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Efficacy of Ethoprop on *Meloidogyne hapla* and *M. chitwoodi* and Enhanced Biodegradation in Soil[1]

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Abstract: Responses of egg masses, free eggs, and second-stage juveniles (J2) of *Meloidogyne hapla* and *M. chitwoodi* to ethoprop were evaluated. The results indicated that J2 were the most sensitive, followed by free eggs and egg masses. In general, *M. chitwoodi* was more susceptible to ethoprop than *M. hapla*. Ethoprop at 7.2 μg a.i./g soil protected tomato roots from upward migrating *M. chitwoodi* for 5 weeks. The zone of protection was extended to 10 and 20 cm below the root zone when 3.6 and 7.2 cm water were applied over 8 days. Ethoprop at 1.8, 3.6, and 7.2 μg a.i./g soil degraded faster and killed fewer *M. chitwoodi* J2 in potato field soil previously exposed to ethoprop than in unexposed soil or sterilized exposed soil. The enhanced biodegradation property of the exposed soil lasted 17 months after the last application of ethoprop. The limited downward movement of ethoprop in the soil, migration of *M. chitwoodi* J2 into the treated zone, presence of resistant life stage(s) at the time of application, and loss of efficacy due to enhanced biodegradation may have a significant effect on the performance of ethoprop.

Key words: biodegradation, Columbia root-knot nematode, ethoprop, *Meloidogyne chitwoodi, Meloidogyne hapla,* migration, nematicide, nematode, northern root-knot nematode, potato, *Solanum tuberosum.*

The Columbia (*Meloidogyne chitwoodi* Golden et al.) and northern (*M. hapla* Chitwood) root-knot nematodes are major pests of potato (*Solanum tuberosum* L.) in the Pacific Northwest. *Meloidogyne chitwoodi* damages potatoes more severely than *M. hapla.* Preplant thresholds are 1 egg/250 cm^3 soil for *M. chitwoodi* (15) and 50 eggs/250 cm^3 soil for *M. hapla.* The lower threshold for *M. chitwoodi* is due to its ability to become active at lower temperatures (6 C) than *M. hapla* (10 C) (4). *Meloidogyne chitwoodi* may complete two to three more generations per year than *M. hapla* on potato (17). Also, *M. chitwoodi* may migrate upward in the soil profile faster and farther than *M. hapla* (12). The nonvolatile organophosphate nematicide ethoprop is registered for use on potato to control *M. hapla* and suppress *M. chitwoodi.* To control *M. chitwoodi,* ethoprop is applied in combination with soil fumigants especially in fields with high nematode population densities distributed deep in the soil profile (11,16). Ethoprop has limited mobility in the soil and must be incorporated into the top 10–15-cm soil layer (17). During the past 10 years, we have evaluated ethoprop for control of *M. chitwoodi* on potato with inconsistent results (17,18). These irregular performances with ethoprop suggested that edaphic and (or) biological factors may have reduced the efficacy of this nematicide in the soil. The half-life of ethoprop may be reduced by ca. 90% in soil with a previous ethoprop application history (20). Also, *M. chitwoodi* second-stage juveniles (J2) may migrate vertically more than 90 cm in a potato field (9). Thus, *M. chitwoodi* populations located below the zone treated with ethoprop move into the treated zone after the nematicidal effects of ethoprop have diminished (8). Another factor is the differential response of *M. chitwoodi* egg masses, eggs, and J2 to ethoprop in the soil (8). The presence of more resistant stages of nematodes in the soil at the time of application may render ethoprop less effective. Studies were initiated to determine 1) the effect of ethoprop on the survival and infectivity of different life stages of *M. chitwoodi* and *M. hapla,* 2) the downward movement and residual effect of ethoprop on the migration of *M. chitwoodi,* and 3) the efficacy of ethoprop to control *M. chitwoodi* in soil with previous ethoprop application history.

