, biomass calculations were made (Table 9). Certain assumptions were made in the calculations. Since the bacteria settle out upon standing, it assumed that they were more dense than the lake (1.230), so a density of 1.300 was selected. Thus an average lake bacterium would have a weight per cell of 4.3×10^{-12} g. The two algae were assumed to have a density of 1.13 which is that of a 6.2 m glycerol solution. The average weights for *D. salina* and *D. viridis* respectively were 2.4×10^{-9} and 6.8×10^{-10} g per cell. The total bacterial biomass in the lake is immense. The average bacterial biomass far exceeds the usually observed algal biomass and is more than 10-fold larger than the maximum observed algal bloom, Table 10. Even reversing the densities in the biomass calculations would not change this relative difference more than a small amount. The biomass of the viable bacteria is very close to the maximum algal bloom biomass and under non-bloom conditions would exceed it.

A number of individual bacteria have been isolated from the lake and all shown to be *Halobacterium* or *Halococcus* species possessing the special membrane lipid and the carotenoid

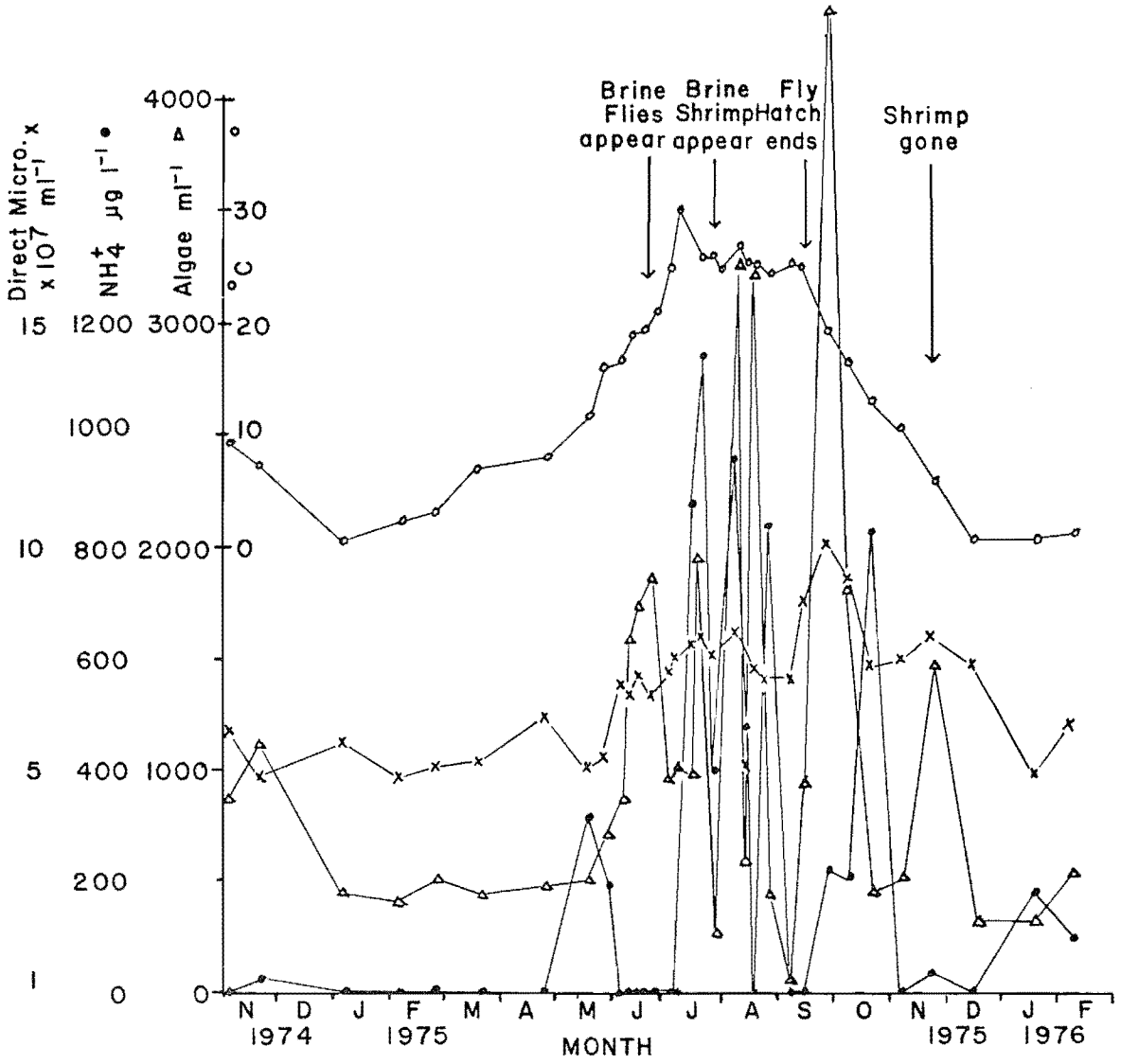


Figure 41. Seasonal changes in algae, direct microscopic count, ammonia, temperature and invertebrate appearance for the period 1974-1976 in the north arm of the Great Salt Lake. (Raw data in Appendix C.)

bacterioruberin (Buchanan and Gibbons, 1974; Larsen, 1967). Some isolates are capable of digesting chitin and some the cell wall of normal bacteria *(Escherichia coli)*. They have broad biochemical capabilities including acid production from carbohydrates by some. Halotolerant organisms are presumed to be present but have not been studied.

One additional factor in the north arm water is the presence of a variety of phages specific for the Great Salt Lake bacteria. Two different halophages specific for lake bacteria and five for strains of *Halobacterium halobium* (provided by the National Research Council of Canada) have been isolated. What role these play in the bacterial ecology must still be determined but it may be important because of the unusually large number of bacteria present.

The Protozoa

Although a number of reports in the literature describe protozoa in the Great Salt Lake prior to the separation of the lake into two parts, none have been found to be reported from systems as saline as the present day north arm. In August 1974 a sediment and water sample was brought back to the laboratory and placed in a 50 cm long x 26 cm wide x 30 cm deep aquarium. Invertebrates were removed and the aquarium has been maintained at 28 C (summer lake temperature) with the water surface 25 cm below two 40 watt daylight fluorescent bulbs with an 18 hour on, 6 hour off cycle. This aquarium has not been fed any chemicals or nutrients since establishment. Only demineralized water has been added to replace that evaporated.

Within six months, the aquarium was heavily colonized with bacteria and algae and protozoa had appeared. These were generally small and attempts to cultivate them outside the aquarium have not been successful. Several types have been observed: 1. A small 4 x 5 μm biflagellated, wedge shaped protozoan highly refractile under phase contrast; 2. A long thin 16 x 4 μm cell with two flagella often intertwined; 3. Two amoeba, one small about 4 μm in diameter forming one pseudopod at a time, and a much larger form, about 16 μm or more in diameter. Both are commonly found in the plankton of the aquarium associated with large clumps of bacteria; and 4. An organism which looks very much like a *Tetramitus* sp. (Kudo, 1946). It is possible that the long flagellated form and the large amoeba are two states in the life cycle of a single protozoan (Dimastigamoeba-like) since one amoeba was seen with flagella-like appendages. This observation has not been confirmed. *Tetramitus* species are also described as having an amoeboid stage. The protozoa occur in rather large numbers (Table 11) and are presumed to be present in the lake but too low in concentration to be observed regularly. A ciliate has been observed in the microcosms lighted and fed regularly but not in the aquarium or the lake. This also is presumed to have come originally from the lake. During three years of examination, no protozoa had been observed in any

Table 9. Physical dimensions of Great Salt Lake north arm organisms as determined from lake samples.

	Average Dimensions μm	Shape	Volume μm^3	Density g cm^3	Weight g $cell^{-1}$
Bacteria	1.2 x 3.0	cylinder	3.3	1.30	4.3×10^{-12}
Dunaliella salina	16	sphere	2143.57	1.13	2.4×10^{-9}
D. vividis	12 x 8	oblate ellipsoid	602.88	1.13	6.8×10^{-10}

Table 10. Concentration and biomass of organisms in the north arm of the Great Salt Lake.

Organism	No. m^{-3}	Biomass g m^{-3}	North Arm Metric Tons[a]
Direct microscopic counts bacteria (average value)	7×10^{13}	300	2.2×10^{6}
Viable bacteria (Max.)	5×10^{12}	22	1.6×10^{5}
Dunaliella salina (Max.)	1×10^{10}	24	1.8×10^{5}
D. viridis (Max.)	2×10^{9}	1.4	1.0×10^{4}
Brine shrimp[b]	1	0.1	7.4×10^{2}

[a]Volume of north arm 7.4×10^{9} m^3 based on Utah Geological Survey data.

[b]Estimated number and weight. Data not available.

lake samples. However in May 1976 one sample showed a single specimen bearing a strong resemblance to a *Uronema* sp. One species (Bick, 1972) is described as tolerating up to 35 gl^{-1} of salts. At that time the lake was 1.198 specific gravity with a solids content of 280 gl^{-1}.

The Brine Shrimp

Artemia salina, the brine shrimp, occurs throughout the world in saline waters. Prior to separation, the Great Salt Lake served as a massive breeding ground for the brine shrimp and eggs were harvested commercially. Since division of the lake production of eggs in the south end has been steadily going down (Sander's Brine Shrimp Company, Ogden, Utah, personal communication) while the north arm has become relatively non-productive. Brine shrimp do occur in the north arm but their origin is obscure. Numerous attempts to hatch eggs in north arm water (including the aquarium described above) have been unsuccessful. Nixon (1969) reports a similar failure in hypersaline climax communities. Clegg (1964) reports that *Artemia salina* exists by producing enough glycerol to raise its internal osmotic pressure above that of the water environment. He also reports a failure to exist in 2 m NaCl or higher. Existment may be beyond the capabilities of the organism in north arm water which is about 4.6 m NaCl. Still, brine shrimp do appear in the north arm (Figure 41) in late June or early July, several months after their appearance in the south arm. Several explanations can be suggested. Fresh rainfall on a calm lake provides a lower salinity surface layer where eggs could hatch. The lake is rarely this calm but such conditions have been observed several times in the lake, most recently in February 1976 when approximately a 10 cm layer of 1.100 specific gravity water overlayed water of 1.210 specific gravity (Figure 42, Appendix C). Attempts to hatch eggs under these conditions in the laboratory have been unsuccessful. Another possibility is margin areas where springs of low salinity (10 percent solids) enter the lake. Such water floats on the surface for some time and eggs could hatch in the saline gradient established. The few springs entering the lake, the rapid mixing by wind, and the large number of shrimp observed would make this little more than a possibility. The most likely source of the shrimp is the water flowing from the south end of the lake. Shrimp hatch in the south arm water and flow into the north arm as nauplii or adults. There is also the distinct possibility that once acclimatized, summer eggs may hatch or live nauplii be produced (Porcella and Holman, 1972) by the females. The fact that no brine shrimp have been reported in the Dead Sea suggests that they are unable to hatch there either due to the high salinity or the high magnesium level. This last would seem important since there are presumably areas of the Dead Sea where the salinity should allow the shrimp to hatch.

