KANSAS BIOLOGICAL SURVEY

ASSESSMENT OF THE QUALITY OF WATER SOURCES AT THE MILFORD FISH HATCHERY

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No. 33 February 16, 1987

Reports of the
State Biological Survey of Kansas

THE UNIVERSITY OF KANSAS 2041 CONSTANT AVE. LAWRENCE, KANSAS 66047-2906 (913) 864-7725

Final Report

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Submitted to

The Kansas Fish and Game Commission

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16 February 1987

PREFACE

This document represents the final report to the Kansas Fish and Game Commission of the project "Milford Hatchery Site Water Quality and Water Management Assessment". We thank Gary Welker and Franz Schmidt for their assistance in conducting the bioassays at the hatchery. We also thank the following personel at the Milford Hatchery for their cooperation and hospitality: Scott Steuwe, Tom Dorzab, Harold Jagerson, and Cecil "C.L." Hazlett. D. D. Dierking, superintendent at Public Utilities of Clay Center, KS, provided us access to one of the municipal wells.

ASSESSMENT OF WATER QUALITY AT THE MILFORD FISH HATCHERY

INTRODUCTION

The Milford Fish Hatchery is located 1 km downstream of the Milford Lake dam in Geary County, Kansas. The hatchery was constructed over a sand and gravel aquifer which receives water from Milford Lake and the Republican River. Water for the hatchery can be drawn from two sources: a nearby 44 hectare outlet lake or from three, 12.2 m deep wells located near the outlet lake. By using lakewater during the warm seasons and groundwater during cold seasons, water temperature at the hatchery is maximized for faster fish growth.

The outlet lake receives water from 74 pressure relief ("weep") wells located adjacent to the base of Milford Lake dam and from direct seepage from the aquifer. Because of the flat topography and sandy soil, surface runoff into the lake is probably negligible. The 12.2 m deep wells also draw water from this aquifer. Thus, all of the water used at the hatchery is ultimately derived from groundwater. Despite the use of what should be naturally purified groundwater, problems with fish survival appeared shortly after start of hatchery operations. The first year of production (1985) encountered disease, poor growth and excessive mortality. Fish were examined for disease organisms, however, the pathology report indicated that stress from poor water quality contributed more to morbidity than disease.

The first sign of distress exhibited by the juvenile catfish was development of a thick layer of mucus on the gills. Experimentation with different types of feed, chemical treatments, and stocking densities failed to improve their condition. These results and the results of water quality analyses performed on samples collected in September 1985 from various

locations around the hatchery, suggested that poor water quality might be the cause of the poor fish production.

Several parameters in those samples had notably high concentrations. They were total ammonia, iron, manganese, and pH. A characteristic odor eminating from the aeration towers at the hatchery suggested that hydrogen sulfide might also be present in the groundwater. Of these, the joint effect of ammonia with pH was felt to be the most likely cause of poor growth. Ammonia exists in two forms: ionized (NH_4^+) and un-ionized (NH_3). In a solution, these two forms are in an equilibrium which is largely pH dependent with the percent un-ionized ammonia increasing with increasing pH. It is un-ionized ammonia which is by far the most toxic to fish. Recommended maximum levels for un-ionized ammonia have been given as 0.025 mg/L by Alabaster and Lloyd (1982) and between 0.03 and 0.05 by EPA (1985) depending on temperature and pH. The maximum level recommended by EPA increases with both increasing temperature and pH. In raceway 12 during September 1985, the total ammonia was 0.85 ug/L and the pH was 8.1. We calculated the un-ionized ammonia concentration to be between 0.041 and 0.057 mg/L which is within the range of the maximum levels recommended by EPA.

The iron concentrations found in one of the supply wells and in the outlet lake near the intake for the hatchery were 2.89 and 0.19 mg/L, respectively. The former value exceeds the criterion of 1.0 mg/L of iron set by EPA (1976) for the protection of aquatic life. Iron can exist in waters in a reduced, soluble form when anoxic or low pH conditions exist as in groundwater, or in an oxidized, insoluble form when oxygen is present. Although considerable iron may be in the groundwater which flows into the outlet lake, most of the iron is oxidized and precipitated out before it

reaches the hatchery intake. Iron precipitate, in the form of $Fe(OH)$, is probably responsible for much of the yellow-orange and red-brown flocculent material in the weep well collecting canal. Iron in the groundwater used by the hatchery may not be oxidized until the water is passed through the areation towers. The resulting precipitate will then enter the raceways where it could be a possible source of irritation to the gills of the catfish.

The concentrations of manganese were found to be 1.02 mg/L in one of the supply wells, 0.76 mg/L near the intake structure in the outlet lake, and 0.42 mg/L in the raceways. The values are comparable to those found to adversely affect fish eggs and fry. Lewis (1976) found that 0.325 mg/L of manganese increased mortality of rainbow trout eggs two-fold over control conditions. Nix and Ingols (1981) found that mortality of rainbow trout fingerlings increased rapidly as concentrations of manganese increased above 0.5 mg/L. They concluded that it was manganese in the oxidized state that was most toxic. Soluble, reduced manganese compounds can enter a fish hatchery water supply via withdrawal from an anaerobic hypolimnion or groundwater. If these water sources are sufficiently aerated, the manganese will eventually be oxidized back to the more toxic form. Fortunately, this reaction occurs at a very slow rate when pH is less than 9.1 (Morgan 1967). Since residence time in the raceways under normal flow rates (250 gal/min/raceway) is less than 40 min, it is unlikely that significant amounts of oxidized manganese will be present in the system when groundwater is being used. The residence time of water in the outlet lake is probably sufficient for significant amounts of oxidized manganese to form and precipitate out. The slow oxidation of manganese could also allow significant amounts of soluble manganese in groundwater to pass through the filtration system inside the hatchery. Oxidation could then

take place in the egg hatching and fingerling rearing section of the hatchery, thus endangering those operations.

Another toxic compound which forms under the anoxic conditions present in groundwater or lake sediments is hydrogen sulfide. This gas is characterized by an unpleasant odor and extreme solubility in water. Intuitively, this compound should not be a problem at the hatchery because the sulfide should be oxidized to sulfate when the source water is passed through the aeration towers. However, hydrogen sulfide is very toxic and concentrations above 0.002 mg/L are considered a long-term hazard to fish and other aquatic life (EPA 1976). Therefore, if the oxidation of the hydrogen sulfide is anything less than total, significant concentrations of hydrogen sulfide may exist in the hatchery water.

Another water quality concern at the hatchery was the eutrophic condition of the outlet lake. Eutrophication can occur when high levels of nutrients such as nitrogen and phosphorus are present in a lake. The resulting high levels of phytoplankton primary production and standing crop can raise the pH of the water and produce large amounts of decaying organic matter. This could be detrimental to the fish at the hatchery if the decay of the organic matter results in an accumulation of ammonia in the water. Under these conditions significant portions of the ammonia would be in the toxic, un-ionized form because of the elevated pH. Even if no ammonia exists in the outlet lakewater, the high pH will cause or significant portion of the ammonia excreted by the fish in the raceways to be in the un-ionized form.

Water samples collected from the weep wells in September 1985 had nutrient concentrations of 0.37 to 1.2 mg/L total phosphorus and 0.68 to 1.0 mg/L ammonia. Nitrate concentrations ranged from not detectable to

only 0.06 mg/L. The ammonia and phosphorus concentrations were high enough to suggest that the groundwater, which is the source of water for the outlet lake, was responsible for the lake's eutrophic condition. The high levels of ammonia and low levels of nitrate in the weep well water resembled the pattern of nutrient concentrations derived from typical lake sediment elutriates. Since the aquifer below Milford Lake is recharged by water passing out of the lake through the sediments, it is possible that the nutients in the groundwater were leached out of the lake sediments.

The objectives of this research were to:

--Determine more precisely the water quality parameter(s) of the source water which might be inhibiting fish growth;

--Determine if seasonal changes in water quality exist;

--Identify the source of nutrients causing eutrophication in the outlet lake; and

--Recommend measures to improve water quality.

METHODS

Bioassays. To determine which water quality parameter might be causing poor fish growth we used a seven-day, static-renewal, larval fish bioassay (Norberg and Mount 1985). Larval fish were used because their relative growth rates are so rapid that it is possible to detect differences in growth among treatments using only a 7-day test. Our standard test organisms were larval fathead minnows. These fish were used because: an extensive amount of toxicological work has already been done on this organism; they are easily obtainable at any time and of known age; they are disease free; they have a known genetic background; and they are highly sensitive to many toxicants. They were obtained from EPA's Newton Toxicology Laboratory near Cincinnati, Ohio. Walleye and catfish larvae were also used when they were available. The walleye were obtained from

the Milford hatchery and the channel catfish came from the Pratt hatchery. A total of seven bioassays were performed during the period from March through December 1986 (Table 1) in an attempt to detect seasonal patterns in response of the fish to the different sources of water.

Five sources of water from the vicinity of Milford hatchery were used in the bioassays. They were Milford Lake, the outlet lake, weep wells, and the hatchery domestic and supply wells. A sixth source was water from the reservoir at the Nelson Environmental Studies Area (NESA) near Lawrence, Ks. Water from the NESA reservoir has been used by us in previous bioassays and is considered to be free of any toxic contaminants. Past experiments using unadulterated NESA water have generally had mortalities less than 10 percent. The purpose of using a variety of water sources was to provide a variety of chemical compositions. The goal was to isolate a chemical agent causing poor growth by correlating growth with one or more chemical parameters.

Water from Milford Lake was collected either from 20 cm below the surface just offshore from the boat ramp nearest the dam on the south shore, or from the main basin approximately 300 m offshore from the middle of the dam at a depth of 5 m. Samples were collected from the main basin when weather conditions permitted the use of a boat. Water from the outlet lake was collected either from 1 m deep in the vicinity of the hatchery intake structure or 20 cm below the surface just off a stone pier at the channel connecting the upper and lower basins, or as outlet lake water entered a raceway. Water from the domestic well was collected from a pressure tank inside the hatchery before the water was treated. Water from the hatchery supply wells came from the head of either well #3 (western-most well, 500 gpm) or well #1 (eastern-most well, 1000 gpm) or was a composite sample taken as the water entered a raceway. Water from the weep

Table 1. Dates and species used in the seven 7-day larval fish bioassays performed at the Milford Fish Hatchery during 1986.

Bioassay	Date	Species
1	12–18 March	Fathead minnow
2	14–22 April	Walleye
3	8–16 May	Fathead minnow
4	24 June–1 July	Channel catfish
5	26 August–2 September	Fathead minnow
6	18–25 November	Fathead minnow
7	4–11 December	Fathead minnow

wells was collected at the head end of lateral canal C (second northern-most canal) and represents a composite of weep wells from near the middle of the weep well array. Water from NESA was brought in at the beginning of each bioassay and stored under refrigeration at 4 degrees C. Water from around the hatchery was collected daily. Before the water was used, its temperature was adjusted to 25 degrees C in a water bath, then the samples were aerated for 20 min in order to adjust the level of dissolved oxygen (DO) to near saturation.

