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Microbial Enhanced Waterflooding Pilot Project, Mink Unit, Delaware-Childers (OK) Field

Topical Report

By
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D. M. Dennis
D. O. Hitzman

August 1991

Performed Under Cooperative Agreement No. DE-FC22-83FE60149

IIT Research Institute
National Institute for Petroleum and Energy Research
Bartlesville, Oklahoma

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U. S. DEPARTMENT OF ENERGY
Bartlesville, Oklahoma

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MICROBIAL ENHANCED WATERFLOODING PILOT PROJECT MINK UNIT, DELAWARE-CHILDERS (OK) FIELD

By R.S. Bryant[1], T.E. Burchfield[1], D.M. Dennis[2], and D.O. Hitzman[3]

ABSTRACT

The first microbial-enhanced waterflood field project sponsored by the U.S. Department of Energy (DOE), Microbial Systems Corp. (MSC), and INJECTECH, Inc., and being conducted in cooperation with the National Institute for Petroleum and Energy Research (NIPER) was initiated in October of 1986. One of the major goals of this project was to develop a technology that could be implemented by independent oil producers; thus, the field site chosen for the pilot test was representative of a mid-continent waterflood operation with stripper wells. The methodology for designing and optimizing microbial enhanced oil recovery (MEOR) field technology has yet to be established; however, literature information and experience with MEOR processes indicate that certain procedures are necessary to implement a microbial waterflooding process.

The site selected for the project is in the Mink Unit of Delaware-Childers field in Nowata County, Oklahoma. This field is typical of mid-continent reservoirs in the United States. The pilot area consists of four adjacent inverted five-spot patterns drilled on 5-acre spacing. There are 21 injection and 15 production wells on this pilot. Four of the 21 injection wells were treated with NIPER's microbial formulation.

Laboratory screening criteria were developed to evaluate microorganisms for this project. Several different microbial formulations were tested in Berea sandstone cores with reservoir fluids to determine oil recovery efficiency. Baseline monitoring of oil production was conducted to establish pre-pilot conditions, and fluid samples were collected on a weekly basis from producing wells.

Injectivity and microbial field survivability tests were conducted during the baseline period on two off-pattern wells, and a chemical tracer, fluorescein, was injected into the four injection wells during the baseline period. Tracer was observed in production wells about 1.8 years after injection, which corresponded reasonably well with the tracer breakthrough predicted from simulation studies.

Methodologies for field applications of microorganisms in ongoing waterfloods were developed as a result of this project. Results from the field pilot showed that microorganisms could be injected into an ongoing waterflood.

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without causing any problems in injectivity. Microorganisms were injected only at the onset of the project, while molasses was injected daily for 2.5 years. Routine injection well backflushing continued to show that the injected microorganisms are thriving in the reservoir. Some of the injected microorganisms were detected at producing wells 32 weeks after injection, which was probably a result of microbial transport through low-volume, high-permeability stringers in the formation.

Microbial treatment did improve oil production rate, and water/oil ratios for producing wells nearest the microbially treated injection wells continue to be more favorable than baseline values. The results from this test are encouraging that microbial-enhanced waterflooding can be applied by independent producers.

INTRODUCTION

A microbial-enhanced waterflood field project sponsored by the U. S. Department of Energy (DOE), Microbial Systems Corp. (MSC), and INJECTECH, Inc., and being conducted in cooperation with the National Institute for Petroleum and Energy Research (NIPER) was initiated in October of 1986. The purpose of the project was to determine the feasibility of injection of a microbial formulation in a mature, ongoing waterflood, and if such an injection could increase oil production rate.

A DOE Fossil Energy report, "Oil Research Program Implementation Plan" has stressed the need for near term oil recovery activities by independent petroleum producers for declining oil fields and stripper wells.[1] According to that study, these activities are particularly important because independent operators produce about 40% of the total oil recovered in the U.S., but cannot conduct needed EOR research. Microbial methods for improving oil recovery are potentially cost-effective and particularly well suited for today's economic climate. The technology is flexible, relatively inexpensive, and can be applied by independent producers. Microbial formulations can be applied in a variety of methods including well simulation treatments, permeability modification treatments, and microbial-enhanced waterflooding. Well stimulation treatments are relatively inexpensive and easy to implement and can provide rapid recovery of nominal investment costs. Microbial-enhanced waterflooding has significant potential for increasing production from aging oil fields that are currently under waterflood. The incremental cost for injecting microbes and nutrient is relatively small in an existing waterflood, which may make this recovery method applicable at low oil prices when more expensive methods are not economically feasible.

The concept of the use of microorganisms to recover oil from depleted petroleum reservoirs is not new. Field and laboratory research has been performed, and patents have been granted for this technology since the late 1940s. Early microbial enhanced oil recovery (MEOR) patents by Zobell,[2] Hitzman,[3] and Updegraff and Wren[4] described the use of microorganisms in reservoirs to produce chemicals that could help to mobilize oil. Several literature reviews on MEOR have been published.[5-8]

Laboratory research has demonstrated that products from microbial fermentation of nutrient can change the chemical and physical properties of oil, selectively plug high-permeability zones to improve sweep efficiency, and increase wellhead pressures in single-well injections. Some microbial species can also significantly improve oil production by helping to remove suspended debris and paraffins from the near wellbore region.

Microorganisms most commonly used for MEOR field processes are species of *Bacillus* and *Clostridium*. These species have a greater potential for survival under petroleum reservoir conditions than other species because they produce spores. Spores are dormant, resistant forms of the cells that can survive more stressful environmental conditions. *Clostridium* species produce surfactants, gases, alcohols and solvents; whereas some *Bacillus* species produce surfactants, acids, and some gases. There are also species of *Bacillus* that produce polymers.

In microbial enhanced waterflood applications, it is important that the microbes be capable of moving through the reservoir matrix and producing chemical products that can mobilize oil. The relative rates of transport of the nutrient and microorganisms will affect the injection strategy and design of the microbial system.

A microbial treatment requires careful design and sound reservoir engineering practice, as does any enhanced oil recovery (EOR) method. The methodology for designing and optimizing MEOR field tests has yet to be established; however, the literature and laboratory experience indicate that certain procedures are necessary to implement a microbial-enhanced waterflood. This particular field experiment was designed to use microorganisms that produced chemicals (surfactants, gases, alcohols, and fatty acids) for improved oil mobilization and had the ability to transport through porous media.

FIELD TEST DESIGN

Since one of the major goals of this project was to develop a technology that could be implemented by independent oil producers, one of the criteria was that the field site chosen for the pilot test should be representative of a mid-continent waterflood operation with stripper wells. The methodology for designing and optimizing MEOR field technology has yet to be established; however, the literature and experience with MEOR processes indicate that certain procedures are necessary to implement a microbial waterflood.

Characterization of the target reservoir is important in designing an MEOR treatment. Microbial treatments can be designed to mitigate channeling and problems with variations in permeability in pay zones. Well log analysis, pressure-transient testing, spinner surveys, and tracer studies can be helpful in identifying channeling or high-permeability streaks in selected reservoirs. Chemical tracer tests can also be useful in identifying directional flow characteristics.

The mineralogy of the rock formation should be characterized by core analysis. The presence of clay minerals in the pores of the rock matrix may increase retention of microbes either by attachment or by filtration. Clay minerals distributed in the pores of the rock matrix may also adsorb surfactants and solvents that are being produced by the microorganisms and decrease the recovery efficiency of the process. In carbonate rocks or sandstone formations containing carbonaceous cementing material, the injection of acid-producing microbes may increase permeability. The effects of rock mineralogy on transport of microbes have not been established but could be important in understanding why plugging has been observed in some field applications.

For microbial treatments, reservoirs should meet some minimum requirements (table 1). Each reservoir has some variation in indigenous microbial populations; therefore, it is necessary to examine the produced water and oil for indigenous microorganisms. These organisms may have an adverse effect on the injected microbial system, or they may be beneficial. The presence of microorganisms in porous media or in injected nutrients will affect the performance of injected microbial systems.[9-10] Some microbes can overgrow and totally eradicate injected microorganisms. Most microbial systems used in prior field tests have been microbes originally isolated from petroleum reservoirs, which can then be adapted to the temperatures, pressures, and salinities normally encountered in a reservoir environment.

Care must be taken when nutrients or sulfate-containing waters are injected in the field to ensure that indigenous sulfate-reducing bacteria (SRB) are either not stimulated or are overgrown by the injected microbes. If a high concentration of sulfate is known to be present in the connate water, then there is a great potential for SRB, and the compatibility of the injected microorganisms must be tested with them. SRB can produce hydrogen sulfide. The deleterious effect of SRB has been well documented by studies of wells that have become soured by microbial action.[11]

As with any EOR application, the production history and characteristics of the reservoir should be studied before microbial treatment (table 1). Fluid samples should be collected and analyzed for trace nutrients such as nitrate and total dissolved solids concentration. Compatibility testing of fluids must be performed with cores under the same conditions of temperature, pressure, and salinity as those in the reservoir of interest. These tests will also yield estimates of oil recovery efficiency. If Berea sandstone cores are used, efforts must be made to match the permeability of the target reservoir. Reservoir cores should be used if at all possible to duplicate the properties of the formation. Chemical tracer tests are needed to identify channeling in the reservoir so that the microbial system can be designed to improve sweep efficiency.

TABLE 1 - Screening criteria for application of MEOR processes in oilfields

Parameter	Recommended range
Salinity	< 10% sodium chloride; total TDS may be higher
Temperature/depth	< 170° F; < 8,000 ft
Trace minerals	< 10-15 ppm of arsenic, mercury, nickel, selenium, copper
Reservoir rock permeability	> 50 millidarcies, unless highly fractured
Indigenous microorganisms	Compatible with injected microorganisms in selected MEOR process
Crude oil type	> 15 °API; not enough information available yet for heavier crude oils
Residual oil saturation	> 25% ; may be some exceptions
Well spacing	< 40 acres; a response can generally be seen sooner on closer well spacing

Very little research has been done on the effects of refeeding or reinjecting microorganisms once they are in place in the reservoir. We have reported that additional nutrient injection after initial injection improves oil recovery.[12] Another area of concern is the quality of the injected nutrients. Grula[13] reported major differences in the composition of molasses, which affects microbial growth and activity; therefore compatibility testing with the nutrient to be used in the field must be done. Molasses with high fiber content can cause plugging of injection wells. Molasses should also be analyzed for any high concentration of trace metals that may be toxic to the injected microbes, and quality control of the molasses must be maintained in the field.

Other criteria for successful MEOR field tests are monitoring and follow-up after results are obtained. Off-pattern wells should be monitored to ensure that migration of the microorganisms does not occur, to protect other nearby formation sites. If any fresh water aquifers are in the area, they should be monitored. No aquifers were near this particular project. Only by consistent monitoring of the microbial process can a credible evaluation be made. A highly desirable characteristic of the microbial system to be injected is that it is distinguishable from indigenous microorganisms present in the reservoir. If this is the case, then the injected microorganisms can also serve as a tracer.

Field Site Selection

After reviewing all laboratory and field data on MEOR processes, the site selection criteria were chosen for this particular field experiment. More than 30 waterflood projects in the Bartlesville, Oklahoma, area were evaluated

as potential candidates for the project based upon the reservoir criteria given in table 1 and other desired characteristics, including:

1. Low brine salinity (i.e., less than 100,000 ppm)
2. Indigenous microbial compatibility with selected microbes from NIPER's microbial culture bank
3. Established oil production decline rate
4. Reasonably uniform water injection rates
5. Favorable well spacing and pattern
6. Availability of reservoir and oil production data

Many waterflood projects in the area met criteria listed in table 1; however, only a few waterfloods had 5-acre spacing instead of 10-acre spacing. We selected those that had the shorter spacing and continued the screening process. When we had narrowed the number of potential candidates to three, brine and oil samples were taken to the laboratory and indigenous microbes were checked for compatibility with selected microbes from NIPER's culture bank. Samples from all three waterfloods showed that the microbes were compatible; however, a B & N Oil Company waterflood in the Mink Unit of Delaware-Childers field used fresh water from the Verdigris River, and this flood was chosen because the selected microorganisms tend to grow better at lower salinity. In many reservoirs with higher salinities, it may be desirable to test the indigenous microorganisms for any properties that may contribute to improved oil mobilization.

Field Data

The Mink Unit site, which includes both the Candy and Sallie Mink leases, selected for the project is located in Delaware-Childers field in Nowata County, Oklahoma (figs. 1 and 2). This particular part of Delaware-Childers field was owned by B & N Oil Company when the project was initiated in 1986. The legal description of the Mink Unit is Section 36, Township 27N, Range 16 E of Nowata County. Delaware-Childers field was discovered in 1906, and by 1911 initial development was essentially complete. The field was produced by primary methods until 1925 when air injection was initiated, and by 1932 air injection was used field-wide. By the 1940s, the field was approaching the economic limit, and waterflooding was begun. By 1945, four small waterfloods were in operation in less prolific areas of the field. During the next 10 years, many waterfloods were iniated throughout the field.

One waterflood, initiated in March 1954, was the Sinclair Oil and Gas Company's Tanner Flood. This project encompassed about 1,200 acres and included the Mink leases, the site of the microbial field experiment. Surface water from the nearby Verdigris River has continued to be the source water for this flood since its initiation. The flood has been in continuous operation, although under various owners, to the present time. Fortunately, more field information exists than would normally be expected for a shallow field which has been producing for over 80 years.

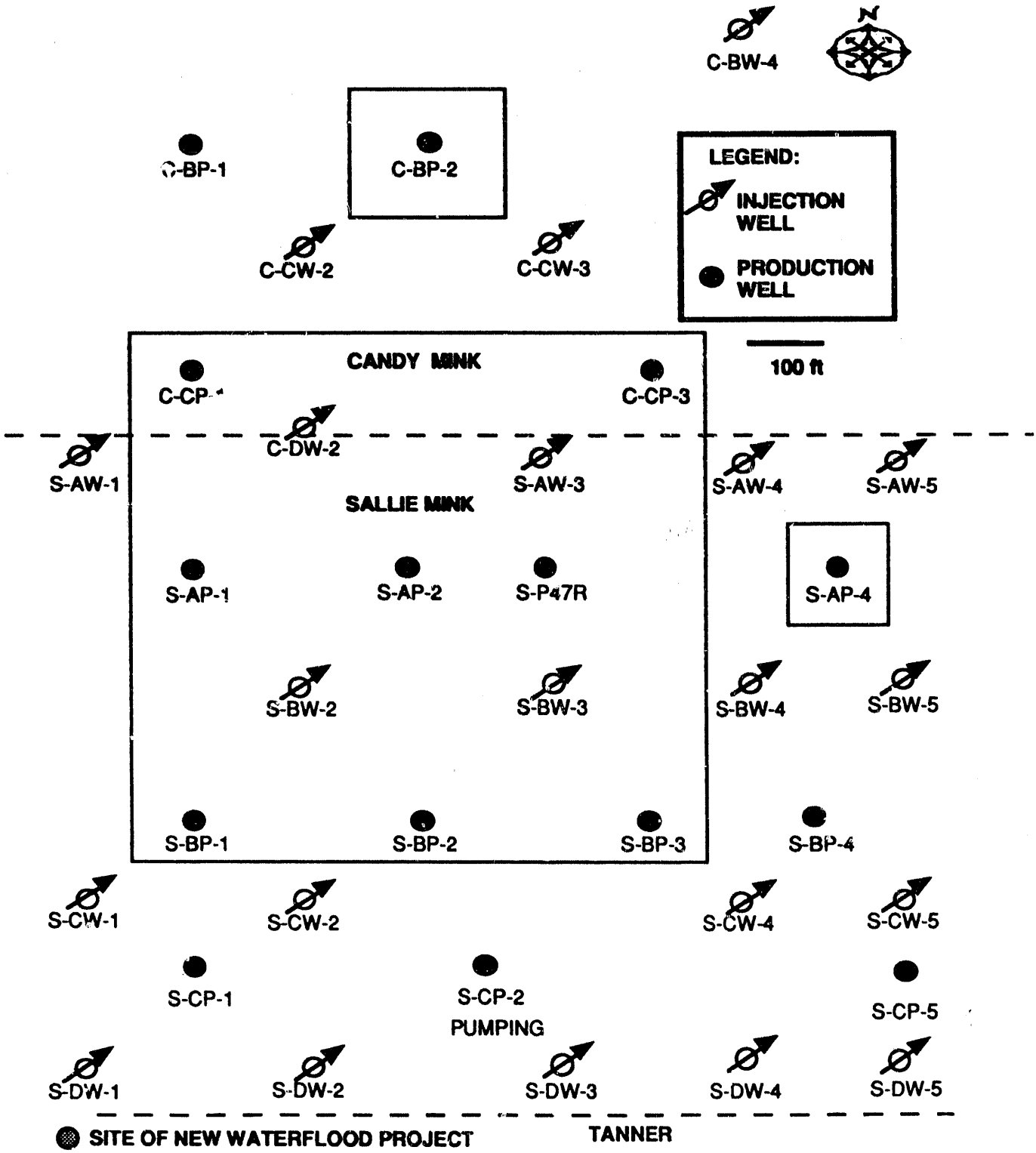

FIGURE 1. – Map of Mink Unit - Delaware-Childers field (S36-T27N-R16E).