The preferred diet of the north arm brine shrimp appears to be the algae although the large bacterial population may also serve as food (Bond, 1933). The massive growth of *D. viridis* in the aquarium (Table 11) may indicate that the shrimp use this alga preferentially when available and *D. salina* only when few or no *D. viridis* are around. However, the shrimp appears to follow blooms of *D. salina* and is often associated vertically only with high *D. salina* populations. Feeding preference studies with the two algae are planned. Figure 42 (and Appendix D) illustrate a typical midsummer and midwinter depth profile for algae, oxygen, temperature, and density. The algal peak occurred in the summer at or near the top of the thermocline and the maximum concentration of brine shrimp occurred at the same depth. It is not known whether the algae or the shrimp rise or fall in the water column in response to diurnal light

Table 11. Standing plankton crop in laboratory aquarium containing Great Salt Lake water (north arm), unfed, after 18 months.

Organism	No. ml^{-1}	No. m^{-3}	Biomass $g\ m^{-3}$	Times Larger Than Lake[a]
Bacteria	7×10^{8}	7×10^{14}	3000	10.0
Dunaliella salina	1×10^{4}	1×10^{10}	24	1
D. viridis	2.5×10^{5}	2.5×10^{11}	230	160.0
Protozoa				
Amoeba, small	3.2×10^{3}	3.2×10^{9}	-[b]	-[c]
Amoeba, large	5.4×10^{3}	5.4×10^{9}	-	-
Long flagellate	1.6×10^{4}	1.6×10^{10}	-	-
"Wedge" shaped	2.1×10^{5}	2.1×10^{11}		

[a] See Table 10.

[b] Not calculated. Based on relative size the entire protozoa group would approximate that of the green alga.

[c] Protozoa not observed in lake samples.

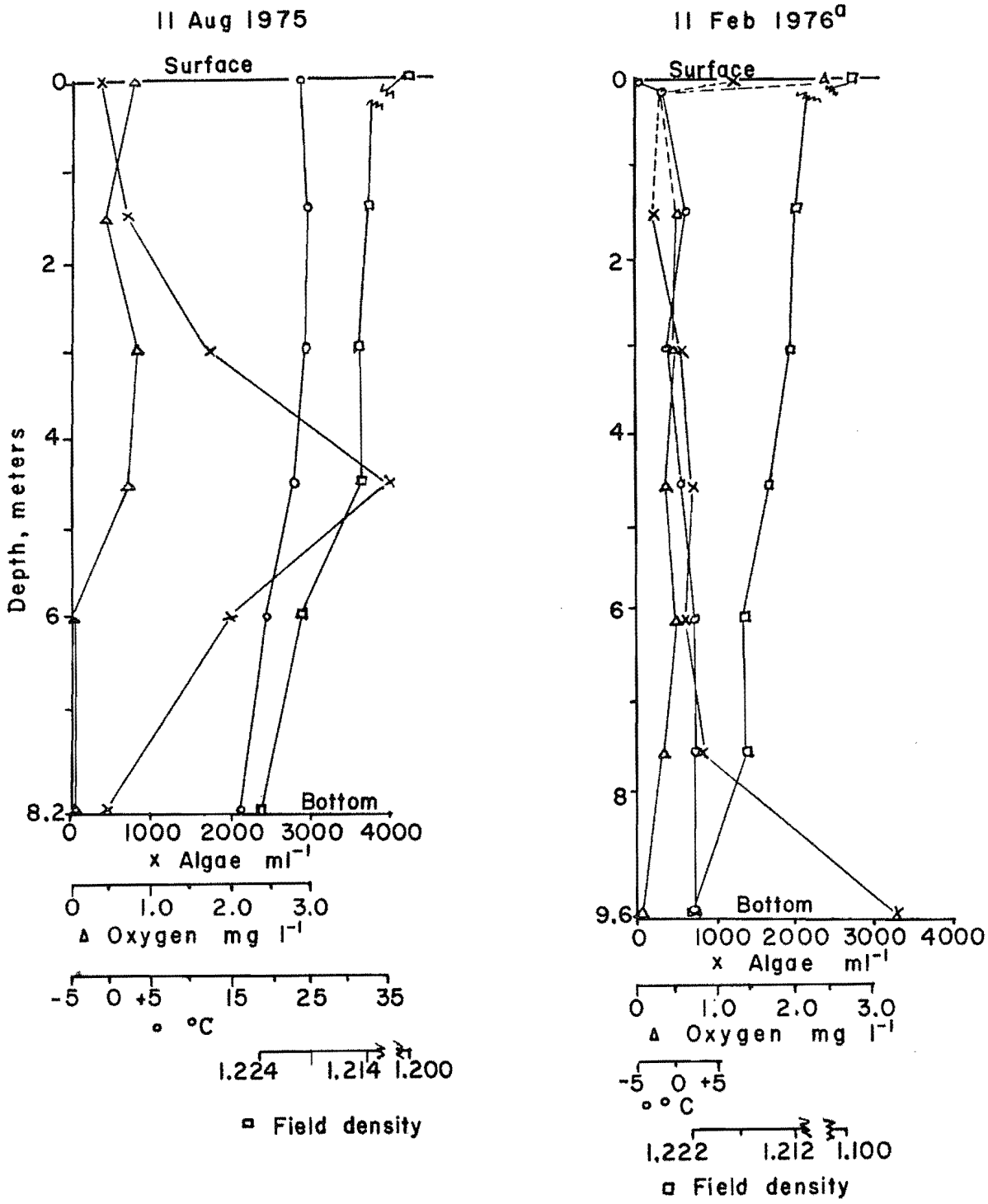


Figure 42. Midsummer and midwinter depth profiles at Great Salt Lake north arm station LVG2 showing oxygen, temperature, algae, and density. (Raw data will be found in Appendix C.)

[a]Surface ice from recent snowfall made boating difficult. The February site was about 3 km south of the usual position. The surface 10 cm had a very low density, 1.100 while below 10 cm the density was 1.210. The top layer had a temperature of -5 C which rose to 0 C at 10 cm. The algae of the surface were entirely *D. viridis* or other south arm types. Below 10 cm no green algae were observed. The surface layer is believed to be less dense south arm water (density 1.100) floating on the top of the lake. The bottom sample consisted of hydrated sodium sulfate (see text). The direct microscopic and total phosphate levels were very high. All algae on the bottom were the red form, mostly in the dormant state carried down by the sedimenting sulfate. Some bacteria may have been carried down also.

levels or whether they are trapped by density gradients. As indicated in Figure 41 oxygen levels were generally high where algae counts were high. Station LVG2 (Figure 3) is on the eastern edge of the water circulation moving north from the causeway.

The low oxygen level of the north arm water has a pronounced effect on the brine shrimp. Virtually all are a bright red color possibly due to an increase in hemoglobin production as a result of the low oxygen levels (Gilchrist, 1954) or due to uptake of carotenoid from the red alga diet (Gilchrist and Green, 1960). Electron microscope sections of the brine shrimp gut wall shows the presence of intracellular bacteria either as symbionts, perhaps aiding nutrition, or as pathogens (Post and Youssef, in preparation). Since the cells with the symbionts are involved in salt transport and balance and are bathed in the high salt water (Croghan, 1958), it is possible that these bacteria come from the lake.

The brine shrimp disappear from the north arm by November when the water temperature falls below 6 C, the lowest temperature where they have been observed.

The Brine Fly

Very little attention has been paid to the brine flies in the north arm. Several species of the genus *Ephydra* live in the lake. Huge clouds of flies cover the water and shores along the margin of the south arm. The north arm is not as productive as the south arm. The reasons for this are not clear since fly eggs have been observed to hatch in north arm water. Sufficient food may be a limiting factor or the proportion of eggs hatching in high salinity may be low. The diet of the larvae is unknown but could include the algae (Figure 41), the bacteria, and the dead brine shrimp. It is possible that the flies and the shrimp compete for available algae. Flies emerge well before the brine shrimp appear and they overlap for a considerable period during midsummer (Figure 41). Discarded pupal cases account for a large part of the lake chitin and organic matter. The fly is the only member of the community which leaves the lake. Most lay eggs on the water surface and die, however many fly or are blown inland and die away from the lake. No brine flies are reported from the Dead Sea (Nissenbaum, 1975). This may be due to the high magnesium level there rather than the salinity level alone.

Ducks, seagulls, pelicans, and other water birds are often observed on the lake but do not apparently feed on the shrimp or the flies in the north arm. These birds are reported to feed on the invertebrates in the water in the south arm and the bird population is considerably higher there. Aside from some feces, an occasional dead bird, or a dropped fish, the bird contribution to the microbial ecology would have to be considered minimal.

Community Metabolism and Lake Chemistry

Nixon (1969) described salterns as chemically rich and of low diversity. The Great Salt Lake fits this description rather well. The dehydrated remains of an extremely large fresh water lake which has cycled numerous times, the Great Salt Lake is rich in organic matter as evidenced by the very high dissolved organic nitrogen load (Table 12). Insoluble organic matter in the form of flies and pupal cases, dead brine shrimp and egg masses is also a large pool but no quantitative measures have been made. Total dissolved organic carbon was not determined because of fouling of the analyzer by the high solids content. If one assumes ratios of 106 C:16 N:1 P in organic matter (Kramer et al., 1972), the dissolved organic carbon would be about 40 mg/l^{-1}. If it is assumed that much of the organic carbon is in the form of glycerol then the ratios of Deevey (1973) might be more appropriate, 1480 C:16 N:1.8 P, giving a carbon level of 555 mg/l^{-1}. Nissenbaum (1975) indicates a ratio of dissolved organic carbon; dissolved organic nitrogen of 25 in the upper 45 m of the Dead Sea where most of the biomass occurred. Applying this ratio to the north arm of the Great Salt Lake gives 150 mg l^{-1}. The actual level is not known but it is presumed to be high because of the observed foaming in the lake. The north arm water foams heavily when disturbed by wind or other physical factors. A power boat produces a wake of bubbles and foam that persists for hours or until dispersed by wind. The high solids content raises the surface tension greatly but this is counteracted in part by the presence of organic matter which results in foaming. Simulated lake water made from reagent grade chemicals does not foam. The microcosm columns of simulated north arm water with microorganisms (no invertebrates) and sediment from the lake were fed ammonium chloride as the sole source of nitrogen. No nitrate or nitrite

Table 12. Concentration ranges of biologically important chemicals in the north arm of Great Salt Lake, 1973-1976.

	Low Value	High Value
o-PO_4 ($\mu g/l$)	290	1600
Total P ($\mu g/l$)	630	4000[a]
NH_3 ($\mu g/l$)	0	1080
Soluble-N (mg/l)	3.0	6.1
Total-N (mg/l)	4.3	7.2
Oxygen (mg/l)	0.00	1.70

[a]Rarely over 1100 except near sediments.

was detected over a year and a half period. Over a two year period no nitrite or nitrate has been found in the north arm itself. Nissenbaum (1975) reports very low levels of both ions in the Dead Sea. Most bacteria isolated from the north arm can reduce nitrate to nitrite and a few can further reduce nitrite to nitrogen gas. Possession of these enzymes suggests frequent encounter with the substrate but perhaps only in waters of much lower salinity. Both the substrate (Stephens and Gillespie, 1976) and the bacteria occur in the south arm, for example. Utilization of nitrate as fast as it is produced by either *D. viridis* or the bacteria is also possible; however, the inability to detect any under controlled microcosm conditions suggests that extremely halophilic or extremely halotolerant nitrifiers may not exist.