For each source of water, four replicate chambers were set up. Each chamber consisted of a 1 L, tall-form, spoutless beaker and was initially stocked with 10 fish between 0 and 24 h old. Each beaker was filled to its maximum capacity then covered with a watch glass to prevent the potential loss of any volatile compounds.

The temperature in the beakers was maintained at 25 degrees C. Beakers were placed in a water bath equipped with a thermostat, four aquarium heaters, a refrigeration probe, and a magnetically driven stirring system which provided water circulation for even temperature distribution. Lighting was provided by fluorescent room lights and from two 40 watt fluorescent lights suspended 60 cm above the water bath. Minimum light intensity was 140 lux when only the overhead lights were used. The light cycle was 16 h light and 8 h dark. Larvae were fed live brine shrimp nauplii at 0800, 1400 and 2000 h each day. Water in the beakers was renewed daily. For each replicate, all but 200 ml of old water was siphoned off, the number of alive and dead larvae were recorded, then the remaining water and live fish were transferred to a clean beaker. The beaker was then filled with the freshly collected water and a clean cover glass was applied.

Residual fresh water from each treatment was saved for measurement of conductivity, pH, total hardness, total alkalinity, and total ammonia. In addition, the DO in each chamber was measured before and after changing the water. Once during each bioassay, a separate set of samples was collected and analysed by an outside laboratory (Wilson Laboratories, Salina, KS or the US Army Corps of Engineers, Missouri River Division Laboratory, Omaha, NB) for total and soluble iron, manganese, and barium. Barium was included in the routine analyses because it is a heavy metal, was frequently detected, and its' concentration correlated with fish growth in the first bioasssay. Sulfide analyses were also performed by us, Wilson Laboratories or the Corps of Engineers. Specific methods for analyses we performed are given in Appendix 1.

At the end of the bioassay, the remaining living fish were preserved in 4% formalin. The fish from each replicate were collectively weighed on an analytical balance and the average weight per fish was calculated by dividing total weight of fish by the number of survivors in the replicate. Assuming that the initial weights of the fish are the same for all treatments, then the final fish weight is proportional to the actual growth rate.

The above description of our protocol represents a typical bioassay performed at the hatchery. Minor variations existed among bioassays in the timing of sample collection, timing of feeding, which samples were aerated, and which chemical analyses were performed. Some of these variations will be discussed later when they produced significant effects.

Outlet lake. Several steps were taken to analyze the eutrophic condition of the outlet lake. A hydraulic budget for the lake was calculated by measuring inflow from the lateral canals, outflow from the outlet lake, and

ascertaining the withdrawal rate by the hatchery. The rate of inflow by direct groundwater seepage was calculated by subtracting the weep well inflows from the total outflow. We assumed that evaporation and loss to groundwater were negligible. In addition, we computed residence time of the water in the outlet lake. This was calculated by dividing the total lake volume by the daily inflow. Lake volume was determined from a bathymetric map using a polar planimeter to create and analyze the hypsographic curve (Lind 1979). The bathymetric map was from the Project Site Plan prepared by Kramer, Chin, and Mayo Inc., Seattle, WA. Nutrient loadings were calculated from measurements of nutrient concentrations in the lateral canals. To determine what nutrient was limiting growth of the phytoplankton in the outlet lake, we measure total inorganic nitrogen (N) and total phosphorus (P) concentration then calculated N to P ratios. To assess the impact of Milford Lake on the groundwater quality, we made additional nutrient concentration measurements on water from the hatchery supply wells and municipal wells in Clay Center and Junction City. We also measured nutrient releases from Milford Lake sediments collected from two locations in the main basin about 300 m from the dam face (see Appendix 1 for sediment elutriations method). To determine if groundwater below other reservoirs was similarly impacted, we visited 13 other reservoirs in eastern Kansas to collect and analyze water samples from their weep wells.

RESULTS

Bioassays. Significant deviations from the standard methodology occurred in bioassays 1 and 4. In the first bioassay, initially, only the domestic well water was aerated. Through the first four days of the bioassay mortality was slight and it appeared that this technique would be acceptable, however, problems began to develop on Day 5. In the control

and Milford Lake replicates, some of the fish developed gas bubbles inside their abdominal cavities. These fish could not regulate their bouyancy and did not eat. Many of the afflicted fish died. No fish with gas bubbles remained alive at the end of the bioassay in the Milford Lake treatment. This problem may have been the result of gas supersaturation in the water. Some measurements made just after adding fresh water to the beakers indicated DO in excess of 9 mg/L. The saturation concentration for oxygen in water at 25 degrees C at the altitude of the hatchery is 7.79 mg/L. This condition was probably caused by the rapid warming of the samples from room temperature to 25 degrees C. On Day 7, the DO in the domestic well replicates dropped to extremely low levels despite the aeration of the sample before use. Two of the replicates suffered 100% mortality and one sustained 60%. Mortality in one replicate was not affected but it is likely that growth was inhibited. These problems compelled us to disregard the survivorship and final weight results from the NESA and domestic well treatments and the survivorship data from the Milford Lake treatment. The fourth bioassay used channel catfish larvae which were approximately ten times larger (dry weight) than the fathead minnows. This larger size and corresponding metabolic requirements were sufficient to cause significant depletion of DO in the beakers after 12 h. Thus, each beaker received an additional 10 min of bubbling 12 h after being placed in fresh water.

Mortality of fish in all seven bioassays was never found to be associated with water source (Table 2). The overall mean and 95% confidence limits for 38 treatments was 85 $\pm$ 5.5% survivorship at the end of seven days. No significant ($p < 0.05$) differences were found among water source treatments in any of the seven bioassays when tested separately (one-way ANOVA used for each). Thus, any water quality

Table 2. Means ($\bar{x}$) and standard deviations (s.d.) of percent of survivors for each treatment at the end of each bioassay (Day 7). Means were of four replicate beakers. Percent survivorship was tested for source water treatment effects using an ANOVA for each bioassay. No differences among treatments were found at $p < 0.05$ for each bioassay.

Bioassay		Water Sources for Treatments:							
		NESA	Domestic well	Supply well 1	Supply well 3	Supply (mixed)	Weep well	Outlet lake	Milford Lake
1	$\bar{x}$	*	*	-	-	100	100	97.5	*
	sd	*	*	-	-	0.0	0.0	5.0	*
2	$\bar{x}$	57.5	50.0	-	-	63.3	46.7	50.0	45.0
	sd	12.6	8.16	-	-	5.77	11.5	8.16	7.07
3	$\bar{x}$	92.5	97.5	-	80.0	-	100	87.5	95.0
	sd	9.57	5.00	-	14.1	-	0.0	15.0	10.0
4	$\bar{x}$	97.5	80.0	-	67.5	-	97.5	97.5	97.5
	sd	50.0	40.0	-	32.0	-	5.0	5.0	5.0
5	$\bar{x}$	100	-	-	95.0	-	100	90.0	92.5
	sd	0.0	-	-	5.77	-	0.0	14.1	5.0
6	$\bar{x}$	95.0	-	87.5	90.0	-	85.0	80.0	72.5
	sd	3.33	-	15.8	6.67	-	16.7	20.0	22.5
7	$\bar{x}$	80.0	-	95.0	97.5	-	100	75.0	92.5
	sd	24.5	-	10.0	5.0	-	0.0	23.8	9.6

* source water tested but mortalities were due to known methodological errors and were therefore omitted

- source water not tested

differences among sources did not affect survivorship in these 7-day bioassays.

Fish growth measured as average weight per fish at the end of 7 days was not consistantly affected by any of the water sources (Table 3). In four of the seven bioassays no significant ($p < 0.05$) differences in growth were found among water source treatments using ANOVA tests for each bioassay. In the other three bioassays (1, 2, and 6) significant differences in growth existed. However, no water sources which were alike or different in fish growth in one bioassay (T-method pairwise comparison tests, Sokal and Rohlf, 1981) behaved similarly in the other bioassays. A correlation between barium and fish growth was observed in bioassay 1, however, it was not found in the other bioassays. When average weight per fish in each treatment was compared to weights per fish in the water from NESA, fish weighed more in some source waters and less in other source waters. Using all bioassays, the differences among water sources were not significant ($p < 0.05$) in an ANOVA test (Table 4). Therefore, fish growth was not found to be affected by the different water sources.

Characterization of the waters used in the bioassays for ionic strength was done by measuring alkalinity, hardness, conductivity, and pH (Figure 1). None of these water quality parameters varied much from the earliest bioassay done in March 1986 to the last in December 1986 (Appendix 3). Thus, means were calculated and used for comparisons among source waters. In general, the pH of lake water samples were higher than those of groundwater samples. An apparent exception was the pH of supply well 1, however, this mean is composed of only two dates from late 1986. Alkalinity, hardness and conductivity were higher and most similar for the groundwater sources, lowest for NESA reservoir water and intermediate for Milford area surface waters. The NESA reservoir water was the lowest in

Table 3. Means ($\bar{x}$) and standard deviations (sd) of average weight (mg) per fish for each treatment at the end of each bioassay. Bioassay 2 was with walleye, 4 with channel catfish, 1, 3, 5, 6 and 7 with fathead minnow. Means were of four replicate beakers. The average weight per fish was calculated by dividing the total dry weight of survivors by number of survivors in each beaker. Means with a common underline were not significantly ($p < 0.05$) different (T-method pairwise comparison tests) after significant ($p < 0.05$) differences were found in an ANOVA. (ns = no significant differences, found using an ANOVA, thus, no comparison tests were done.)