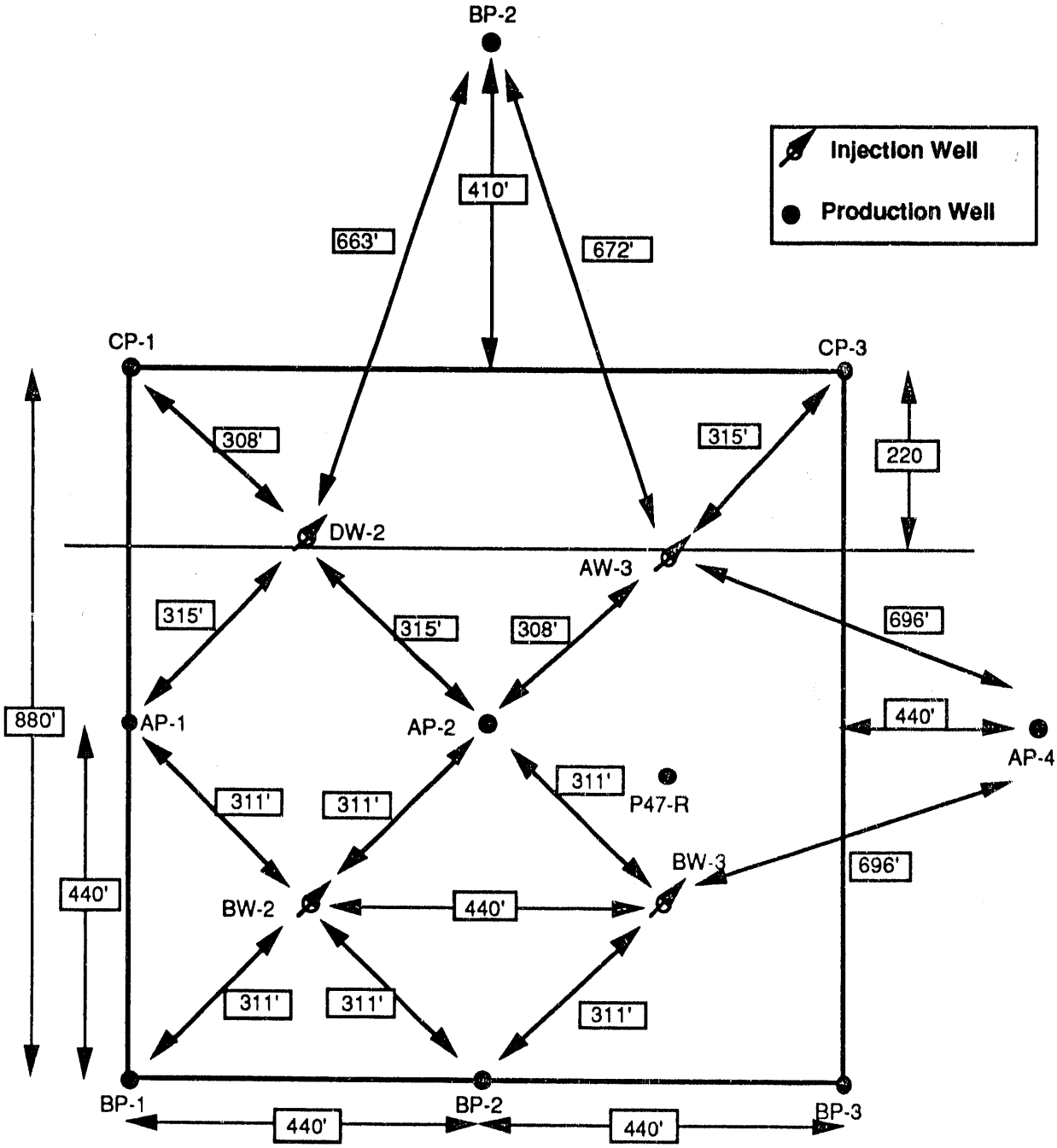

FIGURE 2. – Pilot area of Mink Unit showing well spacing in feet.

This results, in part, from the field size, pioneering secondary recovery efforts, and the close proximity of a petroleum research facility, founded in 1917 as the Bureau of Mines Petroleum Experiment Station, in Bartlesville, Oklahoma.

Reports from the Bureau of Mines provided relevant information about the Mink leases.[14-16] One report[14] shows that most of the Mink leases were developed after primary production was depleted. Another 1955 report[15] lists the production history of the Sinclair-Tanner Flood (annual totals for the 1,200 acres, but not by lease). Drilling/completion reports for about 75 wells on the Mink leases, many of which have since been plugged, were obtained from NIPER's Oklahoma Well Log Library. B & N Oil Company provided core analyses, from cores drilled in 1935 and 1936, from several wells on the Mink leases. Using all available information about the Mink Unit, a net pay isopach map was constructed (fig. 3), an estimate of initial oil saturation was made, and the production history for the unit was determined from initial development to 1952. Actual lease production records from 1953 to the present have been available for continuing this production history.

The Mink leases were determined to have an average porosity of 20%, an initial average oil saturation at the start of the project of 32.6%, and a combined net pay bulk volume of 2,900 acre-feet. The estimated cumulative oil production from the two leases has been 341,217 bbl through 1986. The project area has a surface area of 17.78 acres and a net pay bulk volume of 516 acre-feet.

In 1988, as a result of the sale of this oilfield, cores were drilled on the Mink Unit and Brown leases of this field, and the resulting information was provided by the new owners to NIPER. Figures 4 through 6 show the oil saturation, brine permeability, and porosity reported for cores from the well drilled in the Mink Unit. The average oil saturation of the Mink Unit core was 31.9%, which corresponded to our predictions that had been based on earlier core reports and production history. The average permeability is 90 millidarcies, which was higher than some of the earlier core analyses, and the average porosity is 19.1%.

With an estimated irreducible oil saturation of 25%, the recoverable oil within the leases by waterflooding was 76.3 bbl/acre-foot or about 40,000 bbl in the pilot area, at the initiation of the microbial field project. The Mink Unit covers a 160-acre area of which 110 acres are productive and contain 21 injection wells and 15 production wells drilled on 5-acre spacing (fig. 1). Only one of the producing wells is being pumped. Well completions are open-hole. The average reservoir properties are listed in table 2.

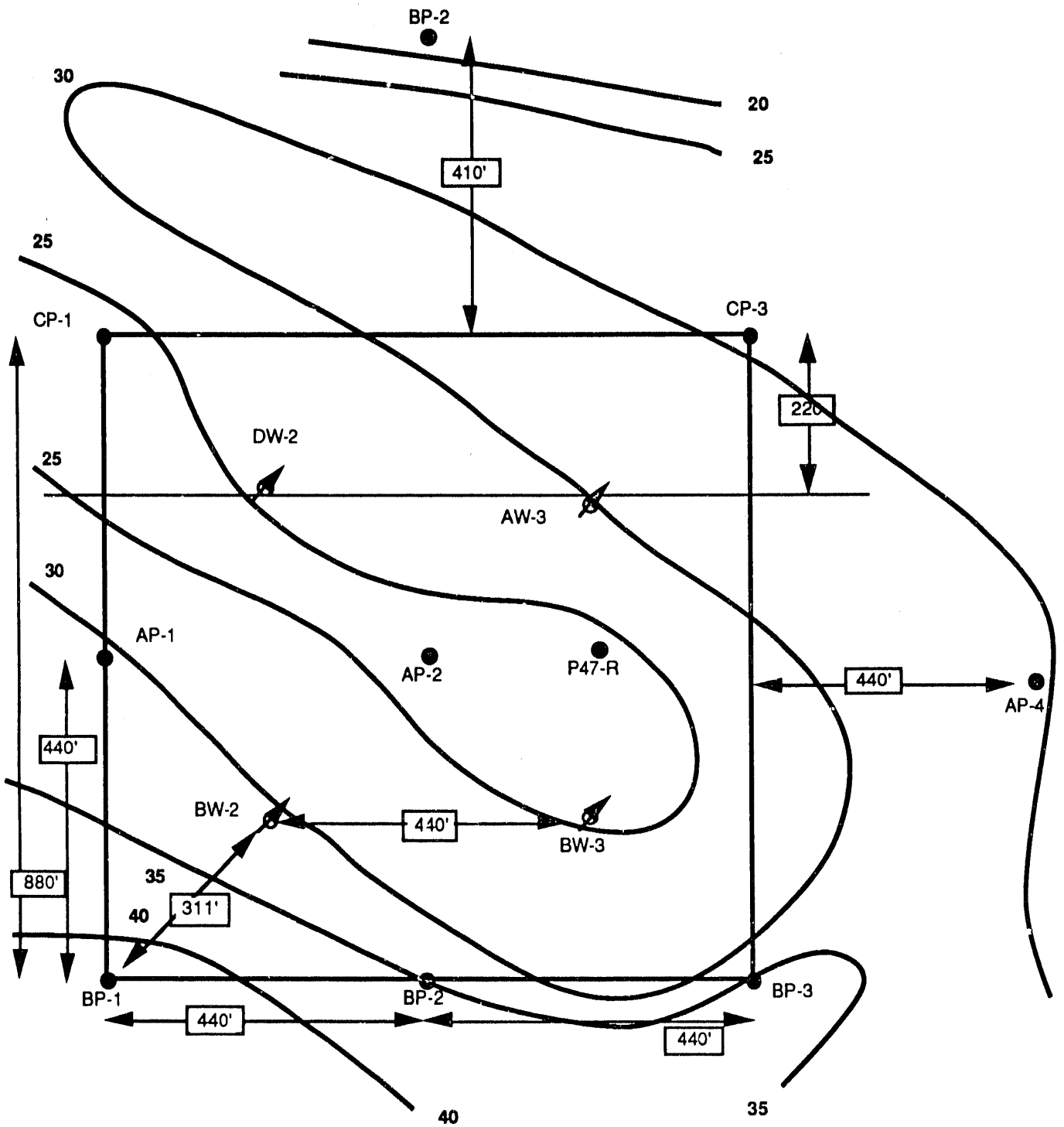

FIGURE 3. - Net pay isopach of Mink pilot area - Delaware-Childers field.

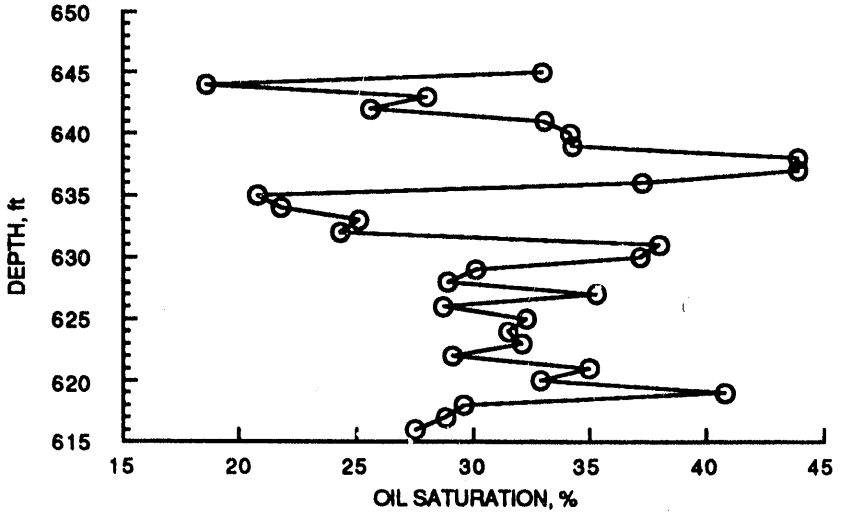

FIGURE 4. – Oil saturation from Mink Unit core data.

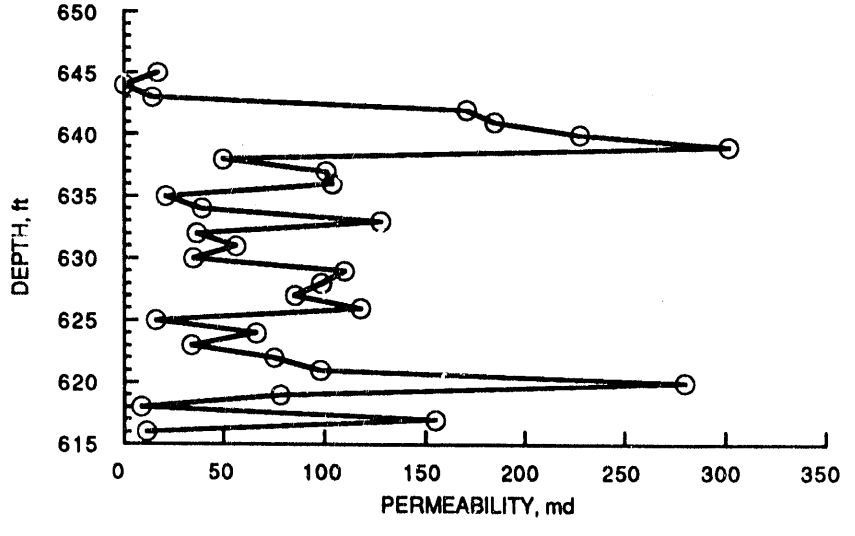

FIGURE 5. – Brine permeability from Mink Unit core data.

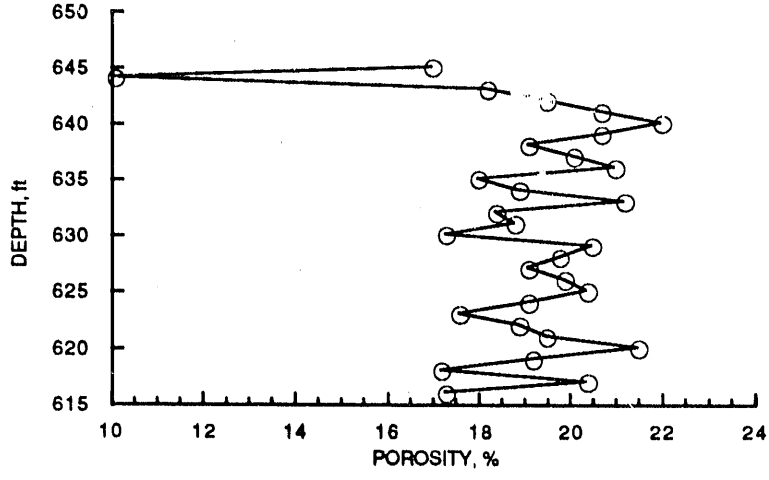

FIGURE 6. – Porosity from Mink Unit core data.

TABLE 2. – Reservoir properties for Mink Unit

Formation	Bartlesville Sandstone
Depth, ft	600
Average net pay thickness, ft	30
Average permeability, md	90
Porosity, %	20
Average formation temperature, ° F (range 65 to 80)	75
Number of injection wells	21
Number of production wells	15
Average water injection rate, bbl/day	40/well
Average injection pressure, psi	530
Average oil production, bbl/day	6.4
Oil gravity, ° API	34
Oil viscosity, cP @ 77° F	7
Total dissolved solids of injection water, %	0.03
Average total dissolved solids of produced water, %	0.5
Average oil saturation, % (at start of project)	30

Reservoir Characterization

The Mink Unit contains the Sallie and Candy Mink leases. Net pay thickness in Mink Unit decreases from approximately 40 ft to less than 10 ft in a northeasterly direction from the southwest corner of the unit. The original oil in place is estimated from historical oil production records to be 1,666,000 bbl of which 341,000 bbl had been produced as of the end of 1986. The remaining 1,325,000 bbl of oil in place in the 2,900 acre-foot of net pay yield an average oil saturation of approximately 460 bbl/acre-foot (30%). The annual oil production rate from the Mink Unit has remained relatively constant since 1982 (fig. 7).

The pilot site for the project was four adjacent inverted 5-spot patterns within the Mink Unit (fig. 2). The pilot site covers an area of 17.8 acres and a net pay volume of 516 acre-foot. The pilot area has four injection and eight production wells. In addition, two off-pattern wells (C-BP-2 and S-AP-4) were monitored as part of this project. Before beginning the microbial waterflood, all possible efforts were made to ensure that no changes in operating conditions or procedures occurred during the pilot test. The normal procedure of backflushing all injection wells each week was continued.