All attempts to demonstrate a potential for nitrogen fixation by acetylene reduction have been unsuccessful. Samples included benthos, anaerobic decomposing brine shrimp, algal-bacterial mats from rocks and pure cultures of bacteria. Unless blue-green algae with heterocysts are active in the bioherms, it appears that no nitrogen fixation takes place.

Ammonia appears to be the key to many of the events observed in the north arm. It is used especially by the red alga as the microcosm studies show. *Dunaliella viridis* prefers nitrate but will use ammonia (Stephens and Gillespie, 1976). The bacteria produce ammonia in laboratory culture from organic nitrogen but are reported not to use it (Onishi and Gibbons, 1965). Free ammonia is generally in very low concentration in the lake (Figure 41) and is detected principally in the early spring and during the summer. Little or no ammonia is detected in the winter due to low temperature effects on metabolism and growth. What is produced is undoubtedly promptly used by the remaining active algae. The tendency of *D. salina* to become dormant may be a response to low ammonia levels rather than to cold. In April, when the water temperature reaches about 8-10 C (Figure 41) ammonia begins to be produced presumably by the bacteria from the soluble and insoluble organic pool. The halobacteria have been reported (Onishi et al., 1965) to use only organic nitrogen but this needs to be verified with lake isolates. The release of ammonia from the pool suggests that usable carbon is in short supply compared to nitrogen so that excess nitrogen is excreted, i.e. the C:N ratio is very low. If the dissolved organic carbon pool is assumed to be high, this suggests that it is some form not readily available to the bacteria or that contains a large excess of nitrogen. The increased biomass of the aquarium (Table 11) indicates that much of it is usable given enough time.

When the lake reaches a temperature of about 10-12 C, the bacterial population rises (Figure 41) followed by the algae and a decrease in detectable ammonia. After the appearance of the invertebrates at about 20 C, the algae decline and ammonia increases markedly. This increase in ammonia is possibly the result of the excretion of the uric acid (Meglitsch, 1967) or other nitrogenous wastes by the brine fly larvae and its subsequent breakdown by bacteria which are also increasing in numbers at this time. A few bacterial isolates from the lake have been shown to use uric acid. The appearance of brine shrimp in late July keeps the ammonia level fairly high. These organisms presumably excrete ammonia as a waste product, a characteristic nitrogenous waste of crustaceans (Chapman, 1971). After the fly hatch ends in September the algae bloom as do the bacteria, followed by an increase in ammonia until the brine shrimp disappear and the various groups return to a winter type population level. Based on our observations to date, the nitrogen cycle consists of an interconversion of organic matter and ammonia with ammonia as the factor limiting the growth of the algae.

Dissolved carbon is rather high in the lake, as indicated above, but there is also considerable carbon as well as nitrogen tied up in insoluble organic matter. Decomposition of this matter is extremely slow. The soluble inorganic bicarbonate pool available to the algae is fairly high (Table 1) and the sediments contain a very large deposit of carbonate (Eardley, 1966). The bacteria are the chief generators of carbon dioxide which probably never escapes from the lake. One possible loss of carbon to the system, in addition to adult flies lost inland, is methane. Methane, and smaller amounts of other gases, were evolved in all of the simulated lake microcosms but have not been measured in the lake itself. It is apparent that halophilic or halotolerant methane producers are found in the Great Salt Lake sediments. Hydrogen sulfide is also conspicuously present in these same sediments. Because of the high sulfate levels methane appears to be produced in the presence of sulfate contrary to the observations of Martens and Berner (1974). This remains to be verified, however. Whether any of the lake bacteria can use methane as a carbon source is an open question. The carbon cycle also appears to be fairly simple, an interconversion of organic carbon and CO_2 with some loss as methane. This loss may be counterbalanced by solution of atmospheric CO_2.

Kaplan and Friedmann (1970b) have speculated that phytane in sediments and petroleum originating from hypersaline environments may be derived from the cell membrane lipid of halophilic bacteria. These bacteria, then, may play some role in the origin of oil. Both the Dead Sea and the Great Salt Lake have deposits of tar-like petroleum which contribute to the organic carbon pool of the lakes. In the Great Salt Lake this tar-like oil oozes into the lake along a fault line at Rozel Point (Figure 3) from a formation

believed to be about 1000 meters below the lake bottom (Utah Division of Natural Resources, personal communication). A number of attempts have been made to mine this oil. Old drill holes and casings leak oil adding to the natural ooze. The oil floats and is pushed shoreward by the prevailing winds (from the west) where it adheres to rocks and forms clumps of tar along the beaches. These clumps and masses adhering to the rocks serve as a favored habitat for the growth of bacteria. Although it has not been tested yet, the extreme halophiles may be an important factor in the metabolism of petroleum in hypersaline environments.

Phosphorus (Table 12) does not appear to be in short supply. Analysis of the sediments indicates an extremely large pool of phosphate, several hundred times that available in the water column. Most of the phosphorus in the water column is particulate and concentration varied considerably. The dissolved orthophosphate remains fairly constant with time throughout the lake and may represent an equilibrium between the sediments, growth, and decomposition.

The north arm is rich in organic matter and is constantly supplied from the south arm with new organic material or the essential ions to make it. There appears to be little or no loss of carbon or nitrogen from the north arm. The question arises as to what the limiting nutrients may be. The laboratory aquarium sheds some light on this question as far as the lake is concerned. When held at 28 C for six months (and now a year and a half) the lake water can be seen to be extremely productive (Table 11). The bacteria and algae counts are one or more orders of magnitude above the highest levels observed in the lake suggesting that the biomass potential of the lake is much greater than actually observed. That it does not reach this potential is perhaps due to two things; the short summer available for growth and the grazing of the invertebrates.

The algae and the bacteria of the lake have fairly slow generation times. In the laboratory, the bacteria have a generation time of 6-7 hours under optimum conditions of temperature (40-50 C) and nutrient supply. In the lake itself, even in summer where conditions are far below optimum (30 C or less) the generation time would be considerably lengthened. Cold weather would extend the generation time even farther, ultimately stopping growth altogether in the winter.

Dunaliella salina also has a slow generation time (not yet determined) estimated to be between 1 and 2 days under laboratory conditions. It should be slower in the lake itself. *Dunaliella viridis* (Van Auken and McNulty, 1973) has an optimum generation time of 10 hours at 32 C in the laboratory and is no doubt slower in the lake. Any conversion of organic matter to a form usable by the algae would be dependent on temperature and its effects on the organisms, especially the bacteria.

Grazing by the invertebrates places a constraint on the population size of the algae thus limiting their organic output, presumably glycerol, and providing organic and inorganic nitrogen for the bacteria and algae respectively. In the aquarium (Table 11) where the effects of the invertebrates and cold temperature have been removed, the biomass has increased significantly as a result. In contrast to the lake, *D. viridis* grows to numbers never observed in the lake itself. The reasons for this are not clear but may be due to preferential grazing by the invertebrates or to better competition for ammonia with the *D. salina*. The microcosm studies show that *D. viridis* will grow on ammonia, although more slowly than on nitrate. Ammonia and the effect of temperature on the bacteria and the invertebrates would appear to be the limiting factors in the lake. The contrast with the Dead Sea is rather interesting (Nissenbaum, 1975). The organic nitrogen pool in the Dead Sea is about half that of the Great Salt Lake (and presumably the organic carbon is less also) but the ammonia level is 10 to 1000 times larger in the Dead Sea. The bacterial population of the Dead Sea is about one tenth the Great Salt Lake but the algal levels are comparable except for *D. viridis* in the former and *D. salina* in the latter lake. Both bodies of water appear to have a high nitrogen organic pool which is relatively resistant to bacterial attack. No doubt some of the difference between the two is in the total organic load but may also be due to the high magnesium level of the Dead Sea. Given the same climate as the Dead Sea, the Great Salt Lake would have a biomass approaching that of the aquarium lessened by the impact of an increased invertebrate population.

SUMMARY AND CONCLUSIONS

Summary

This study demonstrated that nitrate, ammonia, and urea were not capable of directly stimulating the bacterial populations that were present in the microcosms. Bacterial populations were stimulated or maintained at constant levels indirectly by these sources of nitrogen through the stimulation of an associated algal population. The increased levels of dissolved oxygen and organic matter that resulted from the stimulation of the algae produced quite favorable growth conditions for the bacteria. The most stimulating nitrogen source for the green alga of the system, *Dunaliella viridis*, was nitrate while the red alga, *Dunaliella salina*, responded most favorably to ammonia. This could account for the predominance of the red form during algal blooms in the northern arm of the Great Salt Lake. Chemical analyses of water from this portion of the lake have shown that the nitrate concentration is essentially zero and the only inorganic nitrogen form present is ammonia. Thus ammonia may select for the red alga over the green alga.

Ammonia by itself did not stimulate the green algae to a greater extent than nitrate, but rather maintained the population at the level which had been reached while the system was on nitrate. The presence of these algae allowed for the development of a larger number of bacteria than was found in the systems without algae.

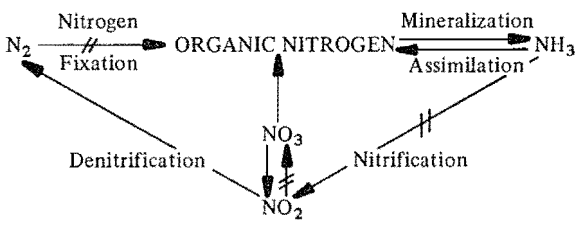
Urea stimulated algal metabolism, but algal numbers did not increase. The increased biological activity resulted in higher levels of dissolved oxygen and organic matter which in turn resulted in stimulation of the bacteria. The increase in bacterial numbers that occurred when algae were stimulated by urea was much larger than the increases in bacteria when the systems had nitrate or ammonia inputs. This indicates that the production of algal materials stimulatory to bacterial growth was greater on a urea source of nitrogen than it was on either nitrate or ammonia. An essential element to algal growth, possibly carbon or a trace element, must have become limiting in the system receiving urea inputs since there was little algal reproduction. The brine shrimp and brine fly larvae found in the northern portion of the Great Salt Lake primarily excrete ammonia or uric acid so urea is more than likely an insignificant source of nitrogen in this aquatic system.

Glutamic acid was the only nitrogen source tested which directly stimulated bacterial growth. This growth was stimulated to such a degree that the illuminated microcosm turned from green to pink indicating a replacement of the green algae by the red pigmented bacteria. This pink color resulting from a large concentration of pigmented halophilic bacteria is also characteristic of the waters of the northern arm of the Great Salt Lake. A high level of organic nitrogen (6-7 mg/l) in the form of amino acids could very well be responsible for this condition.