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Materials and Methods

Hatching and survival experiments: Hatching and survival tests were conducted in polyvinylchloride (PVC) rings (4.5 cm d × 3.5 cm deep) sealed on one end with a 25-μm-pore nylon screen. These screens retained egg masses and free eggs but allowed J2 passage. Egg masses and freed eggs were placed in screens, strained, and set on thin plastic supports in 5.5-cm-d petri dishes. All concentrations of ethoprop are reported as μg a.i./ml or μg a.i./g soil. A geometric series of ethoprop (69.6% emulsifiable concentrate) concentrations (0, 5, 10, 20, 40, 80, 160 μg/ml) were prepared in distilled water, and 10 ml nematicide solution was added to each petri dish to cover the nematodes on the screens. Second-stage juveniles were suspended in 5 ml water and placed directly in petri dishes, and equal volumes of ethoprop at 10, 20, 40, 80, 160, and 320 μg/ml were added and mixed. The treatments were replicated and randomly assigned to four complete blocks placed in trays. The trays were covered with plastic sheets to minimize evaporation and incubated at 18 C. After 4 days ethoprop was removed by repeated immersion of egg masses and freed eggs in tap water. Second-stage juveniles were collected from petri dishes, washed five times by centrifugation at 3,000 *g* for 5 minutes, and resuspended in tap water. Egg masses and J2 were placed around the root system of 3-week-old Columbian tomato (*Lycopersicon esculentum* Mill.) plants to determine nematode infectivity. Tomato roots were harvested after 3 weeks and stained with acid fuchsin (3), and juveniles were counted. Hatch inhibition was quantified by the J2 in the petri dishes. The effective dose that prevented 50% (ED_{50}) of egg hatch or J2 infection of tomato roots was calculated by probit analysis (5).

Four experiments were conducted using *M. chitwoodi* and *M. hapla* inocula. In the first experiment, pieces of tomato root containing three egg masses were exposed to ethoprop for 4 days, after which the egg masses were bioassayed. In the second experiment, *M. chitwoodi* egg masses were exposed to ethoprop for 21 days, the solution was replaced every 4 days, and numbers of J2 that hatched were determined. In the third experiment, 1,000 eggs extracted from tomato roots with 0.5% NaOCl (6) were exposed to ethoprop for 4 days and the numbers of J2 hatched were determined. The remaining unhatched eggs were washed free of ethoprop and incubated an additional 12 days in water, and freshly hatched J2 were counted every 4 days. To test infectivity, *M. chitwoodi* J2 were stored at 12 C and bioassayed on tomato as before. In the fourth experiment, 1,000 freshly hatched J2 (21) were exposed to the ethoprop series for 4 days prior to bioassaying on tomato.

Mobility and residual effects of ethoprop: Downward movement and residual effect of ethoprop to control migrating *M. chitwoodi* were studied in columns constructed of PVC rings (8.25 cm d × 5.0 cm high) (12). Columns were filled with methyl bromide fumigated sandy loam soil (83% sand, 15% silt, 2% clay, pH 6.9, 0.5% OM) and adjusted to field capacity (10% water by weight) at the beginning of the studies.

The effect of water regimes on the downward movement of ethoprop was studied in the first experiment. A 1-cm-thick layer of soil (500 g) was sprayed with a solution of 5 μl ethoprop suspended in 10 ml water and mixed thoroughly resulting in an estimated 7.2 μg/g soil. The actual concentrations of ethoprop measured (19) several times in the course of this study were 6.0–6.9 μg/g soil. The untreated control soil received 10 ml water. The treated or untreated soil was packed (bulk density = 1.4 g/cm^3) into single PVC rings each sealed on one end with 25-μm-pore nylon screen that confined root growth but allowed nematode passage. Three-week-old tomato seedlings were planted one to each sealed ring, and each ring was assembled on top of a seven-ring (35 cm tall) soil column. Four columns per treatment were placed on the surface of dry soil in 10-cm-d clay pots at 18 C, and 2,500 freshly hatched J2 were introduced 35 cm below the top

ring. The columns received 0.45 or 0.90 cm water daily for 8 days. Soil moisture in each ring was recorded by drying 50-g samples at 105 C for 24 hours. Soil residues of ethoprop in the 0.90-cm water regime were determined upon completion of the experiment (19). A portion of soil obtained from each ring of the 0.45-cm water regime was wet sieved to extract nematodes prior to bioassay.