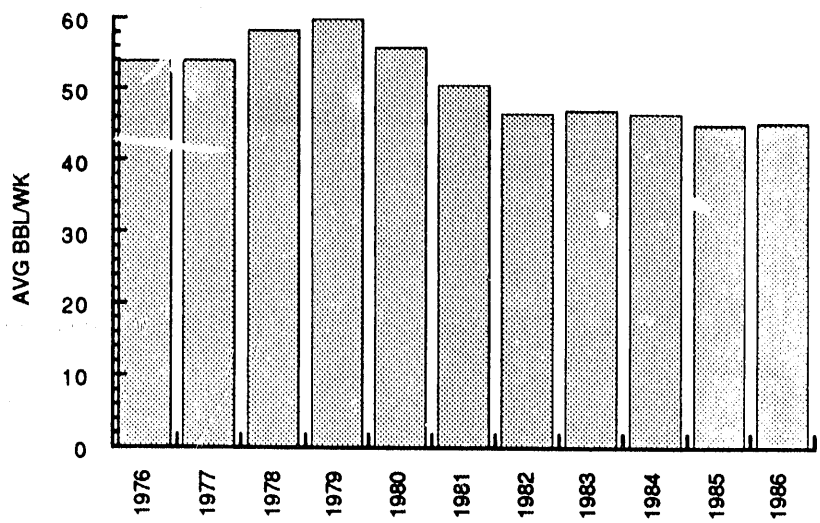

FIGURE 7. – Mink Unit historical oil production.

FIELD OPERATIONS

Baseline Data

The sequence of events in this project is briefly outlined in figure 8. Field sampling began in November 1986, and continued to March 17, 1987. The data from these baseline studies showed that the total dissolved solids (TDS), pH, oil viscosities, and microbial counts were consistent during this period. Field data, including injection pressures and volumes, oil production rate, and water/oil ratios all remained fairly constant during the baseline monitoring period. Since this waterflood had been ongoing since 1953, it is not surprising that the data from the individual wells and the field remain consistent. The same source of injection water, the Verdigris River, has been used since the initiation of the waterflood. The TDS of the produced water from each production well and each injection well remained constant to within ± 0.01%. The pH of the samples remained between 6.4 and 7.0, and trace

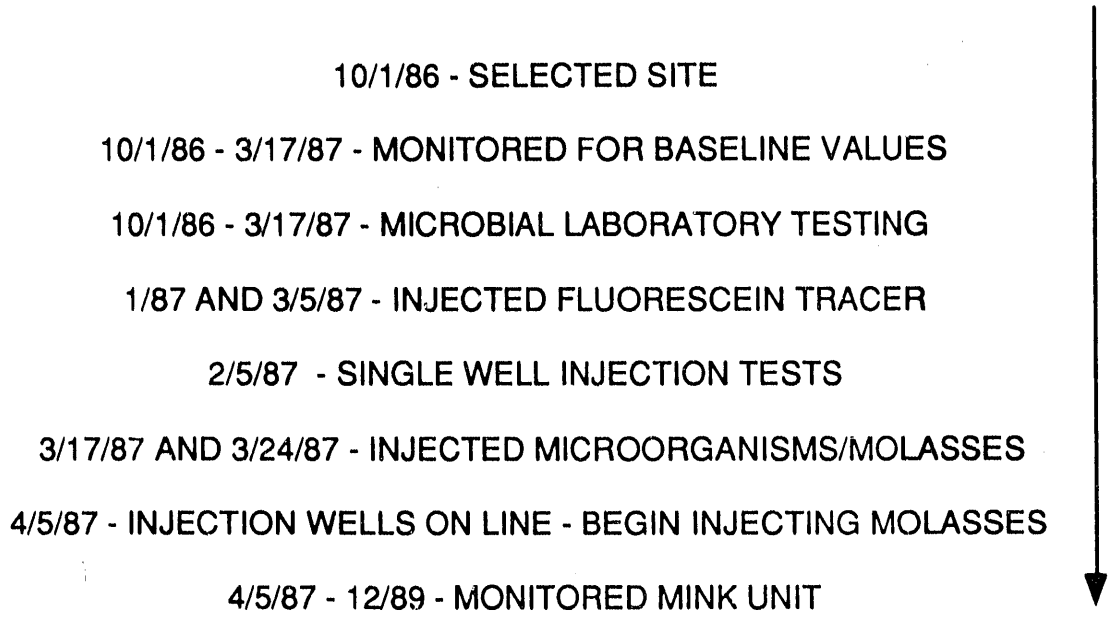

FIGURE 8. – Sequence of events in the Mink Unit project.

mineral and ion analyses indicated no marked changes in concentrations of the following ions: sodium, calcium, magnesium, strontium, barium, carbonate, hydroxide, and phosphate. The microbial counts from the producing wells were consistent throughout the monitoring period. The counts were very low, and ranged from 0-100 cells/mL in the producing wells. Sulfate-reducing bacteria were consistently present at low levels in the tank battery water and intermittently present in the plant injection water. There were sporadic occurrences of sulfate-reducing bacteria in the produced waters. The tabulated data from the baseline studies are compared to the results after microbial treatment in the Results section of this report.

Field data were also monitored during the baseline period. Individual producing well water/oil ratios (WOR), injection plant and injection well pressures, and oil production rates were tabulated for the baseline monitoring (table 3).

TABLE 3. – Baseline field data for the Mink Unit. Oct 28, 1986 - March 17, 1987

Week	Plant inj. pressure, psia	Well inj. pressure, psia	Oil production, bbl/wk
0	–	–	41
1	–	–	42
2	–	–	39
3	–	–	45
4	540	530	39
5	–	–	38
6	–	–	38
7	–	–	42
8	536	526	39
9	–	–	42
10	–	–	40
11	–	–	42
12	538	525	44
13	–	–	48
14	–	–	45
15	–	–	47
16	551	534	37
17	–	–	44

During the baseline period, two single-well injectivity/survivability tests were conducted with two microbial formulations. These single-well tests provided valuable information about the ability of the microbial formulation to survive under actual reservoir conditions and showed that no plugging or increase in injection pressures occurred after microorganisms and molasses were injected.

Chemical Tracer Study

A chemical tracer study was implemented during this baseline period (December, 1986) to determine: (1) the flow patterns of the injected fluids in the Mink Unit; (2) if any gross channeling existed; and (3) if there was communication among all producing wells and the four treated injectors.

Fluorescein was tested in the laboratory with the microbial formulation and reservoir fluids and found to be compatible under reservoir conditions; therefore, it was chosen as the chemical tracer. On Jan. 13, 1987, 27 bbl of a fluorescein solution at a concentration of 714 ppm was injected into wells S-BW-2 and S-BW-3, respectively; and on March 5, 1987, well C-DW-2 was injected with 5.2 bbl of 302 ppm fluorescein, and S-AW-3 was injected with 5.2 bbl of 210 ppm fluorescein solution. Sampling of each producing well was conducted daily for the first 5 days after tracer injection, then biweekly sampling continued until no fluorescein was detected in the production wells.

Samples were protected from light and transported to NIPER where the fluorescein concentration was determined using a spectrophotometric method.

The fluorescein concentration curve was plotted against time for each producing well, and these results showed that there was communication among all of the wells since every well showed some fluorescein response. There did not appear to be gross channeling because the response persisted for a reasonable period of time (2 months) before leveling off to below the detection limit of 100 ppb. The area under each curve was integrated, and a value was obtained. This value was divided by the average number of barrels of produced fluid for that well, and the wells were ranked accordingly (table 4). The tracer studies seemed to indicate a northeasterly flow pattern (fig. 1) because wells C-CP-1 and C-CP-3 and S-AP-4 received fluorescein in greater amounts and more quickly than the other wells. The middle well, S-AP-2, received the highest amount of fluorescein, which was expected since this well is affected by all four injection wells.

TABLE 4. – Fluorescein response from Mink Unit tracer injection

Well	Area of integrated curve ppb · days	Avg produced fluid bbl/d	Ratio*
C-BP-2	2,900	169	17.2
S-BP-2	2,575	115	22.4
S-BP-1	4,250	168	25.3
S-P47R	6,087	193	31.5
S-BP-3	1,225	36	34.0
C-CP-1	2,792	43	64.9
S-AP-1	8,350	76	109.9
C-CP-3	6,525	56	116.5
S-AP-4	5,145	43	119.7
S-AP-2	3,852	27	142.7

*Area of integrated curve/avg produced fluid

Fluorescein was again observed about 1.8 years after injection (table 5 and fig. 9). The persistence of this response seems to indicate that the tracer had just transported through the matrix of the formation, and that the earlier response of tracer was due to low-volume, high-permeability streaks in the formation. Of the wells sampled, the fluorescein appearance again indicate a northeasterly flow pattern, because C-CP-1 and C-CP-3 showed fluorescein in

high amounts, as did S-AP-2, the middle well. Well S-AP-1 was sampled only twice, and probably is not representative of its true tracer appearance.

TABLE 5. – Tracer appearance in the Mink Unit during 1988-89 sampling (All values are in ppb)

Weeks Post Inj.	S-P47R	S-AP-2	C-CP-1	C-CP-3	S-BP-1	S-BP-2	S-AP-1
90	0	-[1]	0	0	0	-	-
94	100	-	0	0	0	-	-
95	100	-	100	180	150	-	-
99	0	250	100	100	0	-	-
101	280	-	100	-	0	-	-
102	100	150	-	150	325	-	-
103	100	0	100	500	120	-	-
104	100	0	100	-	225	-	-
105	100	0	-	100	100	-	-
106	100	0	0	100	100	0	-
107	0	0	0	100	100	375	-
108	100	100	175	100	-	200	100
109	0	0	0	225	0	0	-
110	0	275	0	100	200	0	150
111	0	0	0	50	50	-	-
112	0	-	-	-	-	-	-
113	-	0	0	100	0	-	-
114	0	275	100	0	500	125	-
115	100	-	-	-	-	-	-
116	-	0	0	300	0	100	-
117	0	-	0	130	-	-	-
119	100	-	-	-	-	-	-

[1] Either no sample was available, or sample was too turbid to measure.

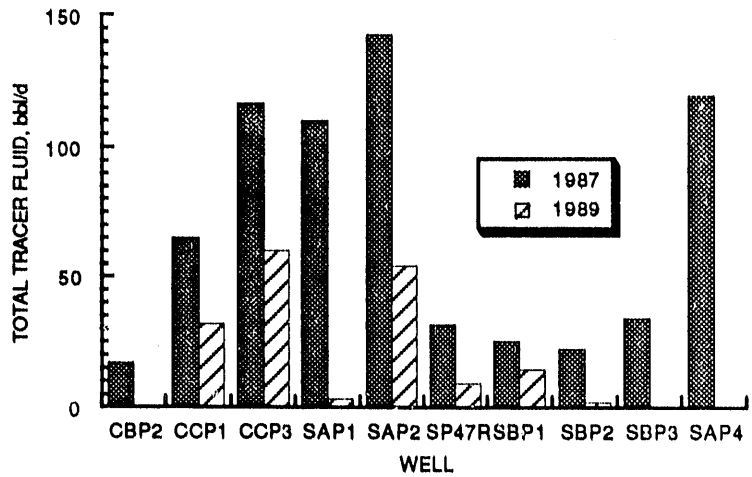

FIGURE 9. – Tracer appearance in Mink Unit producers in 1987 and 1989.

The tracer results obtained are consistent with reports from a micellar-polymer pilot conducted on a nearby lease.[17] Chemical tracers (ammonium thiocyanate and isopropyl alcohol) were used in that study; however, no breakthrough of the tracers was ever detected in the produced water. Later, after injection of the micellar-polymer solutions, polymer was detected in off-pattern wells to the northeast of the pilot site, which indicated a directional permeability flow from the southwest to the northeast. The Bartlesville sandstone is a Cherokee Group, Desmoinesian Series, Middle Pennsylvanian System fluvial-dominated deltaic deposit.[18] Ultimate recovery of oil from reservoirs in the Cherokee Group is affected by facies, bedding boundary and other permeability barriers, and diagenetic changes. Because of these factors, permeability trends such as those observed in Delaware-Childers field, would be fairly common.

LABORATORY SUPPORT

Laboratory Design of the Microbial System

In addition to the field pilot test (SGP-13), a laboratory MEOR research program (BE3) sponsored by the U.S. DOE has been in progress at NIPER for the past several years. Both projects are part of NIPER's integrated program to develop and apply MEOR technology to increase our Nation's reserves of crude oil. A key result from these programs is the development of a microbial culture bank consisting of different types of microorganisms that can improve oil mobilization under a wide variety of conditions.

Based on results from the above programs, a set of conditions was established for the microbial formulation for the field pilot. The optimized microbial formulation for this particular field application must have several characteristics:

1. Be able to survive and "out-compete" indigenous microorganisms in the reservoir
2. Able to utilize inexpensive nutrients for growth and metabolism
3. Have the ability to thrive and produce the desired metabolites for improved oil mobilization under reservoir conditions of temperature, pressure, and salinity
4. Possess the ability to transport through the formation without increasing the injection pressure
5. Pose no threat to humans or wildlife, i.e., nonpathogenic
6. Not produce any harmful metabolite, such as hydrogen sulfide
7. Able to be transported to the field safely and in a cost-effective manner

Porous Media Studies

Studies in porous media using microorganisms have been conducted by NIPER researchers since 1984. It has been well-documented that microorganisms can increase oil production from Berea sandstone cores.[6] Different microorganisms can accomplish this by basically two mechanisms: (1) They can produce chemicals that improve the microscopic oil displacement efficiency; or (2) They can produce polymers and/or biomass that can improve sweep efficiency.

Various microbial formulations isolated by NIPER and INJECTECH, Inc. that satisfied the above criteria were grown with reservoir fluids from the Mink Unit in order to assess the compatibility of the microbial species. Those that appeared to be compatible under reservoir conditions were then tested in Berea sandstone cores to determine their ability to recover crude oil. Data from several microbial corefloods are shown in table 6. Several microbial corefloods were conducted with the individual components of NIPER Bac 1: NIPER 1, NIPER 2, NIPER 3, and NIPER 4. However, these corefloods were conducted before the initiation of this particular field project; thus, the molasses used was different from the molasses selected for the field test. The results from the earlier corefloods did show that the microorganisms could improve oil recovery in Berea sandstone cores. NIPER 3, the *Clostridium* species, showed excellent recovery efficiencies (E_r) in earlier corefloods. NIPER 1 did not have as great an E_r as NIPER 2, even though they are both *Bacillus* species that produce surfactants. However, when assayed in several corefloods, NIPER 2 did not survive as consistently as the surfactant producer that had a recovery efficiency of 25%; it was frequently overgrown by indigenous microorganisms. Since survival, as well as oil recovery, were critical to the success of the project, both microbes NIPER 1 and NIPER 2 were selected for the field test. NIPER 4, the last member of the microbial consortium, is a copious gas producer and very motile. It was selected because of its ability to transport through porous media and because it was very compatible with the other

members of NIPER Bac 1. All four of the above bacteria had been used in coreflooding experiments for several years, and no adverse effects such as plugging had ever been observed.

The other coreflood experiments used various microbial formulations from INJECTECH, Inc. These were primarily *Clostridium* and *Bacillus* species. In the molasses to be used in the field, these microorganisms did not thrive well. INJECTECH microbes were originally isolated for higher salt concentrations; since we had selected a freshwater flood, this may be a reason why they did not grow well. It was also suggested that perhaps the Mink Unit injection water used for the tests contained a higher concentration of oxygen (> 5 ppm) than the strictly anaerobic Clostridia could tolerate. It was then decided to use NIPER Bac 1 for the field test. The composition of NIPER Bac 1 is given in table 7.

Two coreflood tests were conducted using core samples from Delaware-Childers field with NIPER Bac 1 to determine its oil recovery efficiency. Although these cores had not been preserved, they were somewhat representative of the lithology of the formation.

TABLE 6. – Microbial coreflooding experiments for selection of microbial formulation

Core Desig.	k,[1] md	Microbes	Amt Inj. PV	PV,[2] cm^3	S_{orwf},[3] %PV	S_{orcf},[4] %PV	ΔS_{or}[5], %PV	E_r[6], %
M94[7]	332	NIPER 1	0.5	54.0	34.8	27.6	7.2	20.7
M68[7]	180	NIPER 2	0.5	46.0	25.0	11.7	13.3	53.2
M69[7]	134	NIPER 3	0.5	37.8	22.8	8.7	14.1	61.8
M77[7]	344	NIPER 3	0.5	56.7	34.0	18.3	15.7	46.2
M44[7]	304	NIPER 4	0.5	59.6	29.7	19.3	10.4	35.0
M61[7]	293	NIPER 4	0.5	57.5	27.5	19.1	8.4	30.6
B12[7]	271	NIPER BAC 1	0.2	57.7	34.7	30.4	4.3	12.4
B16A[7]	362	NIPER BAC 1	0.2	140.0	35.7	31.9	3.8	10.6
HS2	353	NIPER BAC 1	0.4	568.2	37.8	29.5	8.3	22.0
MSC 10	214	INJECTECH1	0.5	55.7	30.2	28.8	1.4	4.6
MSC 14	180	INJECTECH2	0.5	54.6	38.5	36.6	1.9	4.9
MSC 15	181	INJECTECH3	0.5	54.8	33.9	31.7	2.2	6.5
MSC 21	99	INJECTECH4	0.5	45.9	36.4	35.1	1.3	3.6
MSC 22	133	NIPER BAC 1	0.5	51.0	36.9	30.0	6.9	18.7
MSC 23	52[8]	NIPER BAC 1	0.5	43.8	31.5	22.6	8.9	28.3
MSC 24	162	NIPER BAC 1	0.5	53.0	38.6	34.2	4.4	11.4
MSC 25	35[8]	NIPER BAC 1	0.5	43.8	27.4	23.0	4.4	16.1

[1] Absolute permeability to brine, millidarcies.
[2] Pore volume of core.
[3] Residual oil saturation in core after waterflooding.
[4] Residual oil saturation in core after microbial treatment.
[5] $S_{orwf} - S_{orcf}$.
[6] $(S_{orwf} - S_{orcf})/S_{orwf} \times 100\%$.
[7] These corefloods were flooded with molasses from Pacific Molasses Co.
[8] Indicates core from Delaware-Childers field.