Bioassay		Water Sources for Treatments:					
1		Supply W.	Outlet L.	Weep W.	Milford L.		
	$\bar{x}$	0.432	0.528	0.554	0.565		
	sd	0.030	0.042	0.058	0.062		
2		Milford L.	Supply W.	Weep W.	NESA	Domestic W.	Outlet L.
	$\bar{x}$	0.629	0.720	0.725	0.786	0.942	1.015
	sd	0.138	0.132	0.191	0.091	0.106	0.015
3	ns	Domestic W.	NESA	Weep W.	Milford L.	Supply 3	Outlet L.
	$\bar{x}$	0.630	0.653	0.697	0.699	0.722	0.749
	sd	0.030	0.032	0.041	0.102	0.065	0.032
4	ns	Supply 3	Milford L.	Weep W.	Outlet L.	Domestic W.	NESA
	$\bar{x}$	6.68	7.88	8.09	8.25	9.30	11.0
	sd	1.18	1.41	1.68	1.45	3.95	5.43
5	ns	Weep W.	NESA	Supply 3	Milford L.	Outlet L.	
	$\bar{x}$	0.406	0.413	0.442	0.482	0.521	
	sd	0.065	0.033	0.035	0.058	0.078	
6		Supply 3	NESA	Weep W.	Supply 1	Outlet L.	Milford L.
	$\bar{x}$	0.294	0.320	0.336	0.381	0.400	0.467
	sd	0.046	0.015	0.047	0.094	0.046	0.077
7	ns	NESA	Supply 3	Weep W	Outlet L.	Supply 1	Milford L.
	$\bar{x}$	0.397	0.402	0.406	0.411	0.422	0.450
	sd	0.059	0.023	0.020	0.060	0.045	0.028

Table 4. Grand means ($\bar{x}$) and standard deviations (sd) of fish growth from two to six bioassays with replicates for total sample sizes (n) Weight per fish at the end of each bioassay was relativized by dividing each treatment's weight of fish by the mean weight of NESA fish. An ANOVA yielded a nonsignificant F-statistic. Results from the first bioassay were not included because supersaturation caused gas bubbles to form in the NESA fish.

	Supply well 3	Supply (mixed)	Weep well	NESA	Milford Lake	Outlet lake	Supply well 1
$\bar{x}$	0.860	0.916	0.939	1.00	1.02	1.08	1.13
sd	0.247	0.167	0.211	0.162	0.313	0.277	0.219
n	16	4	24	24	24	24	8

MS among = 0.109
MS within = 0.0612

F = 1.78 d.f. = 7/128, $p > 0.05$ = not significant

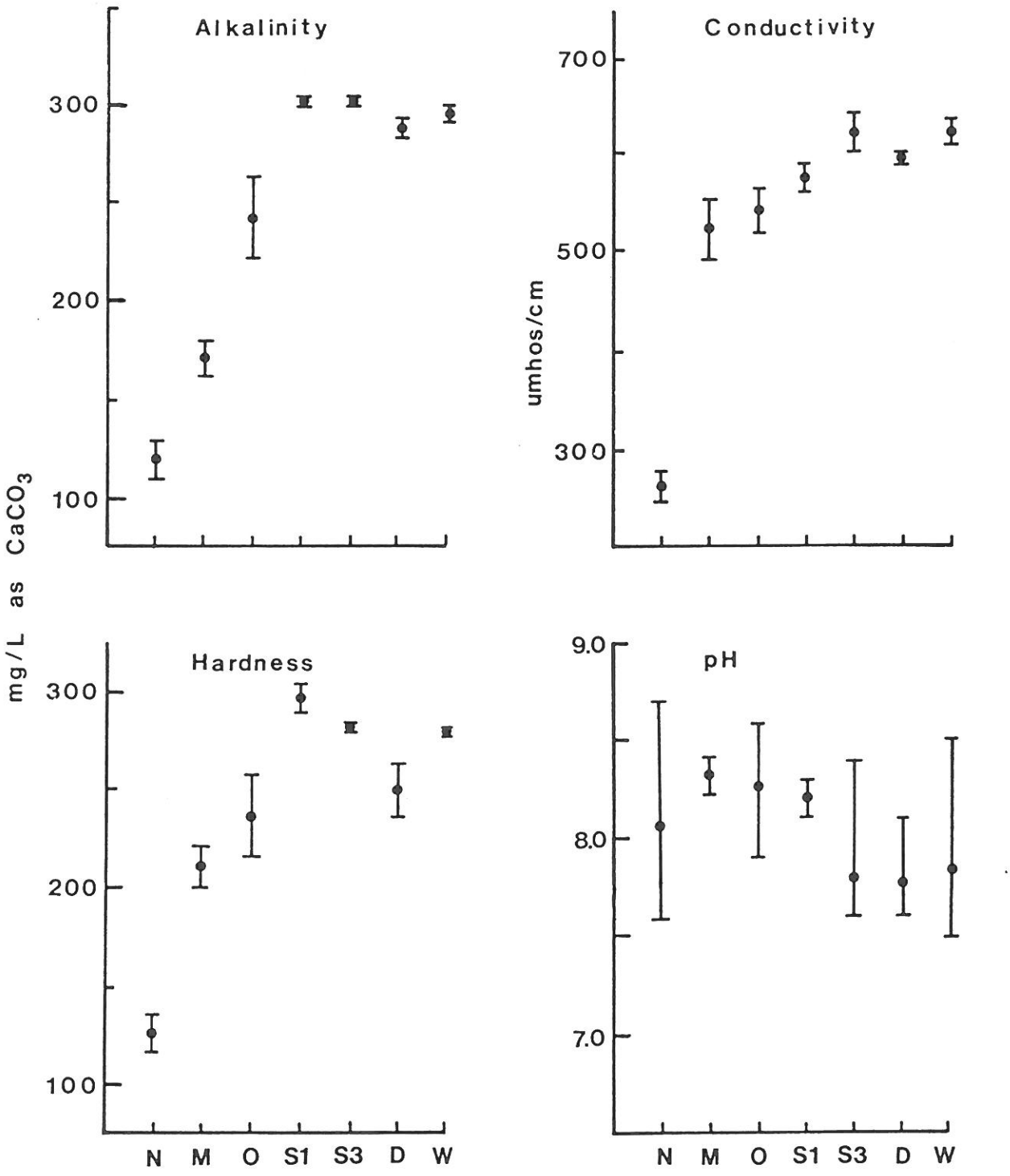

Figure 1. Means ± one standard error of total alkalinity (mg/L as $CaCO_3$), conductivity (μmhos/cm), and hardness (mg/L as $CaCO_3$); and means and ranges of pH for water tested in two to seven bioassays. Test waters used were N = NESA, M = Milford Lake, O = outlet lake, S1 = supply well 1, S3 = supply well 3, D = domestic well, and W = weep well. For each bioassay, values for each water source were means of three to eight measurements taken during the 7-day bioassays.

buffering capacity as indicated by means for alkalinity and hardness of 119 and 125 mg/L as $CaCO_3$, respectively, and for conductivity of 262 µmhos/cm. The groundwater (or well water) sources were always similar to each other with overall means being 295 mg/L as $CaCO_3$ for alkalinity, 280 mg/L as $CaCO_3$ for hardness, and 600 µmhos/cm for conductivity. The outlet lake and Milford Lake had means of 240 and 170 m/L $CaCO_3$ for alkalinity, 235 and 210 mg/L as $CaCO_3$ for hardness and 545 and 525 µmhos/cm for conductivity, respectively. The greatest variations in alkalinity and hardness were observed in the outlet lake water and were due to seasonal changes. In fall and water, values for both parameters were greater than 280 mg/L but less than 180 mg/L in later spring and early summer.

Measurements were made to detect potentially deleterious chemicals in the source waters used in the bioassays. Water samples taken on 12 March from NESA reservoir, the outlet lake, weep wells, a composite of supply wells and domestic well water were all found to have no detectable concentrations of herbicides, insecticides, PCB's, or soluble metals (Appendix 4). However, Milford Lake surface water had 2.9 ug/L atrazine on this date.

Chemical element analyses which were done once during each bioassay revealed iron, manganese, and barium in higher concentrations in the groundwater than in surface water supplies (Figures 2, 3 and 4). Overall means for supply wells 1 and 3, domestic well, and weep well versus NESA and Milford Lake, respectively, were 2.5 vs. 0.33 mg/L for total iron, 0.78 vs. 0.03 mg/L for total manganese and 0.35 vs. 0.08 mg/L for total barium. The outlet lake water mean concentrations for manganese, 0.06 mg/L and barium, 0.24 mg/L, were in between means for groundwater and surface water sources. Outlet lake mean concentration for iron, 0.18 mg/L, was more like the surface water sources. Soluble forms of these elements were usually

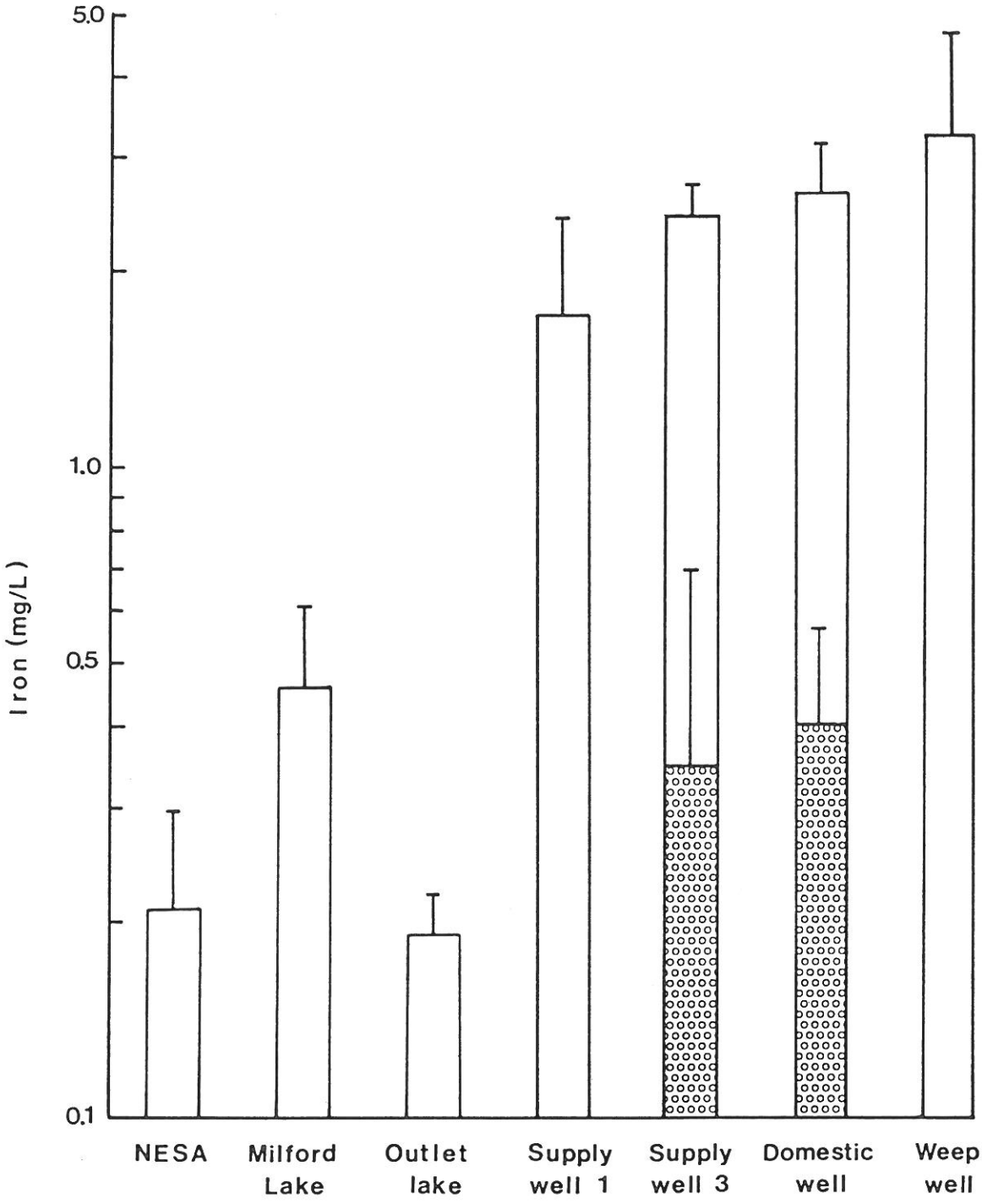

Figure 2. Means + one standard error of total (open bar) and soluble (shaded bar) iron (mg/L) for seven water sources tested in two to seven bioassays.