This study also demonstrated that nitrification probably does not take place in the simulated ecosystem. At least it could not be demonstrated by the methods used here. This means that mineralization of organic matter by bacteria is not followed by the production of nitrite or nitrate, but instead results in the uptake and/or accumulation of ammonia in the system. Since ammonia levels measured in the northern portion of the Great Salt Lake are quite low, the majority of the ammonia produced is evidently utilized by the biological components of the system.

Nitrogen fixation studies performed at Utah State University on samples of water, sediment, and colonized surfaces from the northern arm of the Great Salt Lake indicate that this process does not take place in that system. Therefore, nitrogen fixation cannot be considered as a source of nitrogen for this environment. Inputs must result primarily from precipitation, runoff, and the migration of brine from the southern lake basin into the northern basin.

The following diagram, based on the results of this and other studies, represents the apparent nitrogen cycling that occurs in the northern arm of the Great Salt Lake. Double lines across an arrow indicates the absence of a process from the system.



Nitrification and nitrogen fixation by the available evidence are presumed to be absent from the system. Uptake of nitrate or denitrification of nitrate are potentially possible since many of the bacteria possess these enzyme systems, but since nitrate and nitrite are not detected in the northern lake basin, these two parts of the cycle are probably insignificant or non-existent. The presence of halophilic organisms that have the potential for denitrification in an area where no nitrification is exhibited may seem rather unusual. In fact, there are no known halophilic nitrifiers. However, some halotolerant organisms in the southern arm of the Great Salt Lake possess the nitrifying ability and extreme halophiles have been found within this area. It is possible that extreme halophiles in this system developed the potential for denitrification while existing in close association with halotolerant nitrifiers. The presence of extreme halophiles with the denitrifying potential in an area where nitrification has not been demonstrated and where nitrate is not detected is probably due to the fact that these organisms are not confined to water of extreme salinity, but are also shown to be present in waters of much lower salinity (e.g. the southern end of the Great Salt Lake) where nitrification has been shown to occur (Porcella and Holman, 1972). In any case, the result of the absence of nitrification, nitrogen fixation, or the presence of nitrate for uptake or denitrification is that the production and the utilization of ammonia are the only two significant nitrogen cycling pathways operating in this system. When conditions become favorable for the algae, bacterial numbers increase in response to the increased levels of dissolved oxygen and organic matter. Subsequent mineralization of this organic matter produces ammonia which then stimulates the principal phytoplankter of the system, *Dunaliella salina.* As this cycle of uptake and mineralization continues, bacterial numbers increase to a point where they impart a wine-red color to the water. Thus, the equilibrium established between the conversion of ammonia into organic nitrogen and the production of ammonia from mineralized organic matter seems to be responsible for the conditions which are found in the lake at any one point in time. The limiting factor in this system would then seem to be any condition affecting either algal growth or mineralization of organic matter by bacteria. Some trace element may be limiting to this system, but more work will have to be done to determine this. In any case, organic nitrogen (6-7 mg/l) and phosphorus seem to be plentiful enough to rule either of them out as a limiting nutrient. A total phosphorus determination on a sediment sample from the northern arm of the Great Salt Lake indicated a level of phosphorus in excess of 400 μg P/g of sediment. This, together with an average 500 μg/l orthophosphate and 900 μg/l total phosphorus in the water column, suggests that phosphorus is not a limiting element in this system.

Since inorganic forms of nitrogen do not appreciably stimulate the bacteria of this system, they are probably not limiting to bacteria, but may be limiting to the algae. Since *Dunaliella viridis* is stimulated more by nitrate than by ammonia, the absence of nitrate in this system obviously limits this organism. This, however, is not the case in the southern arm of the lake where nitrate is present. Here it is observed that the dominant phytoplankter is the green alga, *Dunaliella viridis,* and not the red form, *D. salina.* The dominant form of inorganic nitrogen could thus be exerting a selective pressure on the algae of the northern and southern lake basins.

Total phosphorus was a quite sensitive indicator of biological activity in the microcosms. Trends in this parameter closely followed fluctuations in the biological components of the systems. Orthophosphate did not follow these trends and seemed to be rather unaffected by conditions that produced fluctuations in total phosphorus. The orthophosphate in the experimental systems seemed to be a reservoir of phosphorus that acted as a go-between in the exchange of phosphorus between the sediment and the biological components. This condition is also characteristic of the northern arm of the Great Salt Lake where orthophosphate remains quite constant during the year as total phosphorus levels respond to ongoing biological activity. The only time when orthophosphate levels paralleled total phosphorus levels was when glutamic acid was the sole nitrogen source. It was postulated that the exchange of phosphorus in this system was at such a high rate that the orthophosphate never had a chance to equilibrate with the sediment, but responded to the great increase in bacterial biomass that took place rather quickly. The steady increase in orthophosphate could also be due to the release of phosphorus from dead algal cells that had dominated the microcosm before the input of glutamic acid and release of phosphorus from the growing accumulation of bacterial biomass could also have added to this.

The microcosm approach to ecosystem analysis that was employed in this study proved to be an extremely useful and justifiable research tool. The laboratory simulation had defined boundaries, was manageable in size, variables were accounted for and controlled, and the system was replicable. Furthermore, due to the simplicity of the harsh environment that was simulated, the degree of similarity between the natural ecosystem and the microcosm simulation was quite high. Adding to this high degree of similarity was the fact that the sediment and the biological components within the microcosms were obtained from the ecosystem being simulated.

Replicability was exhibited by this microcosm approach in two ways. First, observations of the identical color changes that occurred when three

different pairs of microcosms were on a nitrate nitrogen source indicated that the same processes were occurring in all three pairs with virtually no visible differences. Secondly, analysis of chemical and biological parameters within these three pairs of microcosms indicated that the similarity observed in color changes also existed at the chemical and biological levels.

In addition to this, trends based on known biological principles that were expected to occur did in fact occur, adding to the validity of the approach. Increases in dissolved oxygen with increases in algal numbers or metabolism, absence of photosynthetic growth in dark microcosms, and decreases in the nitrate concentrations in systems switched from nitrate to another nitrogen source are a few examples of such expected trends.

This study also demonstrated the need for several changes in the system used. A reliable stirring or circulating mechanism should be included in the design of the microcosms to ensure the mixing of the aqueous phase. A possible solution might be in the use of a peristaltic pump circulating water from the bottom to the top of the system via an external hose. This might add a significant amount of new surface area to the system, but if an air-tight, closed system is to be maintained and reliable mixing is desired some attributes of the microcosm will have to be compromised to produce a system that meets the requirements of the intended research.

Also, a small volume acid trap (50-125 ml) should be placed in the air hose between the microcosm and the acid-leveling buret. Such an arrangement would prevent acid from being sucked into the microcosm in the event of a decrease in air pressure in the microcosm. Thought should also be given to the use of microcosms with different dimensions than those of the lucite cylinders that were used. The surface area:volume ratio in the lucite columns was quite high compared to that existing in a natural system. The final biomass determination indicated that much of the biological activity in the columns was accounted for by attached populations of algae and bacteria. A higher degree of similarity between the natural system and the microcosms could have been obtained with a lower surface area:volume ratio. This could be achieved in the future by either increasing the diameter of the cylinders or by using standard 10 gallon aquariums as microcosms. These systems would have smaller surface area:volume ratios than the lucite cylinders that were used, but at the same time would have larger volumes. Taking into account the large amount of biological activity that occurs on the inside surface areas of the microcosms, the researcher has to make a decision as to whether or not a low surface area: volume ratio is desired to meet the requirements of the research.

Toward a Lake Model

Work completed so far indicates a relatively simple ecosystem in the north arm composed of four basic groups, the algae, the bacteria, the brine shrimp and the brine fly. Organic matter and ammonia are the key nutrients. Figure 43 illustrates the major interactions. The invertebrates play a role through grazing and contribute to the organic and ammonia pools. The "IMPORT" includes organic and inorganic material from the south arm. The size of this import is not known at the present time but should be small compared to the nutrients and volume of the north arm itself. The ecosystem is further simplified by the apparent absence of nitrogen fixation, nitrification, and denitrification.

With the isolation of axenic cultures of the lake organisms, we are now preparing to determine some of the rates and details of the steps involved in the model. Studies on the fundamental biology of the lake organisms are also being pursued.

Conclusions

1. Nitrate, ammonia, and urea did not directly stimulate the halophilic bacteria in the microcosms.
2. Bacteria in systems receiving nitrate, ammonia, or urea were stimulated indirectly through the stimulation of an associated algal population.

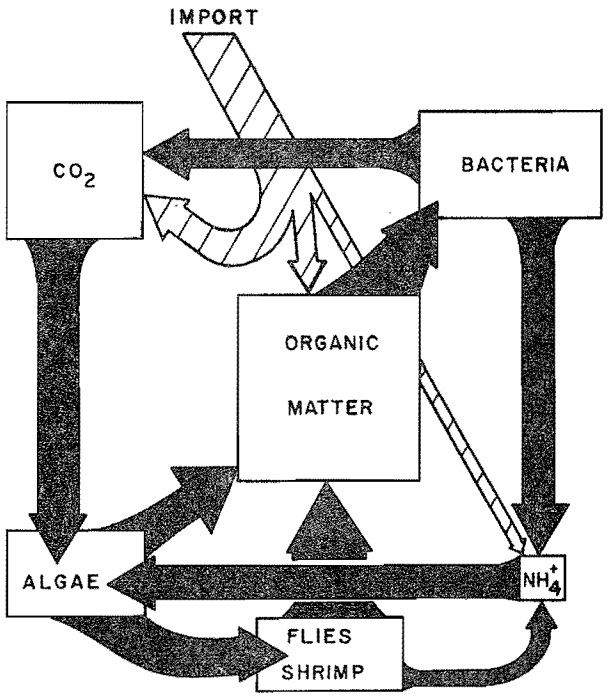


Figure 43. Model of the microbial community in the Great Salt Lake.

3. The green alga of the system, *Dunaliella viridis,* was stimulated most by nitrate while the red alga, *D. salina,* was stimulated most by ammonia. This may account for the observation that the red form is the dominant phytoplankter in the northern arm of the Great Salt Lake where ammonia is the only significant form of inorganic nitrogen.

4. Ammonia and urea were both capable of maintaining constant levels of algae in the microcosms.

5. Algal production of materials stimulatory to bacterial growth was greater on a urea source of nitrogen than it was on either a nitrate or ammonia source of nitrogen.

6. Glutamic acid was the only nitrogen source tested which directly stimulated bacterial growth. This growth was stimulated to such a degree that the illuminated microcosm turned from green to pink indicating a replacement of the green algae by the red pigmented bacteria. This condition is also characteristic of the northern arm of the Great Salt Lake.

7. Nitrification was not demonstrated in the microcosms. This process is probably quite insignificant, if not absent, in the simulated ecosystem and appeared to be absent from the lake also.

8. The production of ammonia from the mineralization of organic matter and the assimilation of ammonia by the algae seem to be the only two significant nitrogen cycling pathways operating in the simulated ecosystem and in the lake.