TABLE 7. – Composition of NIPER Bac 1

NIPER No.	Genus designation	Oxygen requirement	Microbial products
NIPER 1	*Bacillus licheniformis*	Facultative[1]	Surfactant, acids
NIPER 2	*Bacillus* species	Facultative[1]	Surfactant, acids
NIPER 3	*Clostridium* species	Anaerobic[2]	Gases, alcohols, acids, surfactant
NIPER 4	*Shewanella*[3] species	Facultative[1]	Gases, acids

[1] Can grow with or without oxygen.
[2] Can only grow without oxygen.
[3] Tentatively identified, and not confirmed.

The coreflooding procedure was as follows: Core MSC 23 was evacuated and saturated with brine from the Mink Unit. Core MSC 25 used pieces of core epoxied together and placed into a Hassler coreholder. An absolute permeability to brine was determined to be 34.5 md. The cores were saturated with crude oil from the Mink Unit which had a viscosity of 7 centipoises, to an initial oil saturation of 67.7 and 67.6%. respectively, and then flooded with brine to a residual oil saturation of 31.5 and 27.4%. Into each core, 0.2 pore volume (PV) of NIPER Bac 1 and 0.3 PV of molasses was injected and allowed to incubate at 25° C for 3 days. The cores were then waterflooded at 1 ft/d. A graph of the residual oil saturation from coreflood MSC 23 versus injected pore volumes of brine after microbial treatment is shown in figure 10. The microbial treatment recovered 28% of the residual oil remaining in the core after waterflooding. Most of the oil was recovered before the first pore volume of fluid. This was consistent with our observations in Berea sandstone cores.[12] We obtained a recovery efficiency (E_T) of 16.1% for MSC 25, which was acceptable, particularly since this core was pieced together and showed a permeability of 34.5 md. The residual oil saturation values (31.5 and 27.4%) obtained for these cores corresponded reasonably well with the estimated oil saturation value of 30% for the field. The oil saturation range from the 1988 core analysis of the Mink Unit was 18.6 to 43.9%; although the majority of the core plugs showed saturations from 30-35% (figure 4). The permeability value of 52 md was within the range for the Mink Unit reported in core analyses; figure 5 shows that most of the values were in the range of 30-100 md. MSC 25 had a permeability of 35 md, which may have been a little lower due to the piecing together of core plugs.

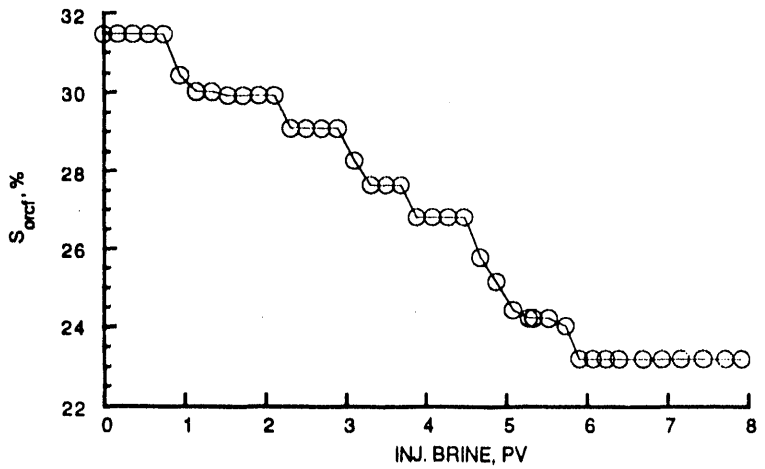

FIGURE 10. – Residual oil saturation after microbial treatment of an unpreserved field core.

After the initial microbial coreflood experiment with the field core, core MSC 23 was flooded with several pore volumes of brine from the Mink Unit, and then re-injected with a consortium of microorganisms isolated from Mink Unit produced water. These microorganisms were characterized as being indigenous to the Mink Unit, and it was important to determine if our microbial formulation, NIPER Bac 1, could survive and out-compete these microbes in porous media. The indigenous microbial consortium was grown in 4% v/v molasses, 0.2 PV was injected into the field core, and the core was shut-in for 3 days. A slug of NIPER Bac 1 (0.2 PV) in molasses was injected, and the core was shut-in for another 3 days. The core was waterflooded, and the effluent was counted for microorganisms and microscopically observed. The results showed that NIPER Bac 1 continued to thrive in the presence of these indigenous microorganisms, even though the indigenous microbes also grew well inside the core.

Experiments with a simulated porous medium, etched-glass micromodels, were conducted to help optimize the microbial formulation by visually observing oil mobilization by NIPER Bac 1. Correlations between microbial coreflooding results and oil mobilization in these micromodels have been reported.[18] A micromodel was saturated with brine from the Mink Unit tank battery and flooded with crude oil from the Mink leases. The micromodel was then flooded with plant injection water until no more oil movement was observed (residual oil saturation). NIPER Bac 1 was injected, and the micromodel shut-in at room temperature for 3 days. The micromodel was then waterflooded and video-taped using a video-enhanced microscopy apparatus. It was observed that there were gas bubbles produced during incubation of the micromodel, as well as some emulsification of the crude oil. When the micromodel was waterflooded after microbial treatment, approximately 60% of the oil initially present was mobilized.

Prior work at NIPER has identified the mechanisms of oil mobilization by certain microbial formulations. Mechanisms that have been shown to be important include wettability alteration, emulsification, solubilization, and alteration in interfacial forces. Recent experiments at NIPER have demonstrated that oil mobilization by microbial formulations are not merely the result of the effects of the metabolic products from the in situ fermentation of nutrient.[19-20] The localized transient concentrations of metabolic products produced by the cells at the oil-water interface are probably a major key to oil mobilization. Although more work is needed to investigate this aspect, it is believed that the presence of the cells in situ, as well as the combined production of surfactants, gases and solvents, are required for successful microbial oil mobilization.

Toxicity and Mutagenicity Testing

Tests for microbial pathogenicity of NIPER Bac 1 were conducted at Oklahoma State University. Mice were used to test for pathogenicity of the microbial formulation, each individual microbial species, and the molasses. Solutions containing the microbes and/or molasses were given both by oral ingestion and intraperitoneal (IP) injection.[21] For IP injection, a 1-mL aliquot was used. For oral ingestion, the mice were deprived of water for 48 hours and then given access to the solution for a period of approximately 20 minutes. All mice were observed more than 2 months, and no ill effects from ingestion or injection were ever noted. Pathogens of the genera *Clostridium* and *Bacillus* produce toxins that have incubation periods of short duration.

The carcinogenic (cancer-causing) potential of many chemical compounds to which humans are exposed in their environment is highly correlated with the ability of the compounds to induce mutation. A culture of NIPER Bac 1 grown in 4% v/v molasses was filtered to remove the microbial cells through a 0.45-micron syringe. The resulting microbial product solution was submitted to the Ames test for detecting mutagenic chemicals.[22] The Ames test, which is used as a prescreen for carcinogenic substances, relies on a series of nutritional mutants of *Salmonella typhimurium*. The assay disk method of the Ames test was used by first placing a thin layer of agar inoculated with *Salmonella typhimurium* over a base agar plate. The *Salmonella typhimurium* strain requires the amino acid histidine. The medium contained only a very small amount of histidine, which should have allowed a few cell divisions to occur. Filter paper disks saturated with NIPER Bac 1 products, sterile water, and nitrobenzene were placed equidistant on the soft agar. The water and nitrobenzene were used as positive and negative controls for mutagenicity. A duplicate plate was prepared, and both plates were incubated at 37° C for 48 hours. Scattered colonies appeared on the surface of both plates. A positive result would be indicated by a relatively high concentration of colonies surrounding the disk. No increase in colony formation was observed surrounding the paper disks with NIPER Bac 1 or molasses.

Environmental Contingency Plan

All reasonable precautions were taken to ensure that there were no adverse effects on the environment from conducting this microbial-enhanced waterflood field experiment. All injected materials were handled in closed containers. A contingency spill plan was formulated by NIPER in the unlikely event of an unwanted microbial contamination. Biocide was stored near the site to be used for killing the injected microorganisms. One of the more beneficial aspects of MEOR technology is that if the microorganisms are not fed, they will disintegrate and die. Molasses injection at this project site was discontinued September 23, 1989, and biocide was injected into the four injection wells on the Mink Unit that had been microbially treated. No large population of injected microorganisms was ever consistently observed at the Mink Unit tank battery. Although some of the injected microorganisms were observed sporadically at Mink Unit production wells, it is believed that the microorganisms never reached the production wells in sufficiently high population that would impact the environment.

Single-Well Injection Tests

In February, 1987, during the baseline period, two single-well injection tests using off-pattern wells were performed to establish certain parameters before injection of the microbial formulation at the Mink Unit site. The goals of these tests were: (1) to determine if any reduced injectivity resulted from microbial plugging; (2) to ensure that the microbial consortium grew well under reservoir conditions; and (3) to develop adequate sampling procedures for the project. An off-pattern injection well was injected with 26 gallons of NIPER Bac 1 (approximately 1.0×10^8 cells/mL) in molasses (4% v/v) and shut-in for 12 days. The well was backflushed, and samples were collected every 10 to 15 minutes until NIPER Bac 1 and molasses were detected. Samples were inoculated into sulfate-reducing bacterial growth media to determine if sulfate-reducing bacteria were stimulated by introduction of molasses and other microorganisms. Gas samples were assayed for hydrogen sulfide. Samples were collected, and pressures were measured at another injection well nearby to serve as controls.

The results from these tests showed the following: (1) the injection rates and pressures of the microbially treated well after the shut-in period were normal and comparable to the control well, indicating no plugging had occurred; (2) all of the injected bacteria in NIPER Bac 1 were detected in high numbers in the backflush samples, indicating that the microbes were still growing and metabolically active after 12 days of incubation under reservoir conditions. No hydrogen sulfide (limit of detection was 5 ppm) was found in any of the gas samples, and no sulfate-reducing bacteria were detected from any of the microbially-treated well samples, and (3) our sampling procedures appeared adequate to measure gas and fluid from the injection and production wells.

Injection of NIPER Bac 1 and Molasses

Twenty-six gallons (0.65 bbl) of the microbial formulation, NIPER Bac 1, were injected into each of the four targeted injection wells, C-DW-2, S-BW-2, S-AW-3, and S-BW-3 (fig. 2). Wells C-DW-2 and S-BW-2 were treated on March 19, 1987, and wells S-AW-3 and S-BW-3, on March 23, 1987. Twenty gallons of molasses diluted to a concentration of approximately 4% was injected into each well periodically during the microbial injection. The molasses and microorganisms were injected by means of a header bypass system (fig. 11). The header bypass allowed injection water from the injection well to flow into the header, mix with the microorganisms or the molasses, and then flow back into the injection well. This system allowed adequate mixing and dilution of the concentrated molasses, and since the ongoing waterflood was used as the injection driving mechanism, no adverse shearing effects on the microorganisms occurred. The four treated injection wells were shut-in until April 3, 1987, when water injection was resumed. The other 17 injection wells in the Mink Unit were still in operation during the shut-in period. After water injection was resumed, the injection wells were backflushed to determine if microbial activity could be observed. Samples of water backflushed from the treated injection wells foamed when shaken, indicating surfactant production and that the microbial populations were viable. Subsequently, the four injection wells received the equivalent of 2 gallons of undiluted molasses per well per day until September 21, 1989.

FIGURE 11. – Injection header system used for the Mink Unit injection fluids.

PROJECT EVALUATION

Sampling of the producing wells was conducted on a weekly basis. Samples were collected from a flowing stream in sterile 4-oz flint glass bottles. Each bottle was filled completely and tightly capped. The samples were taken to the laboratories and processed immediately. The parameters monitored after microbial injection are given in table 8. The results for each parameter given are averages of samples taken during the period of April 3, 1987 through June 1, 1989.

Microbial Counts and Molasses Concentration

The microbial counts have always been surprisingly low for this particular field, particularly since the source water is river water. Oil field brine analyses were conducted of the reservoir fluids, and the results are presented in table 9. The micronutrients available for microbial growth (phosphate, nitrate, and sulfate) were very low in concentration. The baseline counts averaged less than 100 cells/mL for most wells (table 10). During the test, the microbial populations fluctuated greatly, yet on an average, were about 100 times higher after the microbial injection. Some of the injected bacteria from the NIPER Bac 1 consortium were observed at 32 weeks post-injection. Since that time, the same bacterium has been observed periodically from several producing wells. Interestingly, the observation of NIPER 1, which formed distinctive volcano-like colonies, in the producing wells tends to correlate with the observation of fluorescein (figs. 12 and 13). However, a large population of NIPER 1 has never been consistently observed in any producing well or the tank battery.

TABLE 8. – Monitored parameters for field test

Parameter	Sampling frequency	Location sampled
Total dissolved solids	Weekly	Each producer, plant water
pH	Weekly	Each producer, plant water
Surface tension	Weekly	Each producer, plant water
Oil viscosity	Biweekly	Each producer
Interfacial tensions	Biweekly	Each producer
Microorganisms	Weekly	Each producer, plant water
Molasses conc.	Weekly	Each producer, plant water
Microorganisms	Monthly	Each treated injector
Surface tension	Monthly	Each treated injector
pH	Monthly	Each treated injector
Gas analyses	Every few months	Randomly selected producers

TABLE 9. – Trace mineral analyses from Mink Unit water samples taken October 28, 1986

	Injection plant, mg/L	Tank battery, mg/L	Well S-AP-2, mg/L
		CATION	
Sodium	12	1,183	3,176
Calcium	34	156	330
Magnesium	53	289	217
Strontium	0.4	30	20
Barium	0.2	134	144
Potassium	4.5	8.7	14
Iron	0.7	5	2
Copper	<0.1	<0.5	<0.5
Zinc	<0.1	<0.5	<0.5
Nickel	<0.1	<0.1	<0.5
		ANION	
Chloride	17	2,037	5,294
Sulfate	15	12	12
Bicarbonate	135	1,450	1,800
Carbonate	0	0	0
Hydroxide	0	0	0
Phosphate	<0.5	<2	<2
TDS	271.8	5,304.7	11,009

TABLE 10. – Average microbial counts for Mink Unit producing wells. Sampling average period was Oct. 28,1986 - March 17, 1987 for the baseline, and April 3, 1987 - June 1, 1989

Well	Baseline, cells/mL	Post-MEOR, cells/mL
C-BP-2	19	545
C-CP-1	42	2,316
C-CP-3	13	2,533
S-AP-1	50	819
S-AP-2	92	1,018
S-P47R	42	858
S-BP-1	64	636
S-BP-2	27	903
S-BP-3	99	678
S-AP-4	11	387
PLANT	5,595	5,603
TANK BATTERY	[1]	5209

[1]Not sampled or counted before microbial injection.

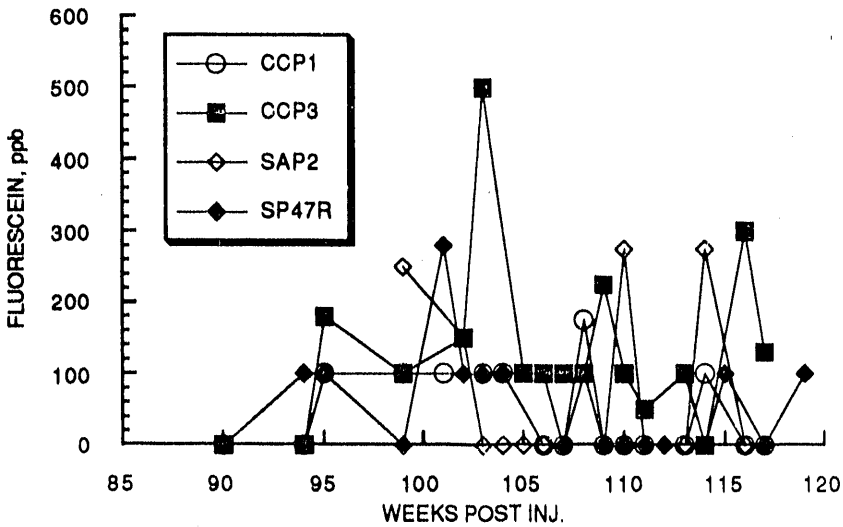

FIGURE 12. – Appearance of tracer in Mink Unit producers.