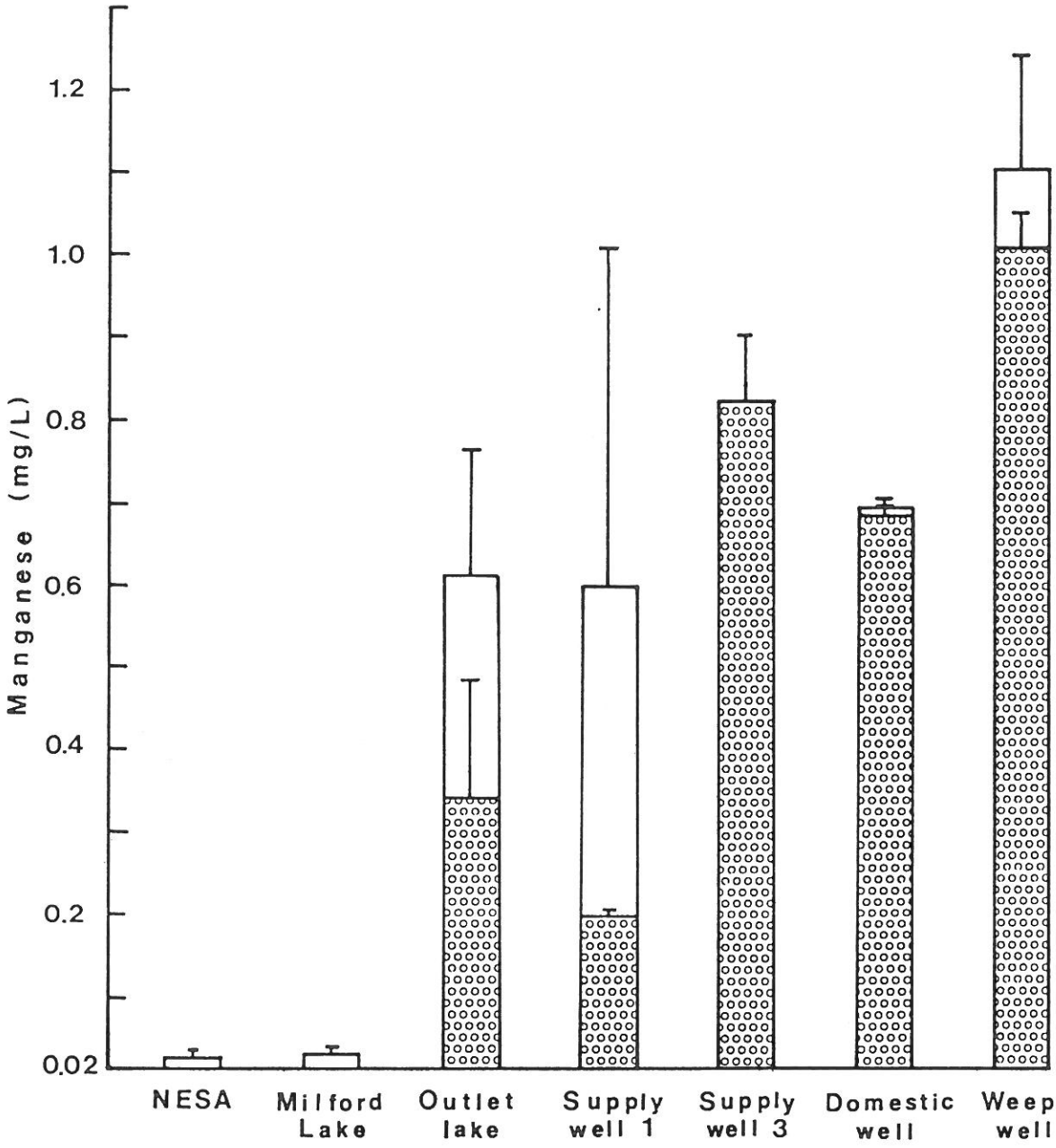

Figure 3. Means + one standard error of total (open bar) and soluble (shaded bar) manganese (mg/L) for seven water sources tested in two to seven bioassays.

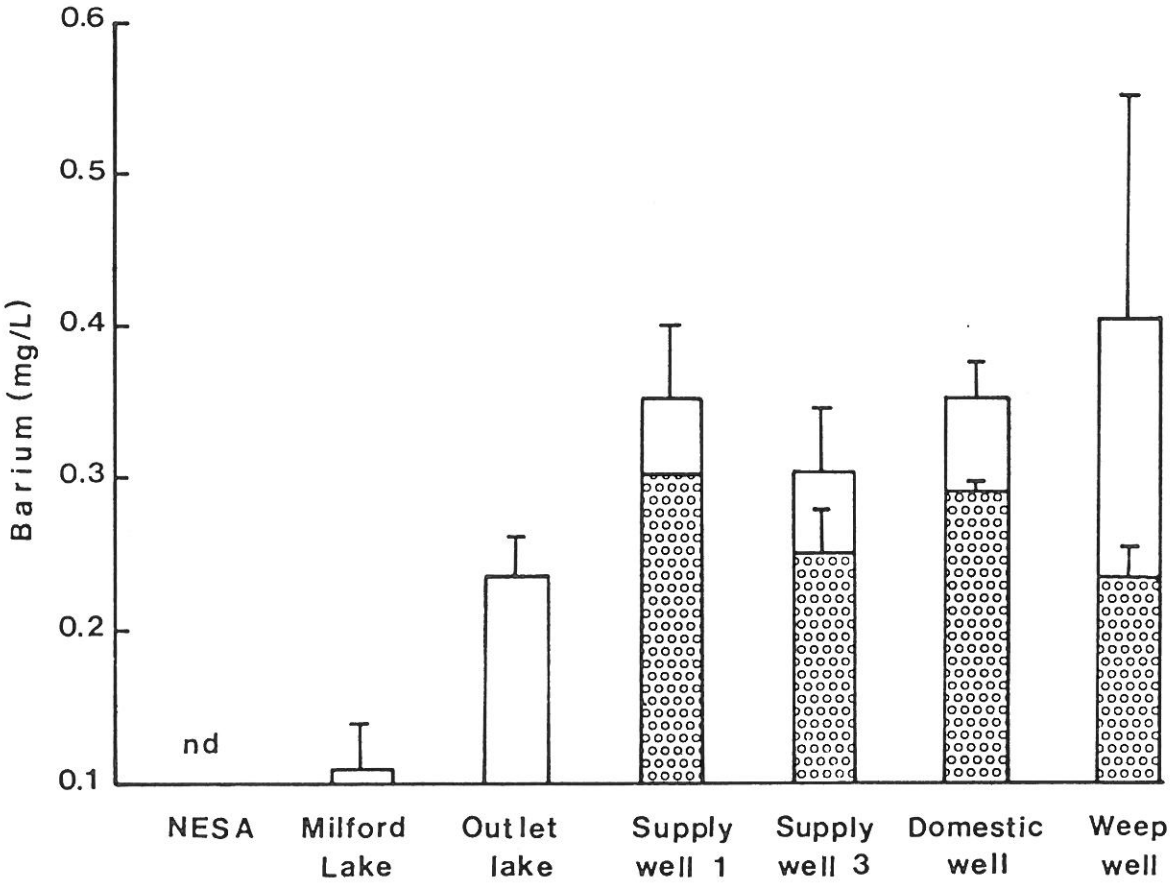

Figure 4. Means + one standard error of total (open bar) and soluble (shaded bar) barium (mg/L) for seven water sources tested in two to seven bioassays. Detection limit for total barium was 0.1 mg/L (nd = not detected).

only present in the groundwater supplies. However, soluble manganese was detected in the outlet lake water. It was the only metal which had a definite seasonal trend, being low in spring and early summer and high in winter and fall (Appendix 5).

The mean ammonia concentration was 0.08 mg/L in Milford Lake while all groundwater sources had ammonia ranging from 0.25 to 0.96 mg/L (Figure 5). The outlet lake and NESA water were usually below the detection limit of 0.05 mg/L. Weep well water usually had the greatest ammonia concentrations of all the water sources. Un-ionized ammonia was usually less than 10% of the total ammonia concentration. However, unionized ammonia in supply well and weep well water was sometimes greater than 0.025 mg/L.

In all of the samples analysed for this project, sulfide was detected in only three. In bioassay 1, sulfide was found in Milford Lake (0.5 mg/L) and NESA (2.0 mg/L) water samples. We believe these values are abberent because these samples are from highly oxygenated environments and subsequent analysis from other bioassays never found sulfide in Milford Lake or NESA water. In bioassay 4, sulfide was detected in the weep well water sample but a concentration was not specified. The detection limits for the sulfide analyses preformed by us and Wilson Laboratories were 0.003 and 0.1 mg/L, respectively. Both of these limits are above the level suggested as safe for aquatic life (EPA 1976). Thus, the possibility exits that deleterious amounts of sulfide were present in some water sources but were undetected.