9. The limiting factor for the simulated ecosystem would seem to be any condition affecting either algal growth or the mineralization of organic matter by bacteria. Both organic nitrogen and phosphorus are plentiful enough in the northern arm of the Great Salt Lake to rule them out as limiting elements but the form may be limiting.

10. The organic pool in the north arm of the lake is quite large but does not seem to be readily available.

11. Nitrogen fixation does not appear to take place in the lake.

12. Nitrogen products excreted by the invertebrates during the summer months results in readily available nitrogen for the algae either directly in the form of ammonia or through decomposition to ammonia through the bacteria.

13. Cold weather, invertebrate grazing and availability of ammonia seem to be the limiting factors for algal and bacterial growth.

14. The bacteria appear to be stimulated by algal activity and invertebrate excretions.

15. A provisional model describing the various interactions is described.

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APPENDICES

Appendix A

Microcosm Chemical Parameters

Table 13. Microcosm No. 1, light.

Date	Day	NO2-N µg/l	NO3-N µg/l	NH3-N µg/l	o-PO4 µg/l	T-P µg/l	pH	D.O. mg/l	Alkal. mg/l
10-14-74	1	0		300		520	7.50	0.40	840
10-23-74	10	0		870		700	7.28	0.30	730
10-29-74	16	0		520	370	1700	6.89	0.35	895
11-04-74	22	0		0	430	640	6.78	0.40	860
11-13-74	31	0		0	370	630	6.75	0.55	910
11-19-74	37	80		0	470	650	6.70	0.58	950
11-25-74	43	80		80	420	560	6.50	0.90	1185
12-04-74	52	22		0	420	920	6.60	1.70	1020
12-10-74	58	31		0	400	540	6.60	2.35	1000
12-16-74	64	14	57	740	300	710	6.50	3.80	1000
12-21-74	69	0	0	340	400	1130	6.60	4.80	1010
12-27-74	75	0	20	850	130	340	6.55	4.10	730
01-03-75	82	0	0	0	0	320	6.60	2.10	1060
01-09-75	88	0	0	0	0	280	6.90	2.50	970
01-15-75	94	0	0	210	0	240	6.87	3.10	1170
01-21-75	100	0	0	0	50	220	6.90	2.85	1010
01-27-75	106	0	0	0	150	310	6.85	3.70	1170
02-05-75	115	0	0	0	75	330	6.85	4.20	1220
02-11-75	121	0	0	0	210	380	6.89	4.60	910
02-17-75	127	0	0	50	370	400	6.90	4.00	935
02-26-75	136	0	0	0	225	380	7.00	5.20	860
03-04-75	142	0	0	130	160	225	6.92	5.00	910
03-10-75	148	0	0	0	200	260	7.05	8.20	965
03-19-75	157	0	0	0	130	350	6.90	7.11	935
03-25-75	163	0	0	0	140	330	6.99	8.00	820
04-01-75	170	0	0	0	220	500	6.80	3.55	865
04-08-75	177	0	0	0	160	390	6.87	3.45	920
04-15-75	184	0	0	125	220	375	6.82	3.90	805
04-22-75	191	17	0	710	180	420	6.87	2.65	775
04-29-75	198	14	0	0	140	520	6.92	5.10	1010
05-06-75	205	14	0	0	200	490	6.95	4.40	990
05-13-75	212	0	0	110			7.05	5.40	900
05-20-75	219	0	0	0	230	575	6.93	7.05	960
05-27-75	226	5	0	970	230	790	6.93	6.05	1070
06-03-75	233	0	0	0	260	620	7.10	4.40	1180
06-11-75	241	0	0	0	220	475	6.83	4.82	1025
06-25-75	255	0	0	0	250	510	6.69	4.75	960
07-02-75	262	7	0	30	200	480	6.92	4.70	990

Table 14. Microcosm No. 3, dark.

Date	Day	NO2-N µg/l	NO3-N µg/l	NH3-N µg/l	o-PO4 µg/l	T-P µg/l	pH	D.O. mg/l	Alkal. mg/l
10-14-74	1	0		0		780	7.59	0.35	780
10-23-74	10	0		770		540	7.27	0.45	790
10-29-74	16	0		0	420	1560	7.00	0.50	870
11-04-74	22	0		0	560	670	6.80	0.45	960
11-13-74	31	0		0	540	710	6.73	0.50	900
11-19-74	37	60		0	550	680	6.69	0.50	950
11-25-74	43	30		200	570	650	6.52	0.35	1070
12-04-74	52	5		0	560	780	6.60	0.40	1030
12-10-74	58	8		0	620	680	6.55	0.45	1110
12-16-74	64	5	915	1680	590	670	6.60	0.45	1005
12-21-74	69	0	840	340	620	1970	6.60	0.65	1000
12-27-74	75	3	837	990	500	680	6.50	0.75	1010
01-03-75	82	0	870	130	330	680	6.60	0.90	980
01-09-75	88	0	840	0	690	570	6.70	0.60	1000
01-15-75	94	17	624	360	530	590	6.63	0.80	1100
01-21-75	100	25	636	0	730	580	6.68	0.35	1030
01-27-75	106	17	834	70	590	660	6.60	0.80	1190
02-05-75	115	3	887	0	500	580	6.57	0.80	1210
02-11-75	121	10	940	970	600	720	6.60	0.40	900
02-17-75	127	16	905	500	710	625	6.70	0.30	910
02-26-75	136	0	800	0	680	580	6.62	0.35	840
03-04-75	142	14	797	325	520	525	6.58	0.60	990
03-10-75	148	10	880	0	580	640	6.65	0.65	930
03-19-75	157	9	951	180	600	670	6.52	0.41	910
03-25-75	163	14	737	120	600	570	6.58	0.42	890
04-01-75	170	21	550	0	670	650	6.51	0.30	880
04-08-75	177	28	583	0	565	625	6.50	0.25	1040
04-15-75	184	35	697	235	620	565	6.45	0.40	815
04-22-75	191	46	656	390	640	600	6.50	0.40	765
04-29-75	198	70	433	160	575	620	6.57	0.40	1180
05-06-75	205	60	293	50	640	640	6.60	0.30	1090
05-13-75	212	16	205	590			6.62	0.30	1000
05-20-75	219	0	0	230	620	570	6.45	0.30	1030
05-27-75	226	0	140	725	610	570	6.57	0.20	1000
06-03-75	233	11	280	140	610	565	6.85	0.20	1280
06-11-75	241	0	430	0	480	525	6.55	0.40	1080
06-25-75	255	12	469	0	590	580	6.47	0.50	1090
07-02-75	262	28	323	120	550	560	6.71	0.40	990

Table 15. Microcosm No. 2, light.

Date	Day	NO2-N µg/l	NO3-N µg/l	NH3-N µg/l	o-PO4 µg/l	T-P µg/l	pH	D.O. mg/l	Alkal. mg/l
10-14-74	1	0		0		420	7.51	0.60	860
10-23-74	10	0		650		500	7.26	0.40	830
10-29-74	16	0		0	400	1560	6.95	0.35	825
11-04-74	22	0		0	370	630	6.78	0.50	835
11-13-74	31	0		0	370	610	6.70	0.45	890
11-19-74	37	70		0	470	630	6.68	0.62	910
11-25-74	43	90		200	470	520	6.60	0.50	1125
12-04-74	52	28		0	410	690	6.60	1.10	1050
12-10-74	58	30		0	420	500	6.60	2.35	1110
12-16-74	64	22	319	880	330	520	6.60	2.45	1020
12-21-74	69	0	20	210	340	1120	6.65	2.90	1000
12-27-74	75	0	0	380	120	470	6.60	2.85	990
01-03-75	82	0	0	0	0	340	6.65	2.30	1070
01-09-75	88	0	0	0	0	220	6.85	2.10	1010
01-15-75	94	0	0	270	0	230	6.83	2.00	1000
01-21-75	100	10	0	0	0	220	6.87	2.30	1100
01-27-75	106	0	0	60	130	300	6.78	2.80	1060
02-05-75	115	0	0	0	110	340	6.77	3.90	1090
02-11-75	121	0	0	0	225	420	6.79	2.70	920
02-17-75	127	0	0	0	330	370	6.79	2.85	810
02-26-75	136	0	0	0	210	425	6.83	3.35	840
03-04-75	142	0	0	120	180	280	6.77	2.90	1010
03-10-75	148	0	0	170	180	260	6.90	4.20	885
03-19-75	157	0	0	0	40	360	6.75	3.53	885
03-25-75	163	0	0	0	130	325	6.79	4.65	950
04-01-75	170	0	0	0	230	460	6.72	2.75	925
04-08-75	177	0	0	0	170	325	6.74	2.35	1000
04-15-75	184	0	0	25	140	330	6.70	2.10	810
04-22-75	191	6	0	285	180	600	6.75	1.95	805
04-29-75	198	0	0	0	150	415	6.80	2.50	1075
05-06-75	205	0	0	0	200	460	6.82	2.60	1030
05-13-75	212	0	0	120			6.88	2.80	1030
05-20-75	219	0	0	360		320	6.72	2.40	920
05-27-75	226	7	0	0	230	470	6.80	3.60	1100
06-03-75	233	0	0	0	175	520	7.05	4.10	1210
06-11-75	241	0	0	0	180	380	6.72	3.60	1030
06-25-75	255	0	0	0	260	520	6.60	2.85	1040
07-02-75	262	0	0	0	190	540	6.86	2.90	1010

Table 16. Microcosm No. 4, dark.

Date	Day	NO2-N µg/l	NO3-N µg/l	NH3-N µg/l	o-PO4 µg/l	T-P µg/l	pH	D.O. mg/l	Alkal. mg/l
10-14-74	1	0		0		490	7.59	0.45	750
10-23-74	10	0		550		540	7.28	0.45	755
10-29-74	16	0		0	420	1640	7.00	0.50	850
11-04-74	22	0		0	500	670	6.81	0.45	865
11-13-74	31	0		170	320	680	6.71	0.50	900
11-19-74	37	50		0	580	650	6.71	0.60	1040
11-25-74	43	70		670	530	600	6.64	0.30	1140
12-04-74	52	0		0	560	710	6.60	0.30	1020
12-10-74	58	15		30	600	540	6.55	0.40	1210
12-16-74	64	5	1015		550	670	6.60	0.55	1020
12-21-74	69	2	948	310	610	1920	6.65	0.65	1025
12-27-74	75	2	918	890	620	680	6.60	0.70	940
01-03-75	82	0	950	0	310	670	6.65	0.90	1060
01-09-75	88	0	920	0	670	720	6.75	0.70	1140
01-15-75	94	0	720	400	530	580	6.70	0.70	1040
01-21-75	100	21	770	790	670	620	6.70	0.30	1000
01-27-75	106	0	810	260	570	580	6.63	0.30	1160
02-05-75	115	3	477	660	500	580	6.59	0.40	1170
02-11-75	121	4	180	560	630	630	6.60	0.30	960
02-17-75	127	8	142	880	625	630	6.62	0.30	910
02-26-75	136	0	0	850	670	675	6.66	0.40	890
03-04-75	142	5	0	1040	620	540	6.60	0.45	1010
03-10-75	148	7	0	610	625	645	6.75	0.85	905
03-19-75	157	0	0	890	570	640	6.59	0.77	920
03-25-75	163	7	0	810	620	575	6.60	0.82	860
04-01-75	170	0	0	1430	620	590	6.58	0.40	875
04-08-75	177	0	0	910	575	615	6.54	0.30	1100
04-15-75	184	0	0	750	620	610	6.52	0.45	835
04-22-75	191	0	0	1300	590	590	6.55	0.40	770
04-29-75	198	0	0	650	570	570	6.64	0.50	1050
05-06-75	205	11	0	140	640	570	6.65	0.70	1050
05-13-75	212	0	0	630			6.68	0.40	1010
05-20-75	219	0	0	825	650	580	6.56	0.40	980
05-27-75	226	0	0	130	560	575	6.59	0.40	1070
06-03-75	233	0	0	110	575	590	6.90		1020
06-11-75	241	0	0	90	590	520	6.60	0.40	1110
06-25-75	255	0	0	180	520	610	4.90	0.40	200
07-02-75	262	0	0	110	540	625	6.48	0.50	820

Table 17. Microcosm "L," light.