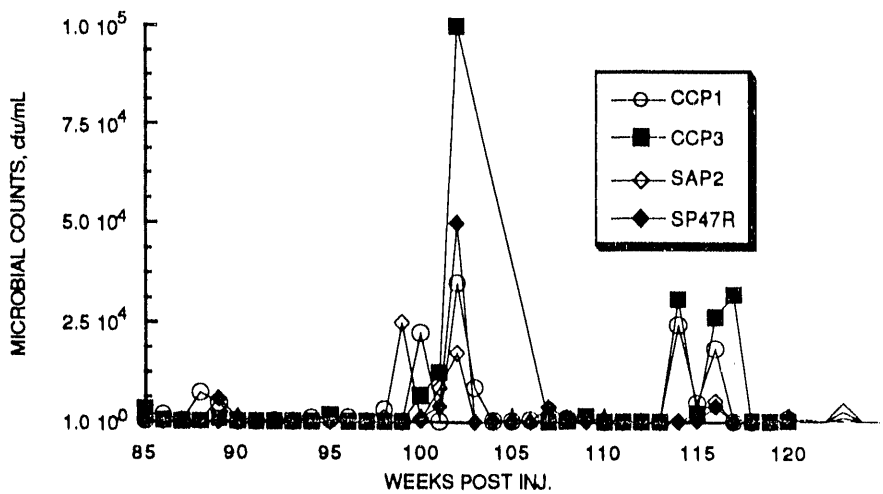

FIGURE 13. – Corresponding appearance of NIPER 1 in Mink Unit producers.

Sulfate-reducing bacteria (SRB) were monitored using lactate-based API sulfate broth. No significant changes occurred in the SRB population after injection of the microbial formulation and molasses. Additionally, no "black water" has appeared before or after injection from the production wells. The low SRB activity is probably a reflection of the low sulfate content of the reservoir (< 15 ppm).

Molasses was assayed in the production well waters using an anthrone carbohydrate assay. One week after injection of the microbial formulation and molasses, molasses was detected in seven producing wells and the tank battery. Interestingly, the injection wells had been shut in during this time period. The injection wells were turned back on 2 weeks after injection, and molasses was subsequently fed twice a week to each of the four injection wells. No molasses was detected in any of the produced water samples after 5 subsequent weeks of its initial appearance. At week 7 of post-microbial injection, molasses sporadically appeared in a few production wells for about 4 weeks, then disappeared. The appearance of molasses early in the test indicated the presence of low-volume, highly permeable streaks within the reservoir. The subsequent disappearance of detectable molasses indicated the establishment of a microbial population that consumed all of the molasses. The presence of the molasses occurred primarily in the north wells of the pattern. This directional flow pattern observed with the molasses analyses corresponded well with the chemical tracer studies.

Total Dissolved Solids

The total dissolved solids (TDS) was measured weekly for every producing well, the water from the Mink Unit tank battery, and the plant injection water since the initiation of the baseline monitoring period (October, 1986). Table 11 shows the average values obtained for each well during the baseline and post-microbial injection. The TDS values did not significantly change during the baseline and post-MEOR periods, and were well within the standard deviations.

pH

The pH of each sample was recorded for the baseline and post-microbial injection periods to determine if microbial injection actually altered the pH of the produced water from the individual wells. Table 12 shows the average of the values obtained during these monitoring times. The pH has not changed significantly since the baseline averages. Although these microorganisms produce short-chained fatty acids such as acetic, propionic, and butyric, these are relatively weak acids produced in such low concentrations that they are probably not lowering the pH. During the backflushing of the injection wells, there seemed to be a significant pH effect, which will be described further in this report. The dissolved oxygen content of the injected water for the Mink Unit was determined to be 5 ppm.

TABLE 11. – Total dissolved solids (TDS, %) from Mink Unit samples. Sampling average period was Oct. 28 - March 17, 1987 for the baseline, and April 3, 1987 - June 1, 1989 for the post-MEOR

Well	Baseline	Post-MEOR
C-CP-1	1.083 ±0.04	0.967 ±0.07
C-CP-3	1.120 ±0.01	1.074 ±0.06
S-AP-1	0.663 ±0.03	0.660 ±0.05
S-AP-2	1.063 ±0.10	1.014 ±0.08
S-P47R	0.275 ±0.03	0.286 ±0.02
S-BP-1	0.479 ±0.03	0.488 ±0.04
S-BP-2	0.557 ±0.10	0.520 ±0.04
S-BP-3	0.478 ±0.02	0.498 ±0.03
S-AP-4	0.730 ±0.03	0.669 ±0.03
C-BP-2	0.656 ±0.02	0.666 ±0.05
PLANT	0.029 ±0.006	0.034 ±0.01
TANK B.	0.500 ±0.02	0.504 ±0.04

TABLE 12. – pH averages for samples taken from the Mink Unit. Sampling average period was Oct. 28 - March 17, 1987 for the baseline, and April 3, 1987 - June 1, 1989 for the post-MEOR

Well	Baseline Avg	Post-MEOR Avg
C-CP-1	6.60 ±0.29	6.85 ±0.38
C-CP-3	6.53 ±0.18	6.83 ±0.31
S-AP-1	6.63 ±0.15	6.79 ±0.31
S-AP-2	6.52 ±0.26	6.79 ±0.34
S-P47R	6.53 ±0.11	6.79 ±0.29
S-BP-1	6.65 ±0.18	6.93 ±0.33
S-BP-2	6.56 ±0.21	6.94 ±0.36
S-BP-3	6.64 ±0.30	6.92 ±0.34
S-AP-4	6.55 ±0.13	6.83 ±0.28
C-BP-2	6.62 ±0.26	6.82 ±0.32
Plant	7.40 ±0.09	7.48 ±0.29
Tank Battery	6.67 ±0.28	6.90 ±0.36

Surface and Interfacial Tensions

Surface tension measurements were conducted twice during the baseline period and weekly on water samples from the Mink Unit post-microbial injection. Interfacial tension (IFT) was not measured during the baseline period, but measurements were initiated soon after microbial injection (May, 1987). Surface and interfacial tension were monitored to provide some indication of microbial surfactant production. Tables 13 and 14 show the results from these measurements. Note the decrease in surface tension and IFT for well S-P47R, the well closest to the microbially treated injection wells. The low value at 38 weeks was probably due to surface active agents produced by the injected microorganisms that traversed quickly through low volume, high permeability streaks in the formation. When the values for 1987, 1988, and 1989 are compared, it is interesting that the 1987 values are the lowest, while the injection plant surface tension was higher in 1987 than in 1988 or 1989. The lower 1987 values are not due to any injection water change from the plant, but rather the microbial injection. The Mink Unit tank battery was not sampled because an emulsion breaker chemical was added to the tank battery which affects the surface and IFT measurements.

TABLE 13. – Interfacial tension values of Mink Unit producing wells. Weeks given are post-MEOR injection. All values are reported in mN/m

Well	1987 38 wk	1988 90 wk	1989 109 wk	Total avg.
C-BP-2	8.97	11.03	11.93	10.64
C-CP-1	8.53	9.63	11.78	9.98
C-CP-3	12.47	13.11	15.21	13.60
S-AP-1	10.01	12.02	14.06	12.03
S-AP-2	10.02	13.36	15.60	13.00
S-P47R	8.38	13.98	15.69	12.68
S-BP-1	9.65	11.06	12.16	10.96
S-BP-2	9.51	10.63	12.56	10.90
S-BP-3	12.61	15.84	16.66	15.05
S-AP-4	11.82	12.55	15.23	13.18

TABLE 14. – Surface tension values of Mink Unit producing wells. Weeks given are post-MEOR injection. All values are reported in mN/m

Well	Baseline	1987 38 wk	1988 90 wk	1989 109 wk	Total avg.
C-BP-2	57.0	53.5	54.6	54.8	54.3
C-CP-1	56.5	51.7	52.6	55.2	53.2
C-CP-3	58.5	55.5	55.2	55.8	55.5
S-AP-1	57.0	54.6	53.6	55.6	54.6
S-AP-2	58.0	53.6	54.5	56.3	54.8
S-P47R	58.6	52.7	56.1	57.7	55.5
S-BP-1	58.0	53.7	54.5	55.4	54.5
S-BP-2	57.0	52.8	53.5	55.1	53.8
S-BP-3	57.5	57.0	56.9	60.6	58.2
S-AP-4	58.0	56.5	56.9	58.9	57.4
PLANT	64.3	63.9	62.5	63.1	63.2

Crude Oil Viscosity

Viscosities of crude oil from each production well were determined weekly during the baseline period, and samples have been measured every 2 weeks since the microbial treatment (table 15). The measurements before and after microbial treatment did not vary significantly (fig. 14). This was expected; although the microorganisms and their products appear to improve microscopic oil displacement efficiency, the amounts of chemicals that they produce probably could not significantly affect crude oil properties. Even in laboratory flask testing, crude oil viscosity is not significantly lowered by microbial growth and activity.

TABLE 15. – Crude oil viscosities (centipoises) from individual wells in the Mink Unit. Sampling average period was Oct. 28 - March 17, 1987 for the baseline, and April 3, 1987 - June 1, 1989, post-baseline

Well	Baseline viscosity, cP	Post-MEOR viscosity, cP
C-CP-1	5.88 ± 1.4	7.12 ± 1.2
C-CP-3	6.71 ± 1.1	7.55 ± 1.3
S-AP-1	5.77 ± 0.8	7.12 ± 1.2
S-AP-2	7.44 ± 1.4	7.58 ± 1.4
S-P47R	7.50 ± 2.5	8.34 ± 2.1
S-BP-1	6.43 ± 1.6	8.13 ± 1.5
S-BP-2	6.23 ± 0.8	7.70 ± 1.3
S-BP-3	6.92 ± 0.9	8.24 ± 1.5
S-AP-4	8.11 ± 1.6	9.69 ± 1.8
C-BP-2	6.79 ± 1.0	8.55 ± 1.4
Total Average	6.78 ± 1.3	8.00 ± 1.5

Injection Pressures

Injection pressures at the microbially treated injection wells have not increased since the beginning of the microbial treatment. In fact, they have decreased since microbial injection, primarily because the injection plant pressures were reduced. Table 16 and figure 15 show pressure values before and after the initiation of the hydraulic fracturing pilot. Injection pressure monitoring was critical to this microbial-enhanced waterflood experiment. In the past, many MEOR researchers have claimed that injection of microorganisms will cause plugging at the face of a

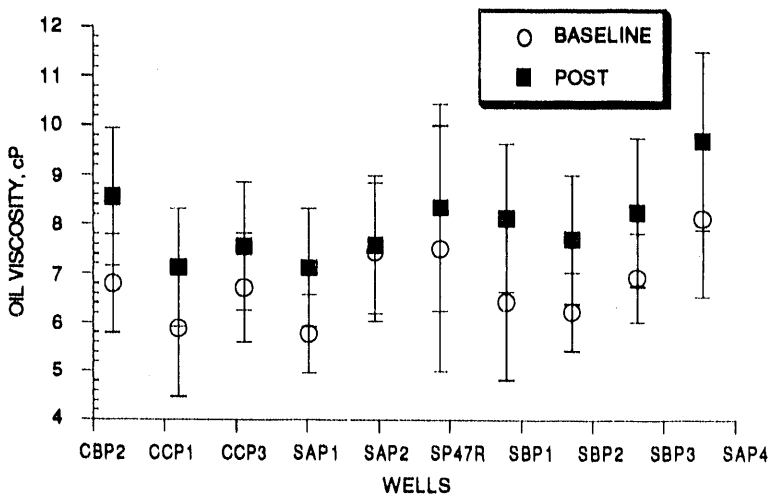

FIGURE 14. – Baseline and post-MEOR oil viscosities of Mink Unit producers.

TABLE 16. – Average injection well pressures (all in psia) of microbially treated Mink Unit injection wells

	1986	1987	1988	1989
Plant	538	544	546	542
Wells	528	532	529	516

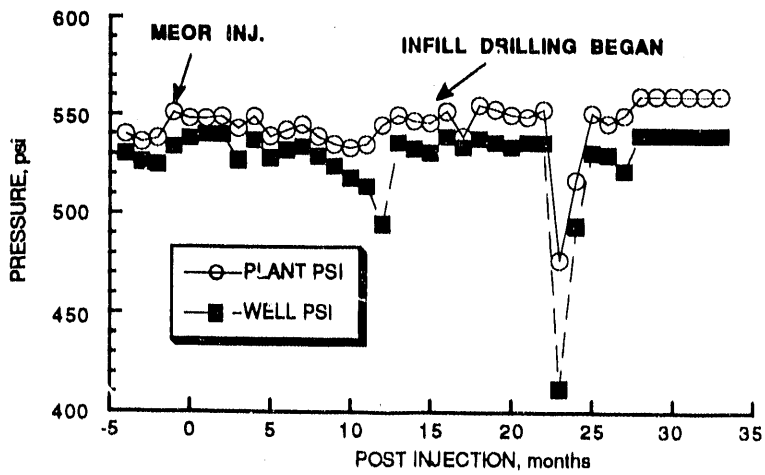

FIGURE 15. – Injection pressures of plant and Mink Unit wells.

formation. In NIPER laboratory coreflooding experiments, no facial plugging was ever observed by NIPER Bac 1. Later coreflood experiments with similar microorganisms have indicated that microorganisms and their products transport at reasonable rates through porous media.[20] Based upon laboratory and field results, we conclude that no adverse plugging effects have occurred because of the microbial injection.

Producing Well Pressures

In May, 1988, Comdisco Resources, Inc. purchased property in Delaware-Childers oil field from B & N Oil Company. The Mink Unit leases were a part of this purchase. The new owners immediately began infill drilling and initiated a hydraulic fracturing project in the nearby Tanner lease (see fig. 1). Wellhead pressures at individual production wells in the project were not routinely measured until the hydraulic fracturing pilot began. However, according to the pumper, all wellhead pressures usually ranged between 20 and 30 psi. When the first well was fractured on the new pilot project (May 29, 1988), a pressure gauge was placed on the nearest well, S-CP-1 (fig. 1), and values were recorded. When the hydraulic fracturing began, the wellhead pressure for well S-CP-1 dropped by about 20 psi. Since that time, individual wellhead pressures have been recorded on a monthly basis (table 17).

Mink Total Produced Fluid

Since the initiation of the project in October, 1986, manual measurement of total volumes was used to provide some indication of the total fluid produced from the Mink Unit production wells. When it became obvious that infill drilling and hydraulic fracturing activity were going to occur, a meter was purchased to record these data. Table 18 and figure 16 show the average fluid production in bbl/d. Again, the drilling and hydraulic fracturing activity affected the fluid production from the Mink Unit. The average for May 1988, before the hydraulic fracturing project began, was twice as high as the numbers have been since that time.

TABLE 17. – Producing wellhead pressures of Mink Unit wells
(values are from 5/88 to 6/89)

Well	Avg wellhead pressures psia
S-CP-1	10
S-BP-1	14
S-BP-2	16
S-BP-3	18
S-AP-1	21
S-AP-2	17
S-P47R	18
C-CP-1	24
C-CP-3	17
S-AP-4	18
C-BP-2	21

TABLE 18. – Total fluid production from Mink Unit

Date	Fluid production, bbl/d
11/86	696
12/86	659
1/87	643
2/87	601
3/87	657
4/87	607
5/87	594
6/87	602
7/87	603
8/87	571
9/87	608
10/87	602
11/87	568
12/87	582
1/88	592
2/88	555
3/88	576
4/88	576
5/88[1]	404[1]
6/88	273
7/88	229
8/88	251
9/88	217
10/88	242
11/88	236
12/88	273
1/89	255
2/89	260
3/89	235
4/89	236
5/89	292
6/89	283
7/89	198
8/89	175
9/89	179
10/89	176

[1]Metering began May/88.