Nutrient chemistry of groundwater and surface water. Nitrogen and phosphorus in groundwater wells were found to form a gradient of decreasing concentration below the dam of Milford Lake (Figure 6). Weep well concentrations were usually greater than in the supply wells. The latter

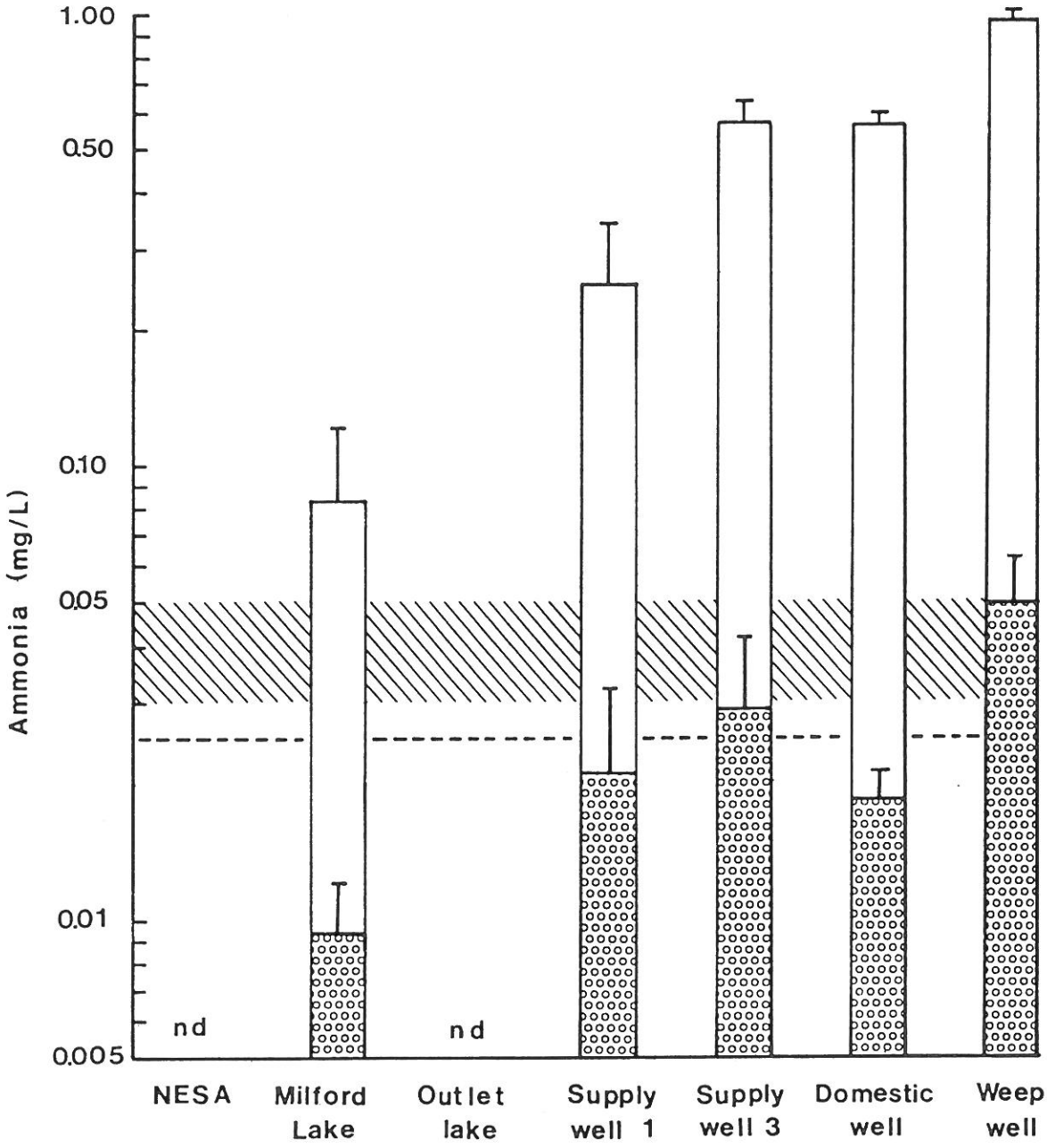

Figure 5. Means + one standard error of total (open bar) and un-ionized (shaded bar) ammonia (mg/L) for seven water sources tested in two to seven bioassays. For each bioasssay values for each water source were means of one to eight measurements taken during the 7-day bioassays. Dashed horizontal line is the maximum level of un-ionized ammonia recommended by Alabaster and Lloyd (1982). Cross hatched area is the range of maximaum concentrations of un-ionized ammonia recommended by EPA (1985). Detection limit for total ammonia was 0.05 mg/L (nd = not detected).

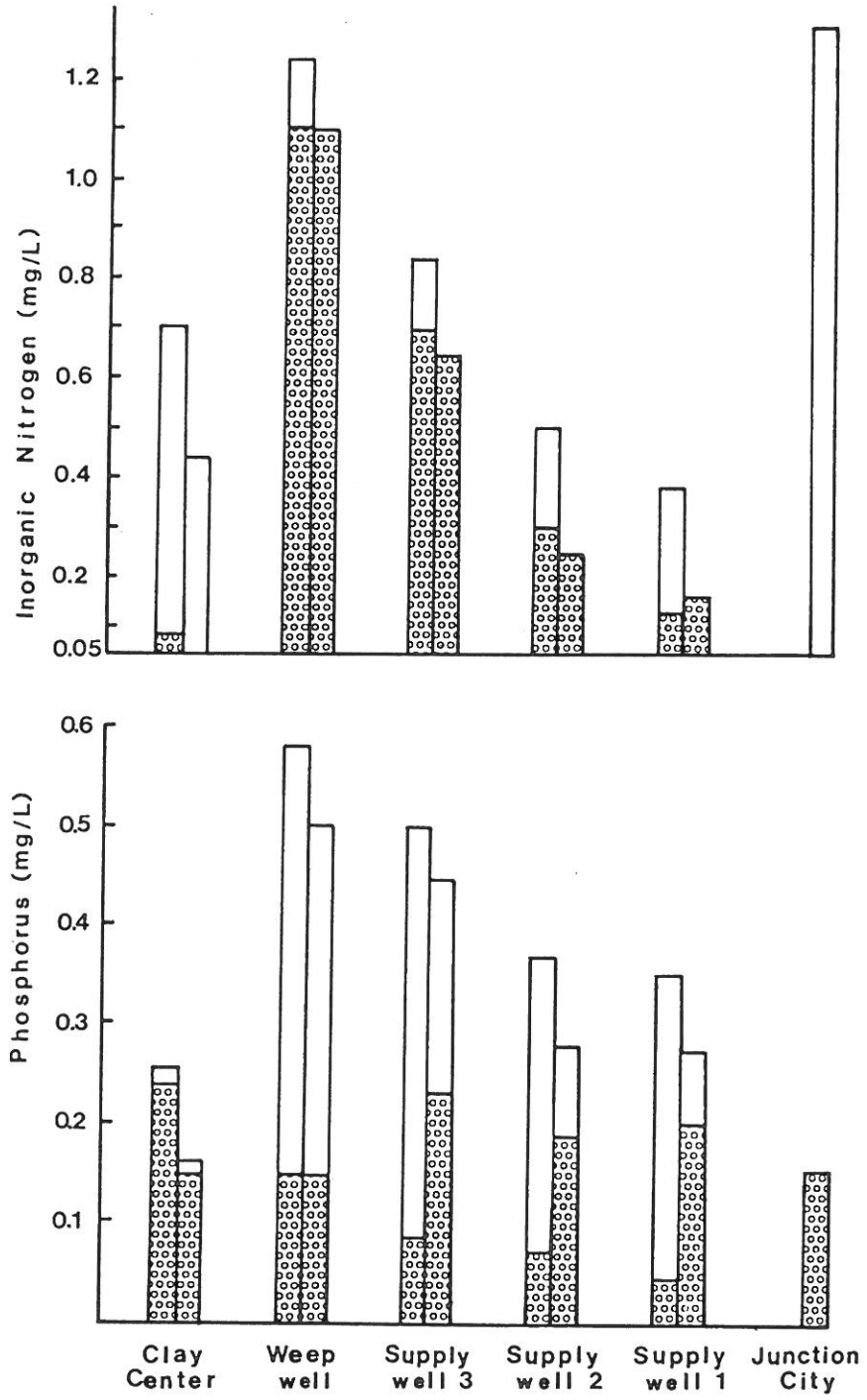

Figure 6. Concentrations (mg/L) of total inorganic nitrogen and total phosphorus (open bars) and total ammonia and soluble reactive phosphorus (shaded bars). Samples were taken in August and November 1986 (left and right adjacent bars, respectively) for six well water sites. Clay Center well is upstream of Milford Lake while other well sites are progressively further downstream (from left to right) of Milford Lake. No data from Junction City well in August.

decreased in order of distance from the Milford Lake dam. Groundwater from a Clay Center municipal well located upstream from Milford Lake had much lower phosphorus and ammonia concentrations than wells from the vicinity of the hatchery and from a well near Junction City which is about 5 km downstream from Milford Lake. When sediments from Milford Lake were tested, the amounts of nutrients released were greater on the south side of the main basin than on the north side (Table 5).

A horizontal gradient existed among the weep wells below Milford Lake along the toe of the dam. The central weep wells (e.g. #36) and corresponding lateral canal C had higher ammonia and total inorganic nitrogen concentrations than the most northern or southern sites (Table 6). Ammonia was also present in groundwater from weep wells below other eastern Kansas reservoir dams, however, the highest concentrations were found at Milford Lake (Table 7). At Tuttle Creek Reservoir, where three samples were taken, a gradient existed with ammonia increasing as distance from the old river channel decreased. These groundwater sources were typically low in dissolved oxygen, neutral in pH, and about 13 degrees C in temperature.

The groundwater from weep wells and nutrients therein feed into the outlet lake via four lateral canals. Rates of inflow to and outflow from the oulet lake were calculated (Table 8) and yielded a water retention time in the lake of 35 days. The rate of inflow from the weep wells is proportional to the elevation of the level of Milford Lake and we expect that the same is true for direct groundwater input. On 10 June 1986, when our measurements were made, the lake elevation was only 0.76 meters above conservation pool. Thus, our estimates of flow from the weep well of 139.6 L/sec should be reasonably respresentative of typical conditions. The nutrient loadings into the outlet lake were estimated to be 12.7 kg/day of total phosphorus and 17.4 kg/day of ammonia (Table 9). We could not

Table 5. Nutrients released (mg) per liter of Milford Lake sediments on 25 November 1986. Sediments were collected from the main basin about 300 m west of the dam.

Sediment source	Total NH_3	NO_2 + NO_3	Total inorganic nitrogen	Soluble reactive phosphorus
North side	3.42	0.575	3.99	2.18
South side	8.50	0.650	9.15	2.82

Table 6. Nutrient concentrations (mg/L) from groundwater sites arranged south to north along the toe of Milford Dam. Weep wells were sampled 25 November 1986 and lateral canals on 29 August 1986. Weep well 6 is near lateral canal A, weep well 36 is near lateral canal C, and weep well 72 is near lateral canal D. SRP = soluble reactive phosphorus.