Date	Day	NO2-N µg/l	NO3-N µg/l	NH3-N µg/l	o-PO4 µg/l	T-P µg/l	pH	D.O. mg/l	Alkal. mg/l
10-14-74	1	0		0		170	7.15	5.60	835
10-23-74	10	0		150		220	7.23	3.75	1050
10-29-74	16	0		0	50	560	7.00	3.80	860
11-04-74	22	0		0	0	170	6.92	4.70	800
11-13-74	31	0		0	0	190	7.00	3.90	850
11-19-74	37	0		500	0	280	6.95	2.75	850
11-25-74	43	0		310	0	270	6.95	3.10	1000
12-04-74	52	0		0	280	1130	6.95	3.30	1080
12-10-74	58	0		0	500	1070	6.80	4.70	1150
12-16-74	64	0	20	410	430	1020	6.40	4.60	995
12-21-74	69	0	0	540	460	1720	6.60	6.15	1010
12-27-74	75	0	0	1050	330	990	6.50	5.32	1040
01-03-75	82	0	0	160	90	570	6.25	3.90	1100
01-09-75	88	0	0	140	130	420	7.10	3.55	1070
01-15-75	94	0	0	560	80	410	7.02	4.00	1055
01-21-75	100	0	0	370	80	620	7.20	3.50	950
01-27-75	106	0	0	580	220	370	6.95	3.70	1010
02-05-75	115	0	0	560	170	440	7.00	3.90	1200
02-11-75	121	0	0	25	190	600	7.01	4.00	1020
02-17-75	127	0	0	125	550	690	6.88	3.80	900
02-26-75	136	0	0	0	440	560	6.90	4.25	935
03-04-75	142	0	0	490	280	490	6.88	4.10	1075
03-10-75	148	0	0	70	310	460	7.05	5.90	930
03-19-75	157	0	0	0	220	510	6.95	4.90	830
03-25-75	163	0	0	75	260	500	6.93	6.18	930
04-01-75	170	0	0	0	340	690	6.71	3.20	940
04-08-75	177	0	0	110	325	560	6.70	3.35	960
04-15-75	184	8	0	0	460	600	6.58	3.20	765
04-22-75	191	6	0	440	300	600	6.68	2.60	760
04-29-75	198	0	0	520	365	730	6.60	3.40	1140
05-06-75	205	0	0	490	360	570	6.74	3.18	1070
05-13-75	212	0	0	800			6.78	2.90	1110
05-20-75	219	0	0	590	310	600	6.60	2.95	960
05-27-75	226	0	0	170	360	575	6.72	2.90	1010
06-03-75	233	0	0	60	420	690	7.00	3.40	1030
06-11-75	241	0	0	0	500	670	6.60	3.90	1135
06-25-75	255	0	0	0	490	780	6.50	2.00	1050
07-02-75	262	12	0	190	420	650	6.68	1.85	980

Table 18. Microcosm "D," dark.

Date	Day	NO2-N µg/l	NO3-N µg/l	NH3-N µg/l	o-PO4 µg/l	T-P µg/l	pH	D.O. mg/l	Alkal. mg/l
10-14-74	1	0		100		370	6.63	0.35	615
10-23-74	10	0		450		360	6.77	0.40	650
10-29-74	16	17		180	550	1100	6.54	0.50	870
11-04-74	22	0		0	500	510	6.40	0.60	815
11-13-74	31	0		0	350	540	6.45	0.50	890
11-19-74	37	30		0	580	560	6.40	0.50	920
11-25-74	43	30		250	550	530	6.38	0.45	1120
12-04-74	52	0		1060	580	1070	6.45	0.40	1040
12-10-74	58	3		60	580	620	6.47	0.50	1390
12-16-74	64	5	775	450	620	880	6.40	0.55	1030
12-21-74	69	10		720	810	1400	6.45	0.55	1030
12-27-74	75	7	743	340	580	740	6.35	0.78	1000
01-03-75	82	0	880	180	270	640	6.45	0.50	1060
01-09-75	88	0	720	480	600	530	6.65	0.50	1060
01-15-75	94	14	487	430	570	570	6.60	0.60	1050
01-21-75	100	3	437	500	580	580	6.63	0.60	990
01-27-75	106	7	693	480	670	660	6.55	0.60	1150
02-05-75	115	0	700	525	475	590	6.51	0.40	1230
02-11-75	121	4	750	0	570	620	6.55	0.30	900
02-17-75	127	21	760	270	820	660	6.60	0.40	960
02-26-75	136	0	700	80	640	630	6.62	0.40	820
03-04-75	142	17	664	410	575	540	6.52	0.60	1065
03-10-75	148	15	706	110	625	625	6.60	0.60	980
03-19-75	157	21	760	200	630	680	6.50	0.68	910
03-25-75	163	20	731	230	660	600	6.55	0.84	950
04-01-75	170	2	628	0	640	625	6.50	0.40	965
04-08-75	177	3	667	0	590	580	6.49	0.35	1035
04-15-75	184	7	772	310	660	590	6.40	0.35	750
04-22-75	191	11	740	575	620	630	6.45	0.50	775
04-29-75	198	15	686	275	660	690	6.49	0.60	1050
05-06-75	205	7	273	280	660	650	6.55	0.30	1060
05-13-75	212	9	0	970			6.58	0.40	1080
05-20-75	219	0	0	450	660	650	6.40	0.85	1010
05-27-75	226	0	0	400	640	620	6.51	0.35	1070
06-03-75	233	0	0	150	600	660	6.87	0.30	1150
06-11-75	241	0	0	240	630	340	6.72	0.40	1110
06-25-75	255	0	0	1480	840	210	6.40	0.40	1090
07-02-75	262	0	0	450	580	640	6.61	0.45	1040

Appendix B

Microcosm Biological Parameters

Table 19. Microcosm No. 1, light.

Date	Day	Bact/ml x 10^6	Algae/ml x 10^3		
			Red	Green	Total
10-14-74	1	80	0.07	0	0.07
10-23-74	10	80	0.07	0	0.07
10-29-74	16	57	0.07	0	0.07
11-04-74	22	43	0	0	0
11-13-74	31	32	0.07	0	0.07
11-19-74	37	30	0	0.40	0.40
11-25-74	43	23	0.07	1.50	1.57
12-04-74	52	22	0	2.70	2.70
12-10-74	58	14	0	4.20	4.20
12-16-74	64	19	0	6.00	6.00
12-21-74	69	31	0.13	8.00	8.13
12-27-74	75	34	0	2.00	2.00
01-03-75	82	28	0.20	1.90	2.10
01-09-75	88	29	0.33	1.90	2.23
01-15-75	94	31	0.20	4.00	4.20
01-21-75	100	27	0.40	3.30	3.70
01-27-75	106	20	0.40	3.30	3.70
02-05-75	115	17	0.40	6.40	6.80
02-11-75	121	12	0.74	9.60	10.24
02-17-75	127	11	0.94	17.00	17.94
02-26-75	136	10	0.74	18.00	18.74
03-04-75	142	13	0.40	5.20	5.60
03-10-75	148	13	0.40	9.20	9.60
03-19-75	157	16	0.47	13.00	13.47
03-25-75	163	15	0.80	15.00	15.80
04-01-75	170	13	1.10	24.00	25.10
04-08-75	177	17	0.94	11.00	11.94
04-15-75	184	11	1.40	17.00	18.40
04-22-75	191	17	0.90	13.00	13.90
04-29-75	198	21	2.00	14.00	16.00
05-06-75	205	20	2.50	13.00	15.50
05-13-75	212	26	4.80	15.00	19.80
05-20-75	219	31	4.40	34.50	38.90
05-27-75	226	24	6.60	81.20	87.80
06-03-75	233	27	0.94	25.00	25.94
06-11-75	241	35	2.00	10.00	12.00
06-25-75	255		1.40	22.60	24.00
07-02-75	262	27	0.94	16.50	17.44

Table 20. Microcosm No. 3, dark.

Date	Day	Bact/ml x 10^6	Algae/ml x 10^3		
			Red	Green	Total
10-14-74	1	78	0	0	0
10-23-74	10	66	0	0	0
10-29-74	16	56	0	0	0
11-04-74	22	37	0	0	0
11-13-74	31	40	0	0.07	0.07
11-19-74	37	33	0	0	0
11-25-74	43	21	0	0	0
12-04-74	52	14	0	0.07	0.07
12-10-74	58	11	0	0	0
12-16-74	64	11	0	0	0
12-21-74	69	11	0	0	0
12-27-74	75	11	0	0	0
01-03-75	82	9	0	0	0
01-09-75	88	14	0	0	0
01-15-75	94	9	0	0	0
01-21-75	100	5	0	0	0
01-27-75	106	7	0	0	0
02-05-75	115	5	0	0.07	0.07
02-11-75	121	3	0	0	0
02-17-75	127	5	0	0.27	0.27
02-26-75	136	5	0	0.07	0.07
03-04-75	142	2	0	0	0
03-10-75	148	2	0	0	0
03-19-75	157	3	0	0.74	0.74
03-25-75	163	5	0.07	0.13	0.20
04-01-75	170	2	0	0.20	0.20
04-08-75	177	3	0	0	0
04-15-75	184	3	0	0	0
04-22-75	191	1	0.07	0.07	0.14
04-29-75	198	3	0	0.13	0.13
05-06-75	205	3	0	0.07	0.07
05-13-75	212	3	0	0	0
05-20-75	219	2	0	0.13	0.13
05-27-75	226	4	0	0.20	0.20
06-03-75	233	2	0	0	0
06-11-75	241	2	0	0	0
06-25-75	255	5	0	0	0
07-02-75	262	3.4	0	0	0

Table 21. Microcosm No. 2, light.