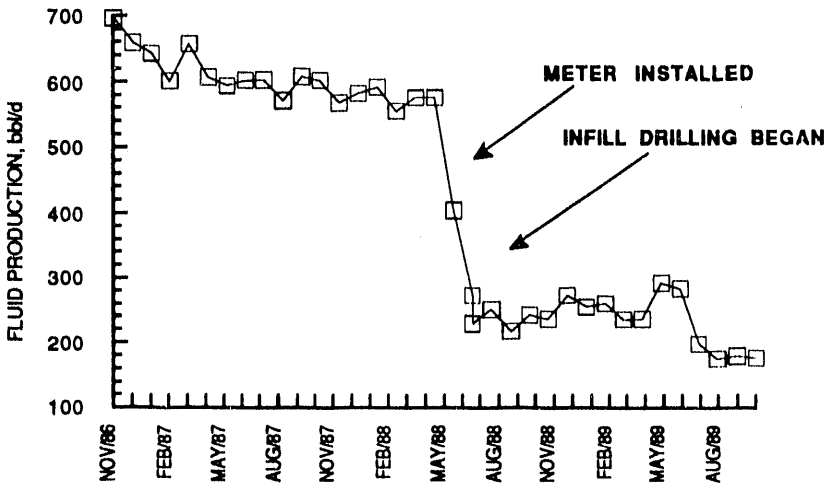

FIGURE 16. – Total fluid production from the Mink Unit.

Injection Well Backflushing

All injection wells on the Mink Unit were being routinely backflushed once a week even before the MEOR project began. This was done routinely to help prevent formation damage and remove debris. The river water is not filtered before it is injected, and the backflushing assists in maintaining good injectivity. In many waterfloods, particularly those with higher dissolved solids and lower permeability, the water must be filtered before injection. Since this process was ongoing, samples were collected every few weeks from the four microbially treated injection wells after 1 hr of backflushing. These samples were analyzed for the following: (1) total microbial populations, aerobic and anaerobic; (2) surface tension; and (3) pH. Samples were observed by phase contrast microscopy in order to visually determine the predominant types of microorganisms. Table 19 and figures 17 through 20 show the results from samples taken during injection well backflushing from 7 to 107 weeks post-injection of the microbial system. Aerobic and anaerobic microbial counts increased to about 10^6 cfu/mL after 22 weeks post injection. Since that time, the counts remained relatively high, which indicates that there was a high concentration of microorganisms in the near wellbore region. This is not surprising, since molasses is continually injected. Because of this high microbial population, pH values are lower than those values measured in the producing wells. Presumably the microorganisms are producing short-chained fatty acids that are lowering the pH. The surface tension values fluctuated throughout the sampling period. At times, the surface tension was low enough to indicate surfactant production by the microbial population; however, the surface tension also rose to values that were comparable to values prior to microbial injection.

TABLE 19. – Mink Unit injection well backflushing results

Weeks Post Inj. 3/24/87	Aerobic, cells/mL	Anaerobic. cells/mL	pH	Surface tension, mN/m
		S-AW-3		
7	5.0×10^3	5.0×10^4	6.1	62.0
10	1.1×10^4	7.4×10^4	6.6	64.0
22	5.0×10^3	5.0×10^4	4.3	45.0
26	1.1×10^7	1.9×10^7	5.55	67.5
34	5.5×10^7	2.0×10^7	5.6	62.5
41	1.0×10^6	1.0×10^7	5.55	53.9
46	1.0×10^7	3.9×10^8	4.1	52.0
50	1.0×10^7	8.1×10^7	4.9	50.5
54	1.1×10^7	1.0×10^8	4.8	51.0
60	5.0×10^8	6.9×10^8	4.4	46.0
74	2.2×10^5	2.1×10^5	6.2	66.2
76	2.0×10^6	5.8×10^6	5.45	55.0
83	1.9×10^6	8.6×10^6	5.3	60.5
86	6.3×10^7	2.3×10^7	5.25	63.8
92	9.4×10^7	1.7×10^8	4.3	48.0
101	2.4×10^6	2.0×10^6	4.8	60.8
107	2.5×10^7	3.2×10^7	5.4	66.0
115	6.6×10^6	2.9×10^7	4.9	58.5
Average	4.4×10^7	8.8×10^7	5.2	57.4
		S-BW-3		
7	5.0×10^3	5.0 x 104	6.05	65.0
10	1.7×10^3	5.1×10^3	6.8	63.1
22	5.0×10^2	5.0×10^4	6.42	53.9
26	3.1×10^6	9.5×10^6	6.35	68.5
34	4.1×10^8	1.3×10^7	5.55	65.0
41	1.0×10^6	1.0×10^7	5.05	46.1
46	1.0×10^7	1.5×10^7	5.9	60.5
50	4.1×10^6	1.0×10^7	6.1	55.5
54	3.0×10^5	5.8×10^5	6.2	58.0
60	6.0×10^6	1.5×10^7	5.6	62.0
74	3.9×10^4	3.6×10^4	5.25	57.2
76	2.0×10^6	8.0×10^6	5.1	51.7
83	1.9×10^5	3.6×10^5	6.2	67.5
86	2.1×10^6	1.5×10^6	5.6	58.2
92	2.6×10^6	3.3×10^6	5.6	59.5
101	2.0×10^5	3.0×10^5	6.0	60.5
107	5.0×10^6	6.3×10^7	5.6	62.5
115	3.0×10^5	5.0×10^5	5.8	68.5
Average	2.5×10^7	8.3×10^6	5.84	60.2

Table 19. – Mink Unit injection well backflushing results (continued)

Weeks Post Inj. 3/24/87	Aerobic, cells/mL	Anaerobic,. cells/mL	pH	Surface tension, mN/m
		C-DW-2		
7	5.0×10^2	5.0×10^4	6.25	57.5
10	3.9×10^3	4.5×10^3	6.7	54.8
22	5.0×10^3	5.0×10^4	4.7	44.3
26	1.0×10^7	4.4×10^7	5.4	51.5
34	1.8×10^7	5.4×10^7	5.35	52.0
41	1.0×10^6	1.0×10^7	4.65	45.0
46	1.0×10^8	8.0×10^8	4.7	48.0
50	1.8×10^7	4.4×10^4	4.8	48.0
54	6.8×10^7	1.1×10^8	4.75	42.0
60	1.3×10^6	2.0×10^6	5.2	51.0
74	1.7×10^4	1.3×10^4	5.25	47.8
76	([1])	2.3×10^7	4.7	49.0
83	([1])	1.0×10^7	4.8	58.0
86	2.1×10^5	7.0×10^6	4.95	49.5
92	1.2×10^6	2.7×10^7	4.6	52.2
101	4.9×10^5	3.2×10^5	5.25	59.8
107	1.0×10^5	2.0×10^6	5.95	62.0
115	1.0×10^5	([1])	5.75	67.0
Average	1.4×10^7	6.4×10^7	5.21	52.2
		S-BW-2		
7	5.0×10^2	5.0×10^3	6.55	59.0
10	4.5×10^3	6.2×10^3	7.40	68.7
22	5.0×10^2	5.0×10^3	6.58	57.0
26	3.7×10^6	7.8×10^6	6.25	65.0
34	7.1×10^7	2.4×10^7	5.9	64.0
41	1.0×10^7	1.0×10^6	5.19	51.2
46	1.0×10^7	4.3×10^8	5.05	57.5
50	1.2×10^7	3.8×10^7	6.03	54.3
54	1.2×10^6	2.8×10^5	6.6	54.0
60	6.0×10^5	1.5×10^6	6.2	65.0
74	2.3×10^5	1.9×10^5	6.25	58.5
76	1.4×10^6	1.5×10^6	6.1	58.2
83	8.3×10^5	4.7×10^6	6.0	64.5
86	1.5×10^7	4.2×10^7	5.65	68.5
92	1.1×10^8	1.4×10^7	5.05	59.0
101	3.6×105	1.0×10^6	4.4	45.2
107	6.0×10^6	2.5×10^7	6.0	61.5
115	3.0×10^6	1.5×10^7	5.4	69.0
Average	1.4×10^7	3.4×10^7	5.9	60.0

([1])Counts were unavailable.

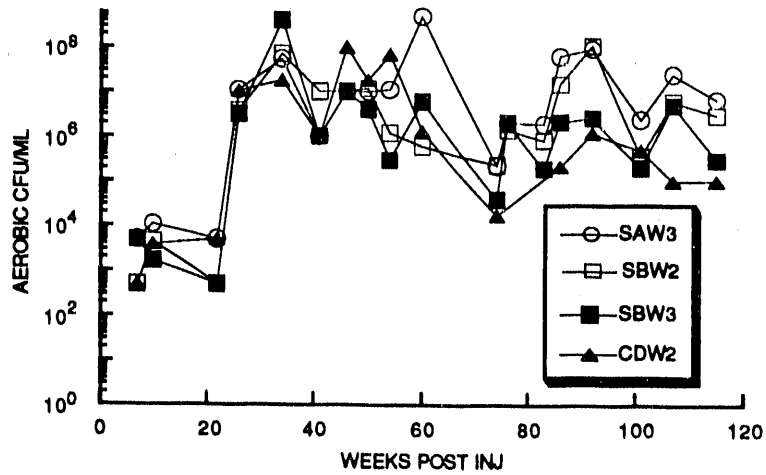

FIGURE 17. – Aerobic microbial counts from routine backflushing of Mink Unit injection wells.

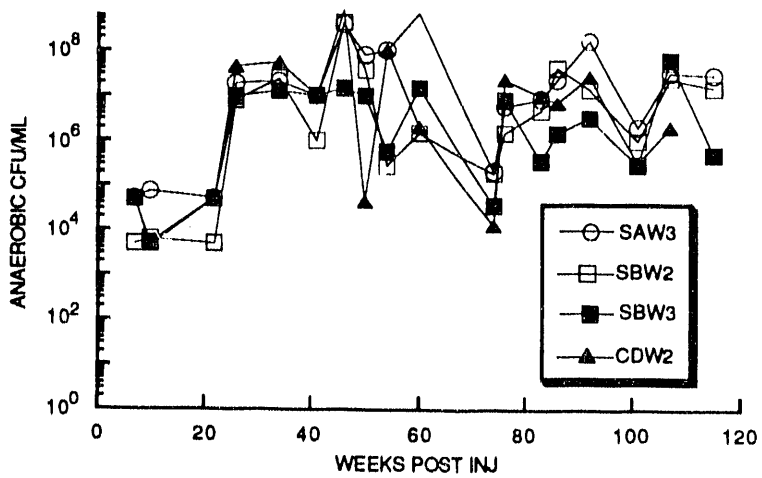

FIGURE 18. – Anaerobic microbial counts from routine backflushing of Mink Unit injection wells.

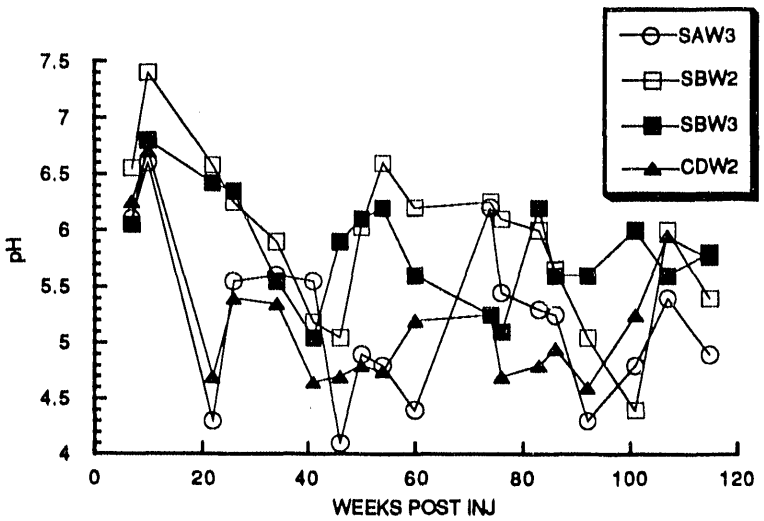

FIGURE 19. – pH of samples from routine backflushing of Mink Unit injection wells.

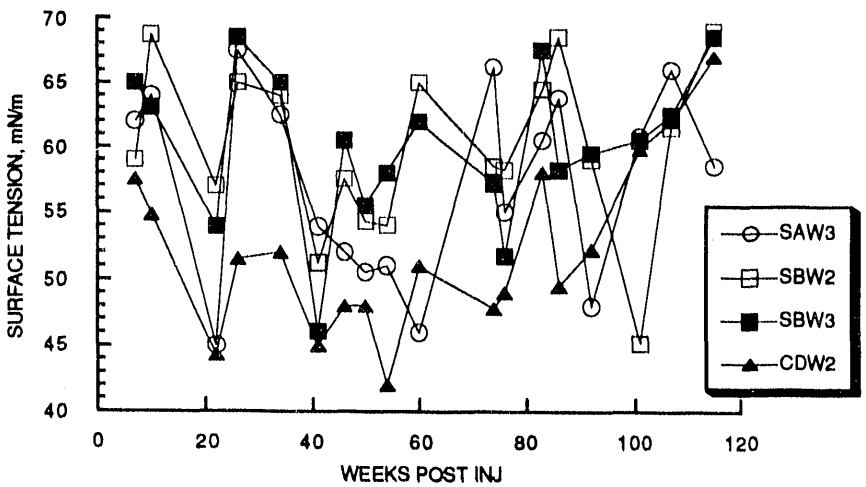

FIGURE 20. – Surface tension of samples from routine backflushing of Mink Unit injection wells.

In addition to the injection well backflush observations, two timed backflush experiments were conducted on the Mink Unit wells. In June of 1988, two of the microbially treated injection wells, S-AW-3 and S-BW-3, were backflushed for approximately 3 hr. Samples were taken every 15 minutes for the first hour, and then every half hour following. The samples were analyzed for microbial populations, surface tension, pH, and microbial products detectable by gas chromatography. The results from the June 1988 timed backflush experiment are presented in tables 20 and 21.

TABLE 20. – Timed backflush results from June, 1988

Time min	pH	Surface tension, mN/m	Aerobic, cells/mL	Anaerobic, cells/mL
			S-AW-3	
0	6.30	67.0	2.0×10^6	1.9×10^6
10	6.35	63.5	4.2×10^6	1.2×10^6
20	6.05	62.0	3.2×10^6	9.2×10^5
30	5.80	63.5	9.9×10^6	1.0×10^6
45	5.75	68.5	7.0×10^6	8.2×10^6
60	5.75	62.0	2.1×10^7	1.1×10^6
90	4.20	51.5	1.4×10^8	7.0×10^7
120	4.20	51.5	4.2×10^7	3.2×10^6
150	4.25	48.5	6.9×10^7	4.5×10^7
180	4.30	48.0	4.0×10^7	4.5×10^7
			S-BW-3	
0	6.25	67.0	4.3×10^5	3.8×10^5
10	4.85	49.5	6.1×10^6	5.7×10^5
20	4.95	48.5	5.2×10^6	5.0×10^5
30	4.95	50.5	4.6×10^7	2.0×10^7
45	5.05	49.5	1.9×10^7	1.3×10^7
60	5.10	50.5	6.5×10^6	7.3×10^6
90	5.15	55.5	8.1×10^6	1.0×10^7
120	5.20	55.0	2.9×10^6	5.2×10^6
150	5.30	53.5	3.5×10^6	5.5×10^6
180	5.40	56.0	2.1×10^6	2.9×10^6

TABLE 21. – Gas chromatographically detectable products from June 1988 timed backflush

Time, min	Compound	Conc., wt %
	S-AW-3	
0	Acetone	0.019
	Propyl alcohol	Trace
10	Acetone	Trace
20	Ethanol	Trace
	Acetone	Trace
	Acetic acid	Trace
	Propionic acid	Trace
30	Ethanol	Trace
	Acetic acid	Trace
45	Acetic acid	Trace
	Propionic acid	Trace
60	None	
90	Ethanol	0.053
	Acetic acid	0.045
	Propionic acid	0.011
120	Ethanol	0.056
	Isopropyl alcohol	0.018
	Acetic acid	0.056
	Butyric acid	Trace
150	Ethanol	0.055
	Isopropyl alcohol	0.013
	Acetic acid	0.057
	Propionic acid	0.015
180	Ethanol	0.061
	Isopropyl alcohol	0.042
	Acetic acid	0.072
	Propionic acid	0.017
	Butyric acid	Trace

TABLE 21. – Gas chromatographically detectable products from June 1988 timed backflush (continued)

Time, min	Compound	Conc., wt %
	S-BW-3	
0	Acetone	0.028
10	Ethanol	0.008
	Isopropyl alcohol	Trace
	Acetic acid	0.037
	Propionic acid	0.029
20	Methanol	Trace
	Ethanol	0.018
	Acetone	Trace
	Isopropyl alcohol	Trace
	Acetic acid	0.034
	Propionic acid	0.022
	Butyric acid	Trace
30	Ethanol	0.012
	Isopropyl alcohol	0.011
	Acetic acid	0.037
	Propionic acid	0.030
	Butyric acid	Trace
45	Methanol	Trace
	Ethanol	0.006
	Isopropyl alcohol	Trace
	Acetic acid	0.018
	Butyric acid	0.007
60	Ethanol	0.007
	Acetone	Trace
	Isopropyl alcohol	Trace
	Acetic acid	0.027
	Propionic acid	0.021
	Butyric acid	Trace
90	Isopropyl alcohol	0.025
	Acetic acid	0.019
	Butyric acid	Trace
120	Ethanol	Trace
	Isopropyl alcohol	Trace
	Acetic acid	0.025
150	Ethanol	Trace
180	Ethanol	Trace
	Isopropyl alcohol	Trace
	Acetic acid	0.023
	Butyric acid	0.012

In September of 1989, a similar timed backflush experiment was conducted on all four of the microbially treated injection wells. The concentration of molasses was also calculated from these samples. Results are presented in tables 22 through 26 and figures 21 through 24. The highest detected concentration of molasses was found in one of the high-volume wells, S-AW-3. This concentration was only 0.05%. The timed backflush minutes and gallons were also converted to radial distance away from the wellbore in feet (table 23). In the high volume wells, this radial distance was determined to be approximately 10 ft away from the wellbore at the end of the timed backflushing in 1989. With the low-volume well, the distance was about 7.3 ft. Comparison of the microbial counts from the 1988 vs. 1989 experiments (fig. 21) indicates that the microorganisms are moving away from the wellbore as expected. The pH results from well S-AW-3 during the 1988 experiment and 1989 experiment showed very dramatically that the pH still dropped in the same manner (fig. 22), but in the 1989 backflushing, the drop was much later during sampling, which again substantiates the theory that the microorganisms and their metabolites are moving away from the wellbore. The results shown in figure 23 also indicated that the surface tension lowering effect was observed much later in the 1989 backflushing.