Nutrient	Weep well 6	Lateral canal A	Lateral canal B	Weep well 36	Lateral canal C	Weep well 72	Lateral canal D
Total NH_3	0.34	0.21	0.71	1.0	1.1	0.93	0.76
NO_2 + NO_3	nd	0.31	0.21	nd	0.13	nd	0.20
Total P	0.47	0.42	0.57	0.51	0.58	0.56	0.52
SRP	0.28	0.44	0.46	0.41	0.15	0.55	0.23

Table 7. Chemistry of weep well water from four Kansas reservoirs on 12-13 November 1986. ($\bar{x}$ = means of weep wells)

Weep well source	pH	Temp. (C)	Dissolved oxygen (mg/L)	Total NH_3 (mg/L)	$NO_2 + NO_3$ (mg/L)	Total inorganic nitrogen (mg/L)
Marion	7.5	14.0	6.8	0.136	0.402	0.538
Tuttle Creek	7.3	-	-	0.140	0.473	0.613
	7.1	13.0	0.5	0.239	0.479	0.718
	7.4	-	-	0.655	0.695	1.35
$\bar{x}$ =	7.3			0.345	0.549	0.894
Perry	6.8	12.4	2.0	0.482	0.568	1.05
Milford	7.5	-	-	1.29	0.700	1.99
	7.5	12.5	0.5	1.43	0.850	2.28
	7.7	-	-	0.542	0.668	1.21
$\bar{x}$ =	7.6			1.087	0.739	1.83

Table 8. Hydrologic inflow and outflow to the outlet lake measured on 10 June 1986.

	Gallons/min	Liters/sec	% Total
Inflow			
Lateral canals			
A	224	14.1	5.3
B	1245	78.5	29.4
C	485	30.6	11.5
D	260	16.4	6.2
Subterranean input	2012	127.0	47.6
Total input	4226	266.6	100.0
Outflow			
Outfall	526	32.2	12.5
Hatchery withdrawal	3700	233.4	87.5
Total outflow	4226	266.6	100.0

Outlet lake volume = 8.0×10^7 m^3

Water residence time in lake = 35 days

measure the concentration of nutrients in the water which entered the outlet lake as direct groundwater input. Our calculations of loadings are based on the assumption that the nutrient concentrations in the direct groundwater input are equal to those in the weep wells. Although nutrient-enriched water enters the outlet lake, concentrations in the outlet lakeare not high (Table 10). Throughout the March to December sampling period ammonia in the outlet lake was only detected on two dates with a maximum of 0.13 mg/L occurring in December (Appendix 5).

Table 9. Total nutrient loading of nitrogen and phosphorus (kg/day) into the outlet lake. These were calculated from nutrient concentrations of each source meansured on 29 August 1986 and hydrologic measurements made on 10 June 1986. Nitrate and nitrite were not detected in weep well water on that date, therefore, all the the total inorganic nitrogen was in the form of ammonia.

Source	Total phosphorus	Total inorganic nitrogen
Lateral canals		
A	0.512	0.250
B	3.87	4.83
C	1.55	2.96
D	0.737	1.08
Sum of lateral canals	6.66	9.12
Subterranean input	6.06	8.29
Total input	12.7	17.4
Areal loading ($g/m^2/yr$)	10.5	14.3

Table 10. Total inorganic nitrogen (mg/L), percent ammonia, total phosphorus (mg/L) and ratio of N:P on two dates from surface water of the outlet lake compared to Milford Lake and weep wells or lateral canals.

	Total inorganic nitrogen	Percent ammonia	Total phosphorus	N:P
29 August 1986				
Milford Lake	0.94	2	0.17	5.6
Lateral canal mean (n=4)	0.91	77	0.52	1.7
Lateral canal C	1.2	90	0.58	2.1
Outlet lake--upper basin	0.33	5	0.57	0.58
lower basin	0.31	5	0.40	0.78
25 November 1986				
Milford Lake	0.86	<1	0.15	5.8
Weep well mean (n=3)	0.61	100	0.51	1.2
Weep well 36	0.81	100	0.51	2.0
Outlet lake--lower basin	nd(0.05)		0.24	<0.04

DISCUSSION

A wide range of concentrations of potentially toxic compounds were present in the water tested in the fish bioassays. However, no significant differences or consistent trends in growth or survival of the larval fish existed among the treatments. Therefore, we cannot isolate a specific chemical water quality parameter or group of chemical parameters (e.g., ammonia, iron, or manganese) which was solely responsible for poor production of fish at the hatchery.

Several plausible explanations exist for the apparent contradiction between the mortality observed at the Milford Hatchery and our bioassay results. The tests we performed did not mimic the physical conditions in the hatchery raceways. Since the bioassays were static tests, water currents were nonexistent and suspended particulate matter was allowed to settle out. The physical crowding of fish was also much less in the bioassays. It is possible that physical conditions of the water interacted with chemical parameters to produce a harmful effect. A hypothetical example is that ammonia and suspended particulate iron combined to produce the observed gill irritation.

In addition, it is possible that incoming source water and its associated water quality constituents become chemically altered (e.g. changes in concentration or species) within the hatchery water delivery system and/or raceways. The presence of microorganisms or other aquatic organisms (e.g. periphyton) within the hatchery system may impact water quality. The uptake and release of various chemical species or parameters might result in a deterioration of the quality of incoming water. For example, ammonia in culture water can and often does originate from the mineralization of organic substances by heterotrophic bacteria and excretion by the animals. Ammonia is the main form of nitrogen excreted by

fish (Smith, 1929) and most aquatic invertebrates (e.g. Robertson 1954; Dresel and Moyle 1950). Certainly the potential exists for the deterioration of various water quality parameters under hatchery conditions. These possibilities were not addressed in this study because we were directed toward the evaluation of the water quality of source waters and not hatchery conditions.

The results obtained in a toxicity test (bioassay), in large part, depend on the conditions and nature of exposure; they are the product of operationally defined procedures. The usefulness and relibility of the 7-day, fathead minnow larval survival and growth test to estimate the effects of toxicants and/or other water quality conditions is well documented in Horning and Weber (1985). It is also well documented that the embryo-larval and early juvenile life-stages of most fish were the most sensitive stages (e.g. McKim 1977; Macek and Sleight 1977). Furthermore, research by Norberg and Mount (1985) with the 7-day fathead minnow larval test indicated that results from this short-term test procedure were comparable to and had confidence intervals that overlapped with, literature values for partial and full life cycle tests. However, it is possible (but improbable) that longer-term exposures of test fish to the various water sources at Milford Hatchery might have prompted different results than those observed during our investigation.

It is possible that the water quality at the Milford Hatchery, particularly in the outlet lake, has improved between 1985 and 1986. Evidence for this are the ammonia concentrations found in the lower basin of the outlet lake. In September 1985 the concentration of total ammonia was 0.91 mg/L. In September 1986 it was 0.07 mg/L. In spring of 1984 the lake was drained for construction of the hatchery and was not refilled until fall 1984. In 1985, it was possible that the balance among such

processes as ammonia input from the source waters, ammonia uptake by the phytoplankton, ammonia release by decay, and ammonia removal by nitrifying bacteria was not yet re-established. The result of these imbalances could have been the accumulation of ammonia in the water. In 1986, it is possible that the microbial, floral, and faunal communities had recovered to the point that they could fully exploit the nitrogen resources of the outlet lake causing the virtual depletion of ammonia.

The large standing crop of phytoplankton in the outlet lake is caused by high nutrient loadings. These loadings of nitrogen and phosphorus are, respectively, approximately one and two orders of magnitude greater than loadings which typically cause eutrophication in lakes (Vollenweider 1968, 1976).

Phytoplankton typically require a ratio of nitrogen (N) to phosphorus (P) in in excess of 10:1. The N:P ratio in the inflowing groundwater is approximately 2:1 which suggests that phytoplankton growth in the outlet lake is nitrogen limited. This condition will cause the depletion of inorganic nitrogen in the water column and favors the development of algae which can fix nitrogen. Many of the nitrogen fixing phytoplankton are bloom-forming, blue-green algae such as Anabaena and Aphanizomenon. Some strains of these algae are known to excrete compounds toxic to vertebrates (Prescot, 1970).

The proximal source of the nutrients for the outlet lake is the groundwater. However, two observations support the hypothesis that the ultimate source of these nutrients is the sediments of Milford Lake. Nutrient concentrations in the groundwater immediately downstream of Milford Lake were greater than those upstream. Also, the same overwhelming abundance of ammonia relative to nitrate and nitrite found in the sediment elutriates is found in the groundwater flowing from the weep wells.

The enrichment of the groundwater flowing out of the weep wells is greatest from near the center of the dam up to the north end. This area corresponds to the area behind the dam where Milford Lake is deepest. This is probably the area of the lake with the greatest sediment accumulation. Nutrient concentrations in the groundwater drop rapidly as distance from the dam increases. Thus, at the furthest east supply well, the nutrient levels are approximately the same as those upstream of Milford Lake.

RECOMMENDATIONS

Hatchery water supply. Based on the results of the bioassays, treatment of the water at the hatchery to remove a particular chemical compound is not justified. However, it is quite likely that the high load of suspended particulate matter (inorganic in the groundwater, organic in the lake water), is interacting with a chemical water quality parameter(s) which by itself is not harmful, to produce a deleterious effect on the catfish. In addition, the suspended material settles out in the raceways and contributes to the growth of periphyton and other attached organisms in the raceways and water distribution system, thus, increasing the amount of time needed for cleaning raceways and water lines. The increase in clarity caused by removal of the suspended matter would improve the ability of visually orienting fish to see their food. This would enhance the production of piscivorous fish, such as largemouth bass, which are among plans for future hatchery operations. In addition, removal of most of the phytoplankton by a clarifying system would allow outlet lake water to be passed through the sand filters inside the hatchery and used in the egg hatching and fingerling rearing operations. Currently, only groundwater or unfiltered lakewater can be used for these activities. Therefore, we recommend the evaluation of the potential benefits derived from the

installation of a water clarifying system similar to sedimentation basins or clarifiers utilized in water and wastewater treatment. A biological assessment would most likely be needed to determine the impact of suspended materials on hatchery production. Ultimately, an engineering study would be required to determine the exact configuration of this system if such a system was warranted.

The identification of a water quality gradient within the hatchery supply wells leads to two recommendations. Even though water from supply well 3 did not cause a significant reduction in fish growth in the bioassays, the concentrations of un-ionized ammonia was frequently within the range of maximum levels recommended by EPA. Considering also the high levels of suspended particulate matter such as oxidized iron, manganese, and barium, that could be interacting with the ammonia, we recommend that supply well 3 be used only when absolutely necessary.

The existing configuration of the Milford Hatchery was not designed for, nor does current demand call for, the use of all 24 raceways simultaneously. However, it is likely that new water sources will be needed in the future. Because of the water quality gradient we observed in the groundwater below Milford Lake, we recommend that any new supply wells for the hatchery be located as far from the dam as possible.