Date	Day	Bact/ml x 10^6	Algae/ml x 10^3		
			Red	Green	Total
10-14-74	1	64	0.07	0.07	0.14
10-23-74	10	82	0	0	0
10-29-74	16	32	0	0.13	0.13
11-04-74	22	49	0	0.13	0.13
11-13-74	31	28	0	0.27	0.27
11-19-74	37	29	0	0.47	0.47
11-25-74	43	31	0	1.40	1.40
12-04-74	52	19	0	1.70	1.70
12-10-74	58	16	0.13	3.30	3.43
12-16-74	64	18	0	5.00	5.00
12-21-74	69	24	0.20	9.00	9.20
12-27-74	75	32	0.07	4.00	4.07
01-03-75	82	26	0.07	3.00	3.07
01-09-75	88	33	0.13	1.90	2.03
01-15-75	94	26	0.13	3.50	3.63
01-21-75	100	22	0.40	3.50	3.90
01-27-75	106	16	0.20	4.50	4.70
02-05-75	115	11	0.54	7.40	7.94
02-11-75	121	11	1.00	11.00	12.00
02-17-75	127	10	1.00	9.00	10.00
02-26-75	136	12	1.20	12.00	13.20
03-04-75	142	7	0.90	3.70	4.60
03-10-75	148	13	1.30	4.50	5.80
03-19-75	157	13	0.87	4.00	4.87
03-25-75	163	7	1.9	6.00	7.90
04-01-75	170	6	2.60	9.20	11.80
04-08-75	177	13	0.87	6.00	6.87
04-15-75	184	14	1.20	8.00	9.20
04-22-75	191	17	1.50	5.00	6.50
04-29-75	198	21	1.90	4.00	5.90
05-06-75	205	27	1.50	5.00	6.50
05-13-75	212				
05-20-75	219	32	1.80	3.50	5.30
05-27-75	226	22	2.60	6.20	8.80
06-03-75	233	38	1.70	3.80	5.50
06-11-75	241	30	2.00	3.30	5.30
06-25-75	255	42	2.10	2.30	4.40
07-02-75	262	43	1.70	3.10	4.80

Table 22. Microcosm No. 4, dark.

Date	Day	Bact/ml x 10^6	Algae/ml x 10^3		
			Red	Green	Total
10-14-74	1	53	0.07	0	0.07
10-23-74	10	64	0	0.07	0.07
10-29-74	16	39	0	0	0
11-04-74	22	34	0	0	0
11-13-74	31	36	0	0	0
11-19-74	37	30	0	0	0
11-25-74	43	24	0	0.07	0.07
12-04-74	52	13	0	0	0
12-10-74	58	10	0	0	0
12-16-74	64	4	0	0	0
12-21-74	69	7	0	0.07	0.07
12-27-74	75	6	0	0	0
01-03-75	82	10	0	0	0
01-09-75	88	7	0	0	0
01-15-75	94	5	0	0	0
01-21-75	100	3	0	0	0
01-27-75	106	6	0	0	0
02-05-75	115	3	0	0	0
02-11-75	121	3	0	0	0
02-17-75	127	2	0	0	0
02-26-75	136	0.6	0	0	0
03-04-75	142	2	0	0	0
03-10-75	148	1	0	0	0
03-19-75	157	5	0	0	0
03-25-75	163	0.6	0	0.07	0.07
04-01-75	170	4	0	0	0
04-08-75	177	2	0	0	0
04-15-75	184	2	0	0	0
04-22-75	191	4	0	0	0
04-29-75	198	2	0	0	0
05-06-75	205	2	0	0	0
05-13-75	212	3	0	0	0
05-20-75	219	3	0	0	0
05-27-75	226	2	0.07	0	0.07
06-03-75	233	3	0	0	0
06-11-75	241	4	0	0	0
06-25-75	255	3	0	0	0
07-02-75	262	3.4	0	0	0

Table 23. Microcosm "L," light.

Date	Day	Bact/ml x 10^6	Algae/ml x 10^3		
			Red	Green	Total
10-14-74	1	52	0.13	10.00	10.13
10-23-74	10	30	0	5.15	5.15
10-29-74	15	23	0	8.50	8.50
11-04-74	22	19	0.07	4.00	4.07
11-13-74	31	11	0	7.00	7.00
11-19-74	37	18	0.13	9.60	9.73
11-25-74	43	21	0.13	8.20	8.33
12-04-74	52	19	0.07	9.90	9.97
12-10-74	58	15	0	12.00	12.00
12-16-74	64	13	0.07	14.00	14.07
12-21-74	69	16	0	19.00	19.00
12-27-74	75	15	0	19.00	19.00
01-03-75	82	18	0	5.00	5.00
01-09-75	88	33	0	4.95	4.95
01-15-75	94	24	0	6.00	6.00
01-21-75	100	6	0.07	9.30	9.37
01-27-75	106	18	0	8.20	8.20
02-05-75	115	14	0.07	14.00	14.07
02-11-75	121	18	0.13	25.00	25.07
02-17-75	127	28	0.07	31.00	31.07
02-26-75	136	30	0.07	31.00	31.07
03-04-75	142	20	0	7.00	7.00
03-10-75	148	15	0	6.50	6.50
03-19-75	157	18	0	16.00	16.00
03-25-75	163	19	0	22.00	22.00
04-01-75	170	19	0.67	16.00	16.67
04-08-75	177	21	0	13.00	13.00
04-15-75	184	18	0	18.00	18.00
04-22-75	191	22	0	9.00	9.00
04-29-75	198	28	0	8.50	8.50
05-06-75	205	32	0.07	8.00	8.07
05-13-75	212	27	0	5.50	5.50
05-20-75	219	30	0	5.00	5.00
05-27-75	226	40	0	4.10	4.10
06-03-75	233	38	0	5.80	5.80
06-11-75	241	38	0	5.50	5.50
06-25-75	255	40	0	0.90	0.90
07-02-75	262	46	0	3.30	3.30

Table 24. Microcosm "D," dark.

Date	Day	Bact/ml x 10^6	Algae/ml x 10^3		
			Red	Green	Total
10-14-74	1	11	0	0.13	0.13
10-23-74	10	3	0	0.20	0.20
10-29-74	16	9	0	0.40	0.40
11-04-74	22	5	0	0.20	0.20
11-13-74	31	2	0	0.60	0.60
11-19-74	37	3	0	0.47	0.47
11-25-74	43	1	0	0	0
12-04-74	52	5	0	0	0
12-10-74	58	1	0	0	0
12-16-74	64	6	0	0	0
12-21-74	69	3	0	0	0
12-27-74	75	3	0	0	0
01-03-75	82	2	0	0	0
01-09-75	88	3	0	0	0
01-15-75	94	5	0	0	0
01-21-75	100	3	0	0	0
01-27-75	106	2	0	0.27	0.27
02-05-75	115	2	0	0	0
02-11-75	121	2	0	0	0
02-17-75	127	1	0	0	0
02-26-75	136	4	0	0	0
03-04-75	142	2	0	0.13	0.13
03-10-75	148	5	0	0	0
03-19-75	157	5	0	0	0
03-25-75	163	6	0	0	0
04-01-75	170	3	0	0	0
04-08-75	177	2	0	0	0
04-15-75	184	2	0	0	0
04-22-75	191	2	0	0	0
04-29-75	198	2	0	0	0
05-06-75	205	3	0	0	0
05-13-75	212	6	0	0	0
05-20-75	219	4	0	0	0
05-27-75	226	3	0	0	0
06-03-75	233	5	0	0	0
06-11-75	241	5	0	0	0
06-25-75	255	5	0	0	0
07-02-75	262	8	0	0	0

Appendix C

Lake Parameters

Table 25. Lake parameters 1974-76.

Date yr/mo/dy	Water Temp. °C	D.O. mg/l^{-1}	Bacterial Count x 10^6 ml^{-1}		Red Algae ml^{-1}		Green Algae ml^{-1}		Specific Gravity	Alkal. Total mg/l^{-1}	Phosphate µg/l^{-1}		Nitrogen mg/l^{-1}			NH_3 µg/l^{-1}	pH
			Viable	Direct	Total	Motile	Total	Motile			Orth.	Total	Part.	Dissol.	Total		
740606	21.0								1.204								
740624	27.0			49	8600	-	2800	-	1.205								
740701	27.2	1.7	0.46	52	2700	2700	0	-	1.206								
740708	23.0	0.73	1.1	48	1400	1400	0	0	1.212								7.65
740715	26.5	0.42	-	41	170	170	0	0	1.205								7.70
740722	29.3	-	5.5	58	320	320	79	79	1.215								7.70
740729	29.8	0.26	4.0	63	320	320	79	79	1.212								7.55
740805	28.0	0.38	3.8	84	130	130	0	0	1.216								7.63
740812	24.2	0.47	0.57	72	480	480	0	0	1.215								7.73
740819	25.5	0.57	0.44	88	300	300	0	0	1.216								7.69
740826	26.2	0.71	2.5	59	540	540	0	0	1.214								7.58
740903	23.2	0.35	4.5	65	420	420	70	70	1.218								7.54
740909	25.0	0.31	1.5	81	140	140	0	0	1.220								7.55
740916	20.5	0.41	1.5	56	140	140	0	0	1.222								7.55
741001	19.8	0.78	4.0	54	43	43	0	0	1.219								7.60
741102	9.8	0.42	4.2	59	690	690	66	66	1.222	800	370	1800				0	7.75
741126	7.0	0.10	4.0	47	950	680	68	0	1.223	850	40	350				20	7.70
750107	0.5	0.70	0.086	56	200	68	140	68	1.218	690	330	970				0	7.70
750294	2.5	0.93	0.054	43	200	68	140	68	1.219	820	400	1080				0	7.58
750225	2.8	0.77	0.21	49	480	340	0	0	1.219	1010	570	990				0	7.65
750318	6.0	0.65	3.2	51	270	270	0	0	1.213	615	550	1020				0	7.65
750428	6.8	1.02	1.4	61	200	200	270	130	1.212	830	480	1025	1.5	5.4	6.9	0	7.49
750520	12.0	0.57	2.5	48	440	150	0	0	1.210	840	580	920	1.6	7.6	9.2	310	7.35
750527	15.8	0.72	2.0	51	680	480	0	0	1.209	1240	470	1020	1.3	12.0	13.3	180	7.65
750603	15.8	0.58	2.5	64	860	790	0	0	1.210	840	460	1060	1.9	4.5	6.4	0	7.47
750610	18.8	1.02	2.5	56	1400	1000	140	140	1.208	810	500	910	1.0	6.3	7.3	0	7.42
750617	18.8	0.82	4.4	67	1700	1300	70	70	1.212	800	570	860	1.2	9.1	10.3	0	7.40
750624	22.0	0.90	2.8	58	1600	1000	270	270	1.204	780	580	820	1.2	9.2	10.4	0	7.59
750701	25.5	0.90	2.7	66	920	750	0	0	1.200	790	520	975	1.0	5.4	6.4	0	7.70
750708	30.2	0.93	4.2	74	1100	750	0	0	1.207	830	900	1130	1.4	5.4	6.8	0	7.65
750715	26.3	0.91	3.4	79	770	600	170	110	1.205	1085	520	825				875	7.75
750722	26.8	1.5	2.8	82	1700	1200	270	130	1.208	931	425	920				1120	7.90
750729	24.5	0.95	3.8	74	220	220	72	72	1.205	890	1600	1920				375	7.98
750805	27.0	0.67	4.9	83	3100	3000	220	220	1.210	915	420	880				925	7.75
750819	24.8	0.53	0.87	66	3000	3000	210	140	1.211	940	330	1260				0	7.70
750826	23.5	0.57	0.21	61	360	360	0	0	1.210	900	330	825	1.1	4.5	5.6	830	7.53
750909	24.2	0.50	0.65	61	43	43	0	0	1.211	933	320	740	1.1	7.8	8.9	0	7.35
750916	23.5	0.43	1.1	86	710	710	140	140	1.212	690	290	630	1.6	4.2	5.8	0	7.40
750930	19.0	0.00	4.2	100	7500	6400	220	220	1.218	992	860	860	1.6	13.6	15.2	225	7.33
751021	13.8	0.58	2.1	69	320	320	0	0	1.210	690	470	840	1.2	5.8	7.0	860	7.50
751104	11.0	0.55	1.4	71	480	480	0	0	1.220		470	715	1.0	3.0	4.0	0	7.63
751125	5.6	0.62	2.8	81	1500	1400	0	0	1.222		360	1120	2.1	5.9	8.0	60	7.60
751216	0.8	0.90	0.21	69	290	220	0	0	1.205		330	770	0.9	3.6	4.5	0	7.71
760121	0.5	0.54	0.9	49	220	54	54	0	1.206		330	720	1.0	6.6	7.6	170	7.60
760309	8.0	1.8	1.4	65	100	100	0	0	1.200			770	1.4			0	7.71
760406	9.0	0.92	0.026	95	350	350	0	0	1.192		445	1030				0	7.62
760511	21.0	0.50	0.018	61	2000	1900	0	0	1.198		320	1030				0	7.69
760525	21.0	1.09		88	1600	1400	0	0	1.200		475	1180				285	7.72
760601	22.3	0.87		62	170	170	0	0	1.200		584	940				20	7.62
760608	24.0	0.55		73	920	780	220	220	1.200		680	1425				0	7.64
760615	17.0	0.75		45	980	980	80	80	1.204		385	795				0	7.55
760622	18.2	0.00		130	3400	2900	0	0	1.212								