TABLE 22. – Molasses concentration from Sept. 1989 timed backflush experiment. Molasses had been injected 1 hr prior to start of backflush

Time, min	S-BW-2	S-BW-3	C-DW-2	S-AW-3
		Molasses conc., wt %		
0	0.004	0.001	0.001	0.002
15	0.001	0.007	0.004	0.001
30	0.010	0.010	0.003	0.006
45	0.012	0.010	0.003	0.007
60	0.014	0.004	0.002	0.007
90	0.014	0.007	0.002	0.007
120	0.019	0.004	0.002	0.011
150	0.019	0.005	0.002	0.012
180	0.021	0.007	0.002	0.015
210	0.019	0.006	0.002	0.035
240	0.020	0.008	0.002	0.051
270	0.023	0.010	0.003	0.053
300	0.023	0.008	0.005	0.050

TABLE 23. – Approximate radial distance from wellbore determined from Sept. 1989 timed backflush experiment

Time, min	S-BW-2	S-BW-3	C-DW-2	S-AW-3
		Radial distance, ft		
0	0.0	0.0	0.0	0.0
15	5.3	4.6	4.6	5.3
30	6.0	6.2	6.2	6.0
45	6.6	6.3	6.3	6.6
60	7.0	6.4	6.4	7.0
90	7.5	6.5	6.5	7.5
120	7.9	6.7	6.7	7.9
150	8.3	6.8	6.8	8.3
180	8.7	6.9	6.9	8.7
210	9.0	7.0	7.0	9.0
240	9.4	7.1	7.1	9.4
270	9.7	7.2	7.2	9.7
300	10.0	7.3	7.3	10.0

Numbers are based upon measuring high- and low-volume wells in 1988.

TABLE 24. – Microbial counts from timed backflush - Sept. 1989. All values are in cells/mL

Time, min	S-BW-2		S-BW-3	
	Aerobic	Anaerobic	Aerobic	Anaerobic
0	9.56×10^6	1.35×10^7	1.30×10^6	9.65×10^5
15	1.12×10^5	1.02×10^6	2.18×10^5	1.95×10^5
30	2.50×10^5	4.15×10^6	2.88×10^5	3.05×10^5
45	9.68×10^5	5.75×10^6	2.15×10^5	2.20×10^5
60	7.67×10^5	5.00×10^6	2.66×10^5	2.25×10^5
90	9.34×10^5	5.55×10^6	2.49×10^5	1.85×10^5
120	1.03×10^6	6.10×10^6	2.14×10^5	1.55×10^5
150	1.05×10^6	5.65×10^6	1.77×10^5	1.65×10^5
180	1.05×10^6	5.55×10^6	1.32×10^5	1.70×10^5
210	8.15×10^5	6.20×10^6	1.07×10^5	1.55×105
240	5.40×10^5	5.55×10^6	6.80×10^4	1.10×10^6
270	1.18×10^6	7.95×10^6	9.20×10^4	9.55×10^4
300	8.65×10^5	9.80×10^6	4.60×10^4	6.00×10^5

TABLE 24. – Microbial counts from timed backflush - Sept. 1989 (continued) All values are in cells/mL

Time, min	C-DW-2		S-AW-3	
	Aerobic	Anaerobic	Aerobic	Anaerobic
0	1.38×10^5	1.20×10^5	1.10×10^6	1.11×10^6
15	7.77×10^4	6.85×10^4	3.60×10^5	6.20×10^5
30	1.03×10^5	7.45×10^4	7.35×10^5	1.18×10^6
45	9.03×10^4	9.00×10^4	7.10×10^5	1.35×10^6
60	9.87×10^4	9.10×10^4	7.65×10^5	9.98×10^5
90	9.24×10^4	7.95×10^4	6.40×10^5	1.39×10^6
120	7.14×10^4	6.10×10^4	8.00×10^5	1.41×10^6
150	7.77×10^4	7.75×10^4	6.10×10^5	1.24×10^6
180	7.98×10^4	8.55×10^4	1.11×10^6	5.36×10^6
210	5.57×10^4	7.55×10^4	7.75×10^6	1.01×10^7
240	5.75×10^4	4.30×10^5	7.26×10^6	1.35×10^7
270	6.25×10^4	3.55×10^5	6.72×10^6	4.18×10^6
300	1.54×10^4	5.15×10^5	5.78×10^6	1.07×10^7

TABLE 25. pH and surface tension values for Sept. 1989 timed backflush

Time, min	S-BW-2		S-BW-3		C-DW-2		S-AW-3	
	S.T.[1]	pH	S.T.[1]	pH	S.T.[1]	pH	S.T.[1]	pH
0	76.6	6.60	75.8	6.50	76.5	6.70	76.0	6.00
15		6.55		5.55		5.70		6.25
30		5.30		6.00		5.75		5.75
45		5.15		6.05		5.80		5.70
60		5.05		6.20		5.90		6.65
90		5.10		6.05		6.00		5.75
120		4.80		6.30		6.00		5.20
150		4.75		6.30		6.00		5.10
180	66.6	4.70	75.4	6.30	76.2	6.00	71.4	4.90
210		4.70		6.25		6.00		4.00
240	65.2	4.70	73.2	6.20	74.6	5.90	60.4	3.90
270		4.65		6.15		5.75		3.95
300	62.7	4.65	74.6	6.15	71.5	5.50	60.9	4.00

[1] Surface tension, mN/m.

TABLE 26. – Gas chromatographic products from Sept 1989 timed backflush. Trace amounts are < 0.01%, except for acetic acid and 2,3-butanediol, which are < 0.05%.

Time, min	Compound	Conc., wt %
	S-AW-3	
0	None	
15	None	
30	Propionic acid/n-butanol	Trace
45	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
60	Propionic acid/n-butanol	Trace
	Acetic acid	Trace
90	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
120	Propionic acid/n-butanol	Trace
150	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
180	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid	Trace
210	Methanol	Trace
	Ethanol	0.01
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.05
240	Methanol	Trace
	Ethanol	0.02
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.05
270	Methanol	Trace
	Ethanol	0.02
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.05
300	Methanol	Trace
	Ethanol	0.02
	Acetic acid	0.05
	Propionic acid/n-butanol	Trace
	Butyric acid	0.05

TABLE 26. – Gas chromatographic products from Sept 1989 timed backflush. Trace amounts are < 0.01%, except for acetic acid and 2,3-butanediol, which are < 0.05%

Time, min	Compound	Conc., wt %
	S-BW-3	
0	None	
15	Acetic acid	Trace
30	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
45	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
60	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
90	Acetic acid	Trace
	Butyric acid	Trace
120	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
150	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
180	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
210	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
240	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
270	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
300	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace

TABLE 26. – Gas chromatographic products from Sept 1989 timed backflush. Trace amounts are < 0.01%, except for acetic acid and 2,3-butanediol, which are < 0.05%

Time, min	Compound	Conc., wt %
	C-DW-2	
0	None	
15	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
30	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
45	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
60	Propionic acid/n-butanol	Trace
90	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
120	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
150	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
180	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
210	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
240	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
270	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
300	Acetic acid	Trace
	Propionic acid/n-butanol	Trace

TABLE 26. – Gas chromatographic products from Sept 1989 timed backflush. Trace amounts are < 0.01%, except for acetic acid and 2,3-butanediol, which are < 0.05%

Time, min	Compound	Conc., wt %
	S-BW-2	
0	None	
15	None	
30	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
45	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
60	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
90	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	Trace
120	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.01
150	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.01
180	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.01
210	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.01
240	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.01
270	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	Trace
	Butyric acid	0.02
300	Ethanol	Trace
	Acetic acid	Trace
	Propionic acid/n-butanol	0.01
	Butyric acid	0.02

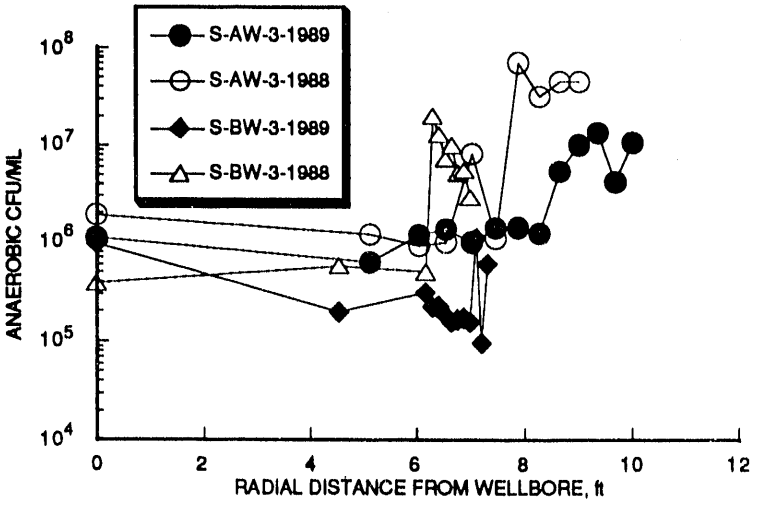

FIGURE 21. – Microbial counts from 1988 and 1989 timed backflush experiments.

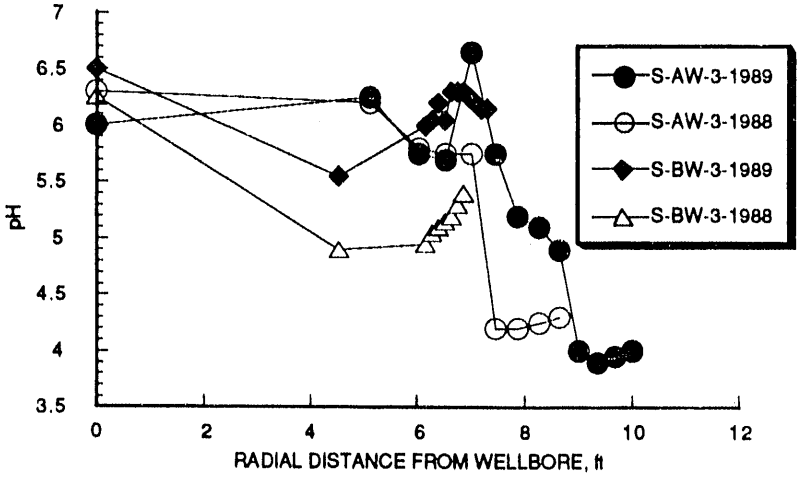

FIGURE 22. – pH from wells S-AW-3 and S-BW-3 during 1988 and 1989 timed backflush experiments.

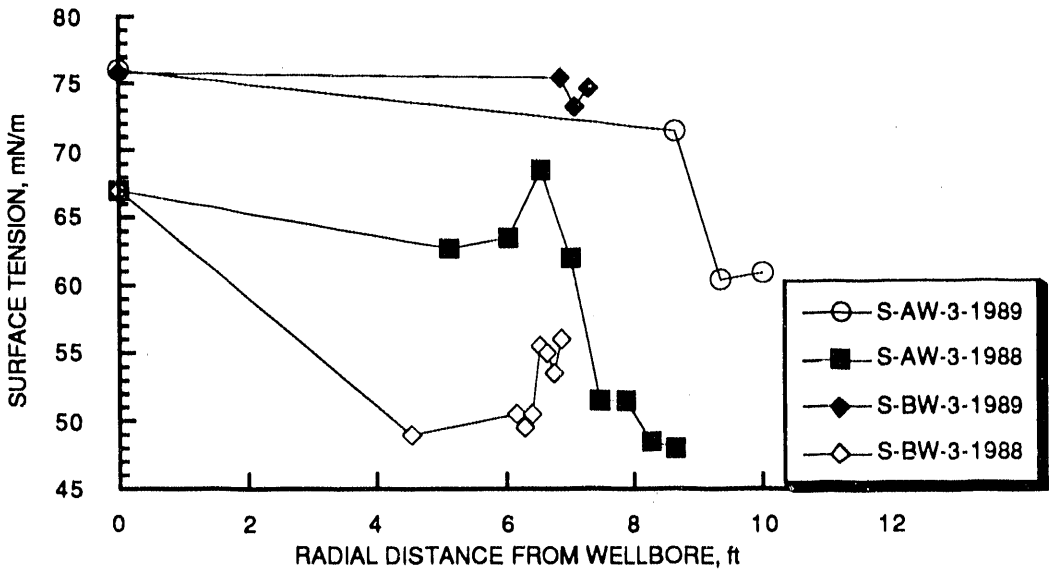

FIGURE 23. – Surface tension of wells S-AW-3 and S-BW-3 during 1988 and 1989 timed backflush experiments.

Gas Analyses

In an earlier report about this field pilot, we reported that gas chromatographic analyses indicated that in two of the producing wells, S-AP-2 and S-P47R, there are compounds present with corresponding retention times to those obtained in Berea sandstone corefloods with the same microbial formulation, NIPER Bac 1.[18] This implies that the microorganisms are metabolizing nutrient in situ and that the products of the fermentation are propagating through the reservoir. Additional information that substantiates this finding is the continued gas chromatographic monitoring of the microbially treated injection well backflush samples. The products from the injected microorganisms, primarily acetic acid, ethanol, and 2,3 butanediol, continue to show a decreasing concentration in the near wellbore region, and a higher concentration further away from the wellbore (fig. 24).

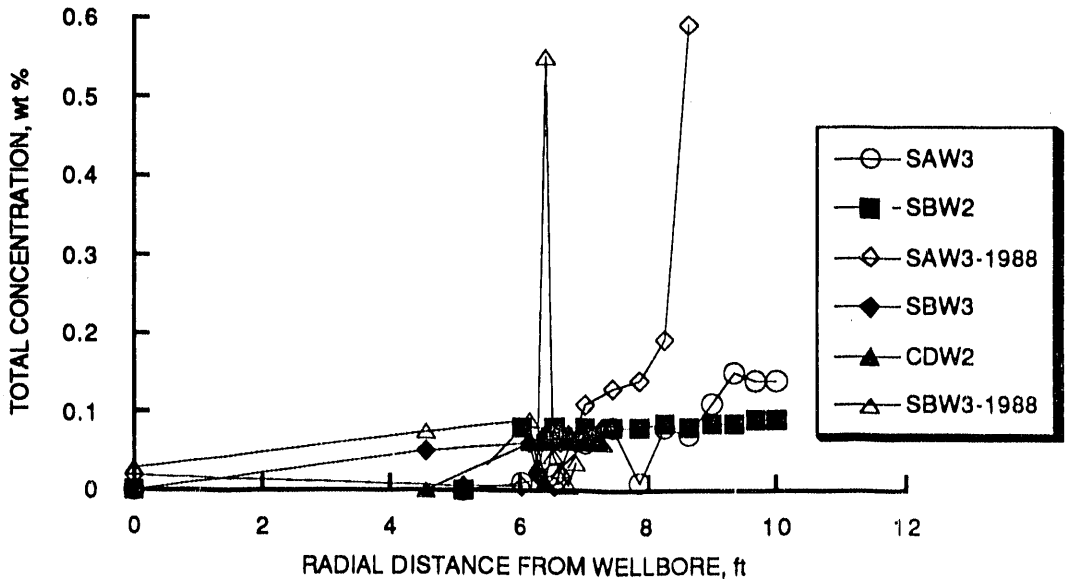

FIGURE 24. – Gas chromatographically detectable products from 1988 and 1989 timed backflushes.