Outlet lake. The nutrient loadings into the outlet lake are extremely high resulting in large standing crops of phytoplankton. The intense photosynthesis which occurs during the day in warm weather results in large diurnal changes in dissolved oxygen and pH. The elevated pH in the day and low dissolved oxygen concentrations at night could create harmful conditions for fish in the raceways. Thus, it would be desirable to reduce phytoplankton production in the outlet lake. Two basic strategies can be

used to moderate the lake's eutrophic condition: one is to curtail the input of nutrients; the other is to directly reduce the standing crop of phytoplankton. The use of one strategy does not preclude the use of the other.

Nutrients enter the lake dissolved in groundwater which supplies the lake. We have measured the nutrients in the weep well water and, for the purposes of calculation, assumed that the same concentration of nutrients existed in the groundwater which enters the outlet lake directly. This is a conservative assumption because the lake is located further from the dam than the weep wells. The existence of a nutrient gradient within the groundwater suggests that the actual nutrient concentrations in the direct groundwater input may be less than in the weep well water. Assuming equal concentrations, the direct groundwater input into the outlet lake contributes slightly less than half of the nutrients entering the lake. It is likely that no feasible method exists to reduce this input. However, it would be relatively simple to divert the flow of weep well water away from the outlet lake and into the old Republican River channel. If this water was diverted, another source would have to be found if the outlet lake were to remain a useful water supply for the hatchery. The only convenient source of replacement water is Milford Lake. Replacing the weep well water with Milford Lake water would require 2.3×10^7 liters of water per day. This is nearly 50% of the required low flow discharge from Milford Lake.

We measured the nutrient concentrations in the water coming out of Milford Lake and found it to contain 0.16 mg/L of total phosphorus and 0.951 mg/L total inorganic nitrogen (means of measurements made in August and November). Only 2% of the nitrogen was in the form of ammonia. If water from the outflow of Milford Lake were used to replace the weep well water, the phosphorus loading in the lake would be reduced by 37% but the

inorganic nitrogen loading would be unchanged. The N:P ratio would increase from 1.7:1 to 2.1:1 which indicates that the growth of the phytoplankton would still be limited by nitrogen. Since the amount of available nitrogen would not be changed, it is unlikely that phytoplankton production would be reduced.

If the amount of water from Milford Lake directed into the outlet lake were increased above what was needed to replace the weep well water, the nutrient loading and the N:P ratio would increase. The concentration of phosphorus in the lake would decrease while the nitrogen concentration would remain the same because of the decreased retention time. The N:P ratio in the Milford Lake outlfow water is 5.8:1. Thus, no matter how much Milford water was diverted into the outlet lake, phosphorus might never become limiting (e.g. N:P > 10:1) and since the concentration of inorganic nitrogen would remain the same, no improvement in the lake would be expected.

This scenario is based on the assumption that N:P ratios are truly indicative of nutrient limitation. However, as is usually the case with ecosystems, simple relationships are the exception rather than the rule. Therefore, in order to predict with more certainty the response of the outlet lake phytoplankton community to diverting the weep well water and adding Milford Lake water, a study designed specifically to address that question would have to be performed.

Although the eutrophic condition of the outlet lake may not be reduced by diverting the weep well water from the outlet lake, adding water from the outflow of Milford Lake would benefit the hatchery. The additional input into the outlet lake would allow more water to be withdrawn for hatchery operations without lowering the outlet lake level. The addition

of Milford Lake zooplankton could play a role in reducing the standing crop of phytoplankton in the outlet lake.

The other strategy of reducing phytoplankton in the outlet lake is to enhance the abundance of organisms that feed on phytoplankton. This technique is called biomanipulation. Two groups of organisms which feed on plankton have been considered for this task: filter-feeding fish and herbivorous zooplankton. A common filter-feeding fish is the gizzard shad. At standard lengths (SL) greater than 2.5 cm, it is an indiscriminant pump filter feeder consuming phytoplankton, zooplankton, and detritus (Kutkuhn 1957, Baker and Schmitz 1971). Unfortunately, the efficiency at which shad eat phytoplankton is not consistent throughout their lives. Small shad, less than 6 cm SL can effectivly filter particles the size of typical phytoplankters (ca 20 μm, Mummert and Drenner 1986). Several food habit studies have found that phytoplankton dominate the diet of small shad (e.g. Tiffany 1921, Cramer and Marzolf 1970, Barger and Kilambi 1980). However, the diet shifts to detritus and zooplankton as the size of shad increases (Kutkuhn 1957, Pierce et al. 1981).

Experimental studies have found that shad only suppressed the populations of large, single celled phytoplankters (Drenner et al. 1974) and either did not effect or actually enhanced smaller algae (Drenner et al. 1984, Drenner et al. 1986). The blue-green bloom forming filamentous algae Anabaena was also not efficiently eaten by shad (Drenner et al. 1986). The impact of shad on the zooplankton community has been shown to be the suppression of rotifers, cladocerans, copepod nauplii, and cyclopoid copepodids and adults and the enhancement of some calanoid copepods (Drenner et al. 1982). The common factor is swimming ability of the zooplankters. Slow swimming types are more easily captured by the pumping

action of the shad whereas the rapid swimming types are more successful at evasion (Drenner and McComas 1980).

A recent experiment comparing the effects of shad and a size-selective, visually orienting zooplanktivorous fish found that both fish reduced zooplankton biomass but only the shad enhanced the phytoplankton biomass (Drenner et al. 1986). The reason for the increase in phytoplankton biomass in the presence of shad was thought to be the release of nutrients from excreta or dead fish.

One of the groups of zooplankters that planktivorous fish selectively remove are Daphnia (Drenner et al. 1986). These relatively large herbivorous zooplankters can have a significant impact on phytoplankton biomass. Removal of Daphnia by fish have caused large increases in phytoplankton biomass (Lynch and Shapiro 1981). Conversely, the removal of fish has caused an increase in the abundance of Daphnia and a reduction in phytoplankton biomass (Shapiro et al. 1983, Shapiro and Wright 1984).

The zooplankton community we observed in the outlet lake was dominated by large, fast swimming, copepods. Large herbivorous cladocerans were absent. This qualitative assessment is based on a sample collected in February 1986 and on observations of zooplankters in the outlet lake water samples collected throughout the course of the study and used in the bioassays. This zooplankton community structure is consistent with predation by filter feeding fish. Although gizzard shad are present in the outlet lake, their numbers have only recently become great. We believe that it was the bigmouth buffalo that was originally responsible for the lack of large cladocerans in the lake. Bigmouth buffalo have food habits very similar to gizzard shad (Starostka and Applegate 1970) and were extremely abundant in the outlet lake as indicated by a recent commercial fish harvest.

The removal of planktivorous fish from the outlet lake might be a useful technique for controlling the phytoplankton. However, several potential drawbacks exist. Although large zooplankton herbivores have been shown to reduce the biomass of many phytoplankton species, Aphinizomenon flos-aquae (a filamentous blue-green alga) was sometimes enhanced (Lynch and Shapiro 1981). A practical difficulty exists with this method in that it may be impossible to remove all or most of the planktivorous fish and prevent their return. Reduction of their numbers may be possible, however, by netting of some species (e.g. bigmouth buffalo, river carpsucker, and common carp) and enhancing the piscivore population by stocking to reduce forage species (e.g. gizzard shad). In conjunction with a fish manipulation, diverting Milford Lake water into the outlet lake would be useful because it would introduce a steady supply of large herbivorous zooplankton (e.g. Daphnia).

The biomanipulation studies we have cited have been done on lakes which were only moderately eutrophic. No experience exists on lakes which receive nutrient loadings as high as the outlet lake. Thus, before any large scale manipulation of the fish population is attempted, it would be prudent to first conduct pilot studies using enclosures in the lake. However, relatively little expense, and therefore relatively little risk, would be involved in continuation of the commercial harvest and the stocking of piscivorous fish. Some requirements of the predator to be stocked would be that it grows large enough in a reasonably short time in order to eat large shad, and that its own planktivorous life stage be as short as possible. Largemouth bass, walleye, northern pike, white bass and white bass/striped bass hydrids might all be useful for controlling the gizzard shad populations.

Among our recommendations are the need for further research to predict the outcome of certain remedial measures. One study would assess the effects of removing suspended particulate matter from the raceway environment. This would probably be accomplished by simulating conditions in a model raceway. Another study would be a detailed analysis of nutrient limitation of phytoplankton in the outlet lake and simulation of possible restoration techniques using in situ enclosures. In addition to these studies, a water quality monitoring regime for the outlet lake should be established. The parameter measured should include pH, ammonia, conductivity, alkalinity, DO, temperature, a measure of clarity, and a measure of phytoplankton abundance (i.e., biomass, chlorophyll, or fluorescence). These parameters should be measured bi-weekly when the hatchery is on groundwater and at least weekly when lake water is being used. Sample collection and measurement should be made in the late afternoon. Additional measurements at pH and DO should be made within a half hour of dawn in order to determine diurnal changes. Monthly samples of phytoplankton and zooplankton should also be collected and analyzed.

Most of the techniques we have suggested to improve water quality at the hatchery and in the outlet lake call for major expenditure for construction, manpower, or both. Further, many of the techniques will require prior study to fully assess their feasibilities. To make matters more complicated, no data exists to ascertain the stability of current water quality conditions. For example, it is unknown if the plume of nutrient enriched groundwater extending downstream from Milford Lake will expand over the life time of the hatchery and impact all the existing supply wells. Phytoplankton abundance within the outlet lake may also change over time perhaps in response to expansion and contraction of the shad population. Given the expense of installing a water quality treatment

facility at the hatchery; the expense and uncertain effects of water diversion and biomanipulation; the uncertainties concerning the future quality of existing water; and the lack of direct toxic effects on fish of the hatchery source waters, we feel that the prudent course of action at this time is to thoroughly review fish production at the hatchery in relation to management techniques. It is possible that the perceived water quality problems can be overcome with appropriate management. Obviously, it will require time to develop the techniques suitable for the set of conditions unique to the Milford Hatchery. Therefore, our final recommendation is that all methods to improve production at the hatchery through refinement of hatchery management techniques should be exhausted before major investments are made to improve water quality.

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APPENDICES

Appendix 1. Methods for analysing water quality parameters. ISE = ion specific electrode.