Table 26. Depth profiles at eight Great Salt Lake stations, 11 August 1975 (750811).

Station Depth Meters	Water Temp. °C	D.O. mg/l^{-1}	Bacterial Count x 10^6 ml^{-1}		Red Algae ml^{-1}		Green Algae ml^{-1}		Specific Gravity	Alkal. Total mg/l^{-1}	Phosphate µg/l^{-1}		Nitrogen mg/l^{-1}			NH_3 µg/l^{-1}	pH
			Variable	Direct	Total	Motile	Total	Motile			Orth.	Total	Part.	Dissol.	Total		
LVG-1																	
Surface	26.1	0.60	1.5	52	270	200	0	0	1.202	680	430	900	1.0	6.0	7.0	175	7.44
1.5 (bottom)	27.2	0.80	1.3	65	420	420	0	0	1.208	700	400	970				25	7.46
LVG-2																	
Surface	25.6	0.80	3.3	57	400	270	0	0	1.198	690	430	980	1.1	5.3	6.4	25	7.27
1.5	26.1	0.45	3.2	57	540	340	0	0	1.212	710	340	910				0	7.50
3.0	25.0	0.70	3.1	74	1700	1300	130	130	1.214	720	390	950				0	7.52
4.5	24.4	0.60	1.5	79	4000	2700	140	140	1.214	710	320	980				0	7.35
6.0	21.7	0.00	1.5	71	2100	1100	66	66	1.218	690	310	950				220	7.38
8.2 (bottom)	17.8	0.00	1.2	147	140	140	140	140	1.224	805	460	2210				25	7.40
LVG-3																	
Surface	26.1	0.80	3.2	71	140	140	0	0	1.204	680	450	920	1.3	3.0	4.3	0	7.46
1.5	26.7	1.00	3.6	64	910	840	70	70	1.208	680	460	930				0	7.43
3.0	26.1	0.80	4.0	66	1300	1100	0	0	1.208	700	330	910				0	7.40
4.5	25.6	0.40	3.3	67	1500	1100	140	140	1.212	700	410	970				300	7.42
6.0	22.2	0.00	2.7	71	2900	2000	140	140	1.218	700	415	1130				0	7.42
7.5	18.9	0.00	2.7	69	400	68	0	0	1.220	710	440	1250				0	7.40
8.8 (bottom)	16.1	0.00	2.5	96	140	140	0	0	1.222	700	540	1170				75	7.40
LVG-4																	
Surface	26.1	0.75	0.98	72	900	600	140	140	1.212	710	375	950	1.4	5.4	6.8	0	7.47
1.5	25.6	0.70	1.2	70	800	500	0	0	1.210	690	410	970				0	7.45
3.0	25.6	0.62	1.9	68	1300	800	0	0	1.212	690	360	960				0	7.42
4.5	25.0	0.60	2.0	77	700	500	68	68	1.214	1000	420	880				0	7.43
6.0	18.9	0.35	-	74	1200	820	0	0	1.216	690	375	970				70	7.41
7.5	20.0	0.00	1.1	87	700	410	68	68	1.218	710	515	1140				275	7.40
9.5 (bottom)	16.1	0.00	1.0	166	200	140	0	0	1.222	750	575	3500				425	7.42
LVG-5																	
Surface	26.1	0.60	0.02	60	660	660	0	0	1.216	700	330	970	1.4	4.4	5.8	0	7.45
1.5 (bottom)	26.1	1.00	0.11	87	1100	1000	130	130	1.216	720	370	1180				0	7.47
NML																	
Surface	26.7	1.00	1.5	56	560	280	0	0	1.214	740	440	860	1.1	6.1	7.2	0	7.35
1.5	26.7	1.30	1.3	71	790	500	70	70	1.218	710	330	850				0	7.36
3.0	26.1	1.18	1.1	71	1100	900	0	0	1.218	680	375	880				200	7.35
3.8 (bottom)	26.1	1.00	1.7	114	1200	1100	0	0	1.220	635	390					1080	7.35
RD-2																	
Surface	26.1	0.90	3.5	50	560	420	0	0	1.210	685	400	950	1.1	6.1	7.2	300	7.20
1.5	26.7	0.60	4.1	61	660	370	0	0	1.210	680	380	1060				0	7.35
3.0	25.6	0.70	3.6	42	1300	930	0	0	1.210	700	380					20	7.31
4.5	25.0	0.20	4.8	60	1500	770	140	140	1.212	660	375	990				0	7.27
6.0	22.8	0.00	2.9	53	1700	1000	68	68	1.216	680	480	1000				0	7.35
7.0 (bottom)	20.6	0.00	3.3	144	600	390	98	98	1.220	970	630	4000				170	7.41
LVH																	
Surface	27.8	1.43	1.5	62	420	280	0	0	1.208	690	375	890	1.1	5.9	7.0	0	7.44
1.5	26.7	1.52	1.7	70	340	340	0	0	1.210	700	330	990				75	7.51
3.0 (bottom)	25.0	1.02	1.3	76	2700	2300	200	200	1.212	720	340	1100				500	7.42

Table 27. Depth profiles at three Great Salt Lake stations, 9 October 1975 (751009).

Station Depth Meters	Water Temp. °C	D.O. mg/l^{-1}	Bacterial Count x 10^6 ml^{-1}		Red Algae ml^{-1}		Green Algae ml^{-1}		Specific Gravity	Phosphate µg/l^{-1}		Nitrogen mg/l^{-1}			NH_3 µg/l^{-1}	pH
			Viable	Direct	Total	Motile	Total	Motile		Orth.	Total	Part.	Dissol.	Total		
LVG-1																
Surface	15.2	0.2	2.1	84	1300	880	150	150	1.216	430	860				610	7.78
1.5 (bottom)	14.5	0.20	1.4	94	1400	1100	0	0	1.216	710	890				310	7.80
LVG-2																
Surface	15.8	0.60	2.6	83	1700	1200	150	150	1.212	450	830				220	7.80
1.5	14.8	0.70	2.1	94	1500	810	0	0	1.214	480	830				230	7.80
3.0	14.8	0.44	3.2	89	1600	1100	0	0	1.214	430	840				130	7.85
4.5	16.9	0.34	2.5	69	1600	1100	200	200	1.214	400	960				180	7.80
6.0	15.8	0.34	2.3	110	1400	1000	140	140	1.216	430	860				210	7.83
7.9 (bottom)	16.3	0.00	2.7	85	2100	1200	280	280	1.218	425	930				180	7.78
LVG-3																
Surface	15.8	0.16	2.1	92	1700	1100	71	71	1.216	390	900	1.5	6.4	7.9	180	7.79
1.5	15.8	0.00	2.3	100	1700	1100	71	71	1.216	400	940	1.6	17.3	18.9	170	7.71
3.0	15.8	0.00	2.6	82	1000	510	150	150	1.214	385	1150	1.5	8.2	9.7	250	7.79
4.5	15.8	0.00	3.0	79	2500	1000	71	71	1.216	440	870	1.7	8.2	9.9	270	7.78
6.0	15.8	0.00	2.6	100	1400	590	73	73	1.216	430	900	1.5	3.6	5.1	80	7.70
7.5	15.8	0.00	1.8	95	1700	1000	73	73	1.216	450	970	1.6	6.0	7.6	175	7.83
8.8 (bottom)	15.8	0.00	3.0	160	2200	1200	0	0	1.218	525	1360	1.6	8.3	9.9	210	7.83

Table 28. Depth profile at Great Salt Lake station LVG-2 (approximately) 11 February 1976 (760211).

Station Depth Meters	Water Temp. °C	D.O. mg/l^{-1}	Bacterial Count x 10^6 ml^{-1}		Red Algae ml^{-1}		Green Algae ml^{-1}		Specific Gravity	Phosphate µg/l^{-1}		Nitrogen mg/l^{-1}			NH_3 µg/l^{-1}	pH
			Viable	Direct	Total	Motile	Total	Motile		Orth.	Total	Part.	Dissol.	Total		
LVG-2																
Surface	-5.0	2.60	Not done	14	68	68	1200	1200	1.100	140	300	0.6			90	8.18
1.5	1.0	0.70	0.011	61	204	0	0	0	1.210	390	520	1.3			0	7.77
3.0	0.8	0.80	1.5	59	610	540	0	0	1.212	300	600	1.4			170	7.75
4.5	1.1	0.63	0.023	60	770	0	0	0	1.214	200	580	1.4			0	7.78
6.0	1.5	0.70	1.5	58	630	0	0	0	1.216	290	825	1.2			90	7.78
7.5	2.2	0.40	0.13	86	630	140	0	0	1.216	260	940	1.6			0	7.76
9.6 (bottom)	2.8	0.00	0.36	240	3300	0	0	0	1.222	250	1810	4.0			0	7.72