Water-Oil Ratio

The average water-oil ratios (WOR) at all monitored production wells in the Mink Unit have decreased when compared to the averages during the baseline period (table 27 and fig. 25). These WOR's have high standard deviation values, primarily because of gas production in the wells, which causes large fluctuations, but the overall averages have definitely decreased, and in wells S-P47R and C-CP-3, the decrease is significant. Note that in the two off-pattern wells, S-AP-4 and C-BP-2, the WOR has not decreased; thus, the microbial treatment has probably affected those wells closest to the injectors. Figure 26 presents a graph of the WOR for S-P47R vs. the WOR for S-AP-4; the WOR for S-P47R is obviously decreasing, while that of the off-pattern well, S-AP-4, is increasing.

TABLE 27. – Average water-oil ratios of monitored producing wells in Mink Unit

Well	Baseline, WOR	Post-MEOR, WOR
C-CP-1	22 ± 11	19 ± 6
C-CP-3	46 ± 12	36 ± 11
S-AP-1	18 ± 7	15 ± 4
S-AP-2	51 ± 21	43 ± 11
S-P47R	162 ±70	106 ± 16
S-BP-1	44 ± 19	33 ± 7
S-BP-2	32 ± 18	30 ± 8
S-BP-3	46 ± 12	38 ± 12
S-AP-4	77 ± 31	80 ± 16
C-BP-2	72 ± 19	77 ± 14
Mink Unit[1]	98 ± 14	77 ± 11

[1]WOR was calculated from the numbers before infill drilling occurred.

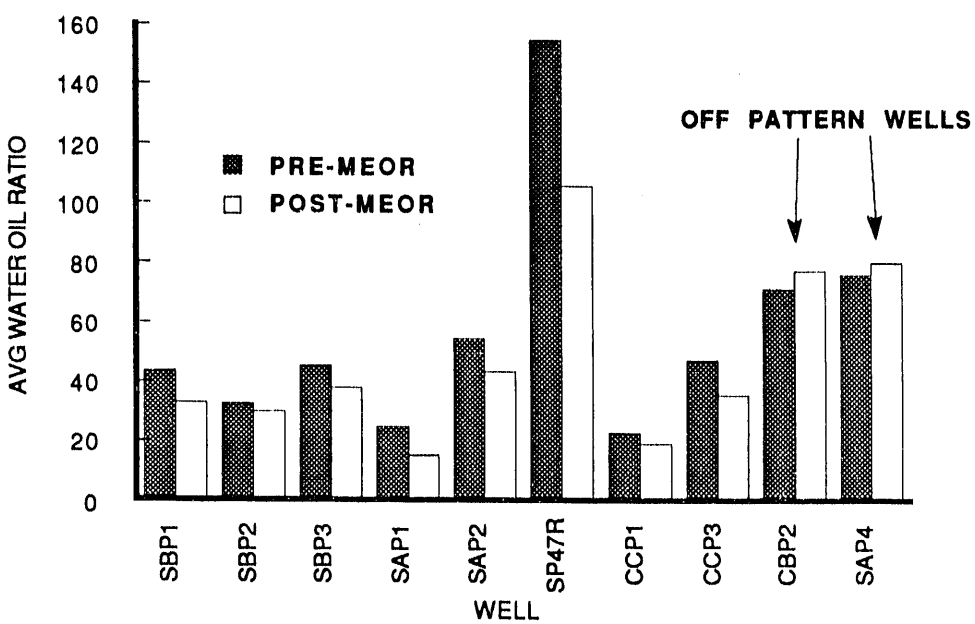

FIGURE 25. – Average water-oil ratios of monitored Mink Unit producers.

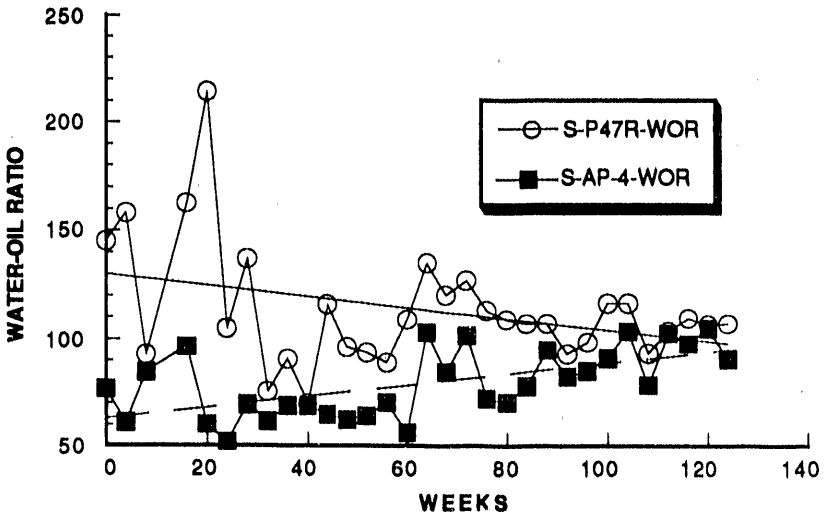

FIGURE 26. – Average WOR of S-P47R and S-AP-4 (off-pattern) wells.

Oil Production Rate

Oil production increased since the microbial injection through May, 1988 (fig. 27). After the infill drilling and hydraulic fracturing occurred, the wellhead pressures at some of the nearest Mink Unit producers were much lower, as was the total produced fluid. The total yearly oil production is presented in figure 28. The MEOR injection obviously had a positive effect on oil production until the drilling and hydraulic fracturing activity and completion of the water injection plant on the nearby Tanner lease. Table 28 and figure 29 show the predicted and actual average production for the years 1981-1990. Since that time, actual oil production has dropped significantly below the predicted decline curve.

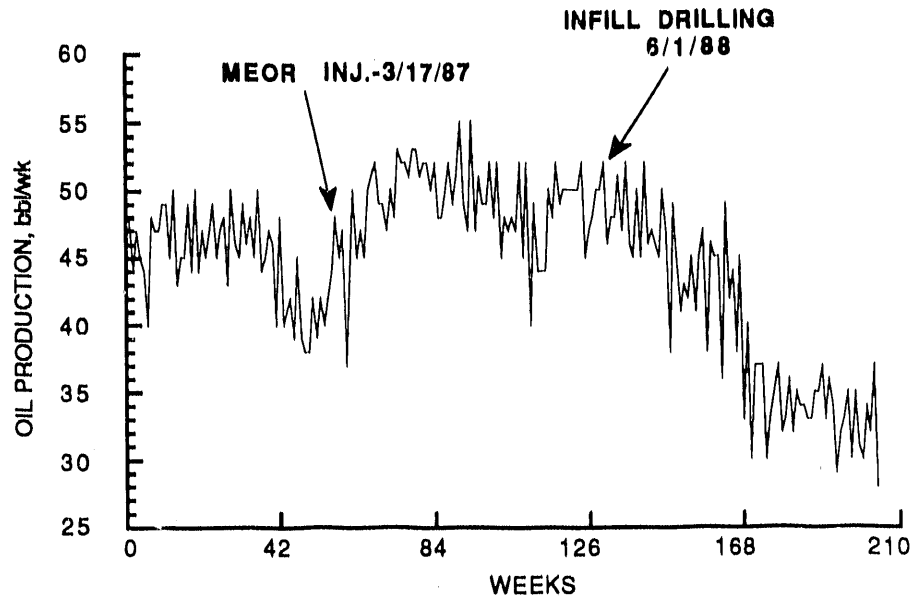

FIGURE 27. – Oil production from Mink Unit from 1985-1989.

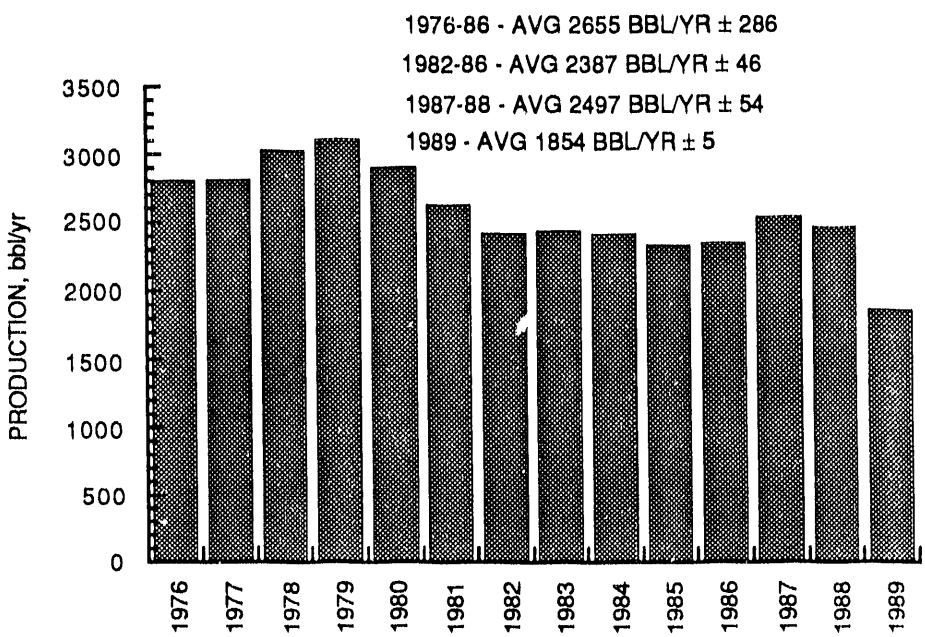

FIGURE 28. – Total yearly oil production for Mink Unit.

TABLE 28. – Predicted and actual oil production rates for the Mink Unit

Year	Production, Predicted avg bbl/wk	Production Actual avg bbl/wk
1981	50.0	50.5
1982	48.8	46.5
1983	47.7	46.8
1984	46.6	46.4
1985	45.6	44.8
1986	44.5	45.1
1987	43.5	48.8
1988A[1]	42.6	48.2
1988B[2]	42.6	46.5
1989	41.7	36.3
1990	40.7	

[1]1988A - Jan. 1 - May 31, 1988.
[2]1988B - Jun. 1 - Dec. 31, 1988.

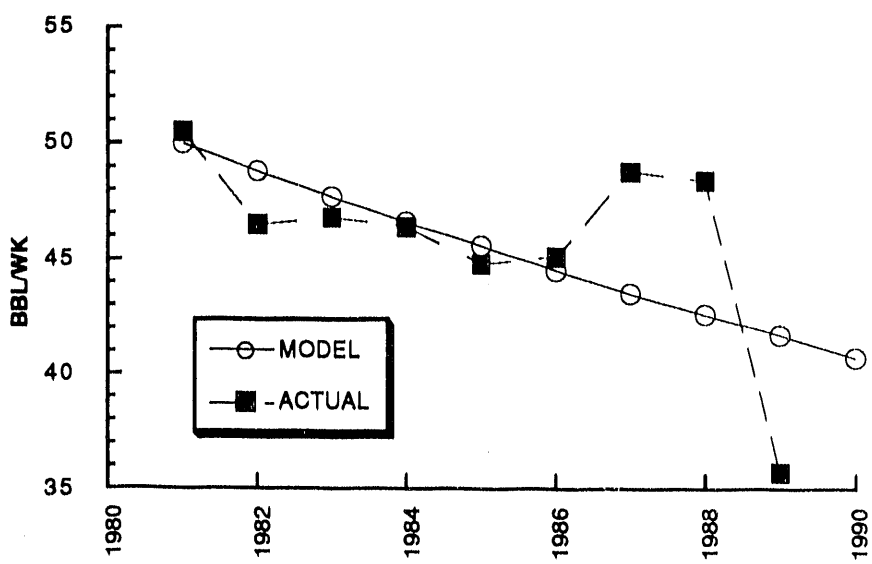

FIGURE. 29. – Predicted and actual average oil production for Mink Unit during 1976-present.

Economic Analysis

Limited economic analyses of this field pilot showed that the major cost of a microbial-enhanced waterflood would be the nutrient support for the microorganisms. Data from more than 50 laboratory microbial coreflooding studies indicate that the cost of nutrient per incremental barrel of oil is about $3-$4/bbl (fig. 30). These data may be somewhat misleading, however, for several reasons: (1) In a microbial coreflood, the amount of nutrient injected

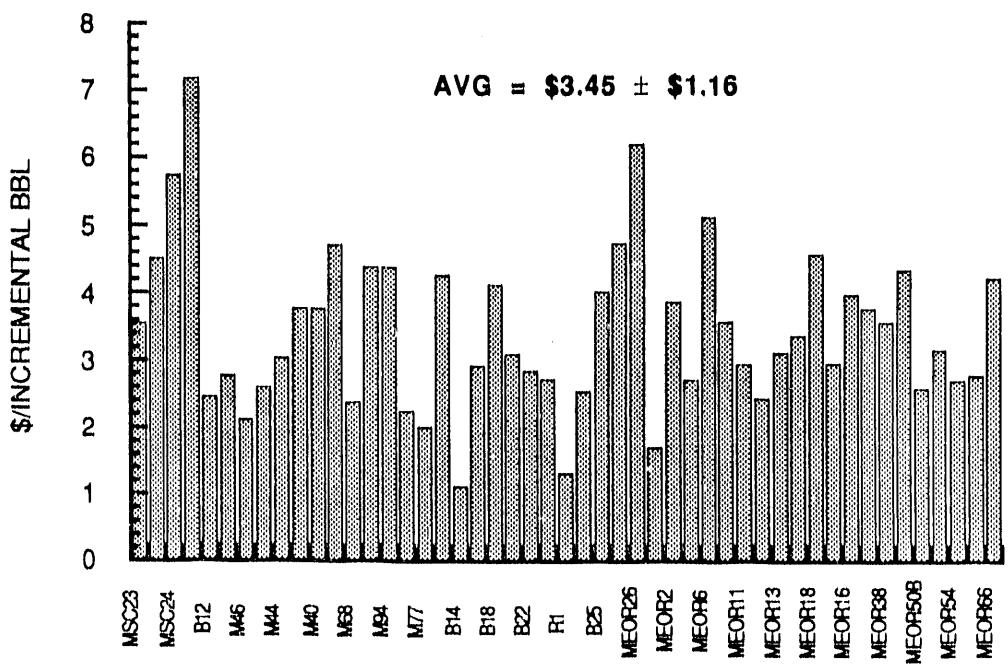

FIGURE 30. – Economics of laboratory microbial coreflood experiments.

is probably an "overkill" situation. Many of these corefloods used almost 0.4 of a pore volume of molasses; this slug size would not be used under reservoir conditions, and (2) In these microbial corefloods, there was usually a 100% sweep efficiency, i.e., the injected fluids contacted virtually all of the rock and, thus, all of the trapped oil. On the other hand, in a reservoir where microorganisms are injected with the waterflood, there may be improvements in microscopic sweep efficiency such that previously uncontacted crude oil is placed in contact with the microbial formulation.

When determining the cost per incremental barrel of oil for the Mink Unit, the following assumptions were made: (1) No cost was assigned for the research and development of the microbial formulation; (2) The cost for

equipment for this particular microbial injection was less than $500; (3) We assumed that we were not overfeeding the microbial population; and (4) We also cannot assume that the total effect of the microbial injection has been attained. Since the chemical tracer just began to appear 1.8 years after injection, based on preliminary data from early breakthrough of tracer, microorganisms should have begun to appear in the production wells about 0.6 - 0.8 years after the tracer appearance. Unfortunately, this would have been about the time that infill drilling near the Mink Unit began, and our sampling period ended.

During the 14 months of microbial/nutrient injection prior to infill drilling and hydraulic fracturing activity, 577 incremental bbl of oil were obtained when compared to the predicted oil recovery by waterflooding alone. A total of 18.7 tons of molasses was injected during this period. Using a nutrient cost of $100/ton, this is equivalent to $3.24 per bbl of incremental oil. This does not take into account any other injection costs, although for this particular project, the costs were fairly minimal. However, this cost also does not include any projected recoveries beyond the time of infill drilling. Since fluorescein was detected in the Mink Unit producing wells after the infill drilling in the Tanner lease, one may assume that the microbial treatment had not yet transported through the formation matrix; thus, the complete effect on incremental oil production that may have occurred would have been masked.

CONCLUSIONS

This microbial-enhanced waterflood field project demonstrated the feasibility of microbial technology in a manner that an independent operator could implement. It is noteworthy that no operating problems were encountered before or during this project, until the initiation of infill drilling and hydraulic fracturing activity nearby in the Tanner lease which was beyond our control. No corrosion problems were experienced; in fact, the sulfate-reducing bacterial populations remained relatively low compared to the baseline counts. There were no problems with injectivity. During the project, laboratory and field data were correlated to develop and document a methodology for conducting microbial-enhanced waterflood field projects. This particular microbial formulation, NIPER Bac 1, and molasses injection improved oil production rates by about 13% and decreased water/oil ratios for producing wells nearest the injection wells up to 35%. All in all, this project has shown promise, particularly for improving oil production from mid-continent stripper wells.

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