Parameter	Method
Dissolved oxygen	Clark polarographic cell with meter, YSI model 5739 probe and YSI model 57 or 58 meter.
pH	Orion model EA 940 ISE meter and Orion Ross electrode.
Conductivity	YSI model 33 meter.
Alkalinity	Colorimetric titration with sulfuric acid (Lind 1979).
Hardness	Colorimetric titration with EDTA (Lind 1979).
Phosphorus: SRP, total	Spectrophotimetric analyses using molybdate blue/ascorbic acid method (Wetzel and Likens 1979).
Ammonia: total	Orion ISE meter and Phoenix or Orion ammonia electrode (APHA, 1986).
Ammonia: unionized	Calculated, see Appendix 2.
Total inorganic nitrogen	Orion ISE meter and ammonia electrode after reduction of all inorganic nitrogen to ammonia using titanous chloride.
Nitrate plus nitrite	Calculated, total inorganic nitrogen minus total ammonia.
Sulfide	Orion ISE and Phoenix sulfide electrode.
Sediment elutriation	Two hundred ml of fresh sediments and 800 ml of distilled water were place in a 1 L screw cap plastic jar. The jar was placed on a device and rotated at 25 rpm for 24 h. The jar was then centrifuged at about 4000 rpm for 10 min. Chemical analyses were performed on the supernatant (EPA/Corps of Engineers 1977).

Appendix 2. Calculation of un-ionized ammonia concentration, from total concentration, pH, temperature, and conductivity (from Colt and Tchobanoglous 1978).

$$NH_3 = \frac{(\text{total ammonia})}{1 + 10^{(pK'a - pH)}}$$

$$pK'a = pKa - \log\gamma_{NH_4}$$

$$pKa = 0.9018 + 2729.22/(T + 273.15)$$
where T = degrees C

$$\log\gamma_{NH_4} = AZ^2\sqrt{I}/(1 + \sqrt{I}) - 0.3(I)$$
where A = 0.5
Z = 1
I = 0.013(EC)
where EC = conductivity in millimhos/cm at 25 degrees C

Appendix 3. Standard water quality parameters in the bioassays. Each value represents the mean of 3 to 7 replicates analysed during the course of each bioasay. - = treatment not included in bioassay.

Bioassay	NESA	Domestic well	Supply well 1	Supply well 3	Supply (mixed)	Weep well	Outlet lake	Milford Lake
				Conductivity (umhos/cm)				
1	254	591	-	-	591	634	599	601
2	322	594	-	-	600	611	499	601
3	304	594	-	604	-	617	464	601
4	287	622	-	628	-	650	467	554
5	240	-	-	698	-	690	593	508
6	221	-	597	632	-	600	593	385
7	205	-	565	580	-	588	597	432
				pH				
1	7.6	7.6	-	-	7.7	7.8	8.2	8.3
2	8.2	8.1	-	-	7.9	8.1	8.3	8.4
3	8.2	7.8	-	7.7	-	7.9	8.2	8.4
4	8.0	7.7	-	7.6	-	7.5	7.9	8.2
5	8.7	-	-	7.7	-	7.6	8.4	8.2
6	8.4	-	8.1	8.0	-	8.1	8.6	8.4
7	8.1	-	8.3	8.4	-	8.5	8.6	8.4
				Total Hardness (mg/L as CaCO)				
1	136	269	-	-	289	288	297	246
2	165	261	-	-	283	282	198	242
3	159	268	-	279	-	281	183	242
4	121	206	-	287	-	274	163	197
5	92	-	-	278	-	273	235	181
6	100	-	305	284	-	279	288	179
7	102	-	288	284	-	274	291	191
	Alkalinity (mg/L as CaCO), total (phenolphthalein)							
1	119	283	-	-	287	283	285	192
2	156	281	-	-	290	284	199	191
3	155(4)	300(0)	-	304(0)	-	302(4)	197(5)	199(10)
4	119(0)	288(0)	-	301(2)	-	303(0)	157(3)	153(3)
5	88(7)	-	-	303(0)	-	310(0)	253(11)	148(1)
6	102(6)	-	305(7)	304(5)	-	295(5)	299(16)	150(5)
7	95(0)	-	302(5)	301(9)	-	293(12)	305(15)	161(5)

Appendix 4. Compounds and elements analyzed for but not detectable in any of the water sources used in bioassay 1. Detection limits are in parentheses, units for heavy metals are mg/L, all others are ug/L.

Herbicides	Insecticides	Organics	Metals
2,4-D (0.5)	Aldrin (0.1)	PCB-1242 (1.0)	Arsenic (0.01)
Ramrod (1.0)	Lindane (0.1)	PCB-1254 (1.0)	Cadmium (0.01)
Dual (1.0)	Chlordane (1.0)	PCB-1221 (1.0)	Chromium (0.01)
Lasso (1.0)	DDT (0.1)	PCG-1232 (1.0)	Copper (0.02)
	DDE (0.1)	PCB-1248 (1.0)	Iron (0.1)
	DDD (0.1)	PCB-1260 (1.0)	Lead (0.005)
	Dieldrin (0.1)	PCB-1016 (1.0)	Selenium (0.005)
	Endosulfan (0.1)		Silver (0.01)
	Endrin (0.1)		Zinc (0.02)
	Heptachlor (0.1)		
	Toxaphene (5.0)		

Appendix 5. Concentrations of potentially deleterious compounds in water used in bioassays. nd = not detectable with the detection limits in parentheses. na = not analysed. - = treatment not included in bioassay. Metals analyses for bioassays 3 and 4 were performed by the Army Corps of Engineers.

Bioassay	NESA	Domestic well	Supply well 1	Supply well 3	Supply (mixed)	Weep well	Outlet lake	Milford Lake
				Total ammonia (mg/L)				
1	nd(0.1)	0.6	-	-	0.3	1.1	nd(0.1)	0.1
2	0.060	0.445	-	-	0.196	0.766	nd(.05)	0.135
3	nd(.05)	0.567	-	0.625	-	1.04	nd(.05)	0.249
4	0.14	0.62	-	0.44	-	1.05	nd(.05)	nd(.05)
5	nd(.05)	-	-	0.58	-	0.94	0.06	nd(.05)
6	nd(.05)	-	0.16	0.56	-	0.90	nd(.05)	nd(.05)
7	0.13	-	0.34	0.68	-	0.91	0.13	0.10
				Un-ionized ammonia (mg/L)				
1	nd	0.012	-	-	0.0076	0.0346	nd	0.0093
2	0.0046	0.027	-	-	0.0077	0.0467	nd	0.0155
3	nd	0.018	-	0.0158	-	0.0409	nd	0.0286
4	0.0071	0.016	-	0.0088	-	0.0168	nd	nd
5	nd	-	-	0.0145	-	0.0188	0.0069	nd
6	nd	-	0.0098	0.0274	-	0.0549	nd	nd
7	0.0082	-	0.0318	0.0782	-	0.1279	0.02217	0.0116
				Total iron (mg/L)				
1	na	na	-	-	na	na	na	na
2	0.02	2.2	-	-	2.1	2.1	0.1	0.2
3	0.224	2.133	-	2.553	-	1.653	0.200	0.095
4	0.620	3.460	-	2.315	-	10.100	na	0.143
5	0.1	-	-	3.0	-	2.5	0.3	0.7
6	0.1	-	1.0	1.5	-	0.9	0.1	1.0
7	0.2	-	2.4	2.8	-	1.9	0.2	0.6
				Soluble iron (mg/L)				
1	nd(0.1)	nd(0.1)	-	-	nd(0.1)	nd(0.1)	nd(0.1)	nd(0.1)
2	nd(0.1)	0.4	-	-	nd(0.1)	nd(0.1)	nd(0.1)	nd(0.1)
3	0.011	0.363	-	0.022	-	0.035	nd(0.1)	nd(0.1)
4	0.036	0.827	-	0.014	-	0.046	na	0.023
5	nd(0.1)	-	-	1.7	-	nd(0.1)	nd(0.1)	nd(0.1)
6	nd(0.1)	-	nd(0.1)	nd(0.1)	-	nd(0.1)	nd(0.1)	nd(0.1)
7	nd(0.1)	-	nd(0.1)	nd(0.1)	-	nd(0.1)	nd(0.1)	nd(0.1)
				Total manganese (mg/L)				
1	na	na	-	-	na	na	na	na
2	0.02	0.71	-	-	0.26	1.06	0.22	0.03
3	0.058	0.650	-	0.837	-	0.940	0.426	0.013
4	0.058	0.708	-	0.551	-	1.780	na	1.121
5	nd(.02)	-	-	0.985	-	0.95	0.56	0.09
6	0.02	-	1.01	0.21	-	0.99	0.90	0.02
7	nd(.02)	-	0.19	0.96	-	0.95	1.00	nd(.02)

Appendix 5. (continued).

Bioassay	NESA	Domestic well	Supply well 1	Supply well 3	Supply (mixed)	Weep well	Outlet lake	Milford Lake
				Soluble manganese (mg/L)				
1	nd(.02)	0.69	–	–	0.32	1.11	0.46	nd(.02)
2	nd(.02)	0.69	–	–	0.26	1.07	nd(.02)	nd(.02)
3	nd(.005)	0.65	–	0.716	–	0.940	nd(.005)	nd(.005)
4	0.008	0.704	–	0.543	–	1.250	na	nd(.005)
5	nd(.02)	–	–	0.95	–	0.95	0.15	nd(.02)
6	nd(.02)	–	0.21	0.96	–	0.86	0.72	nd(.02)
7	nd(.02)	–	0.18	0.94	–	0.92	0.72	nd(.02)
				Particulate manganese (mg/L)				
1	na	nd	–	–	na	na	na	na
2	0.02	0.02	–	–	0.0	0.0	0.22	0.03
3	0.058	0.00	–	0.121	–	0.0	0.426	0.013
4	0.05	0.004	–	0.008	–	0.53	na	0.021
5	nd	–	–	0.0	–	0.0	0.41	0.09
6	0.02	–	0.0	0.5	–	0.13	0.18	0.02
7	nd	–	0.01	0.02	–	0.03	0.28	nd
				Total barium (mg/L)				
1	na	na	–	–	na	na	na	na
2	na	na	–	–	na	na	na	na
3	0.128	0.377	–	0.367	–	0.319	0.244	0.113
4	0.129	0.330	–	0.015	–	0.992	na	0.128
5	nd(0.1)	na	–	0.3	–	0.2	0.2	nd(0.1)
6	nd(0.1)	–	0.3	0.3	–	0.2	0.2	0.1
7	nd(0.1)	–	0.4	0.4	–	0.3	0.3	0.2
				Soluble barium (mg/L)				
1	0.10	0.28	–	–	0.34	0.26	0.26	0.12
2	na	na	–	–	na	na	na	na
3	0.128	0.286	–	0.303	–	0.258	0.203	0.109
4	0.128	0.299	–	na	–	0.295	na	0.106
5	nd(0.1)	–	–	0.2	–	0.2	0.2	nd(0.1)
6	nd(0.1)	–	0.3	0.2	–	0.2	0.2	nd(0.1)
7	nd(0.1)	–	0.3	0.3	–	0.2	0.2	0.1