To test the residual effects of ethoprop, a two-phase experiment was conducted. For the first phase, rings with 25-μm-pore nylon screens were filled with ethoprop-treated or untreated soil as in the previous experiment. A 3-week-old tomato seedling was planted in each ring, and a plastic bag was placed over the ring to minimize moisture loss. The rings were placed on the surface of dry soil and incubated at 18 C with a 14-hour photoperiod for 48 days. The plants received 0.90 cm water weekly, and excess water drained freely into the dry soil. For the second phase, every 6 days four replicates of treated or untreated rings were assembled on top of three-ring (15 cm tall) soil columns. A suspension of 2,500 J2 in 5 ml water was injected into the column through a port in the second ring from the bottom. Tubes were irrigated with 0.45 cm water daily for 8 days before the rings were separated, and the content in each ring was bioassayed with tomato seedlings.

Enhanced biodegradation of ethoprop in field soil with previous ethoprop history: Soil used in three experiments from a commercial field near Plymouth, Washington, was a Quincy sand (mixed, mesic xeric torripsamments) which had received ethoprop (13.2 kg/ha) in 1987 and 1988. Soil was collected in February 1989, 9 months after the last application of ethoprop (exposed soil). Soil was also collected from an adjacent area containing native vegetation with no previous cropping or ethoprop history (unexposed soil). Both soils were a loamy sand (79% sand, 19% silt, 2% clay, pH 6.1, 0.5% OM) and were held at field capacity (10% moisture by weight). The soil samples were used immediately or stored at 15–18 C up to 10 months.

The first experiment was conducted to evaluate enhanced degradation of ethoprop. A solution of 10 ml water containing 1.25 μl ethoprop (ca. 1.8 μg/g soil) was sprayed on the surface of 500 g steam-sterilized exposed soil (120 C under 103.4 KPa for 2–3 hours and aerated for 48 hours), unsterilized exposed soil, and unexposed soil; then treated soils were mixed thoroughly in plastic bags. Exposed and unexposed soil sprayed with water served as controls. The soil in each plastic bag was infested with 2,000 *M. chitwoodi* J2 in 2 ml water 1 week after ethoprop treatment, and the bags were shaken gently to uniformly distribute the nematodes. After 1 week incubation at 15–18 C, the soil from each bag was placed in a 7.5-cm-d clay pot and a 3-week-old Columbian tomato seedling was transplanted. The treatments were replicated five times and arranged in randomized complete blocks on a greenhouse bench. The pots were maintained for 3 weeks at 22–26 C before roots were washed free of soil and stained with acid fuchsin (3). The survival rate was determined by counting the nematodes in the roots. The data were subjected to analysis of variance, and means were separated by Duncan's multiple-range test.

The second experiment was conducted to determine the effect of incubation period following treatment on the efficacy of ethoprop in exposed soil. The procedures were similar to the previous experiment, except that the nematodes were added to unsterilized exposed soil immediately after ethoprop treatment or 1 week later.

The third experiment was conducted to determine the effect of time on enhanced biodegradation of ethoprop. Ethoprop (1.8 μg/g soil) was added to the exposed soil 0, 8, 9, and 10 months after collection from the field. This corresponded to 9, 17, 18, and 19 months after the soil was last treated with ethoprop. Control treatments included exposed soil treated with water and

TABLE 1. Effective dose (ED_{50}) of ethoprop on second-stage juveniles (J2), free eggs, and egg masses of *Meloidogyne chitwoodi* and *M. hapla*.

Life stages	Exposure (days)†	ED_{50} (μg/ml) *M. chitwoodi*	ED_{50} (μg/ml) *M. hapla*
Infectivity			
Egg masses‡	4	160.0 a	160.0 a
J2	4	5.6 d	6.8 d
Hatching			
Egg masses	21	0.2 e	
Free eggs	4	46.0 c	83.3 b

The values used for probit analysis to calculate ED_{50} were means of four replicates. The values in columns and rows followed by different letters had different ($P < 0.05$) variances of log ED_{50}.

† Nematode life stages were exposed to ethoprop at 0, 5, 10, 20, 40, 80, and 160 μg/ml at 18 C.

‡ ED_{50} for infectivity tests of egg masses exposed for 4 days were beyond the highest concentration of ethoprop (160.0 μg/ml) tested.

TABLE 2. Percentage of *Meloidogyne chitwoodi* second-stage juveniles (J2) that hatched from 1,000 free eggs exposed to ethoprop for 4 days and placed in water for 12 days and percentage of hatched J2 that infected tomato roots.

Ethoprop concentration (μg/ml)	Hatched	Infected tomato roots
0	63 a	26 a
5	47 ab	25 a
10	41 ab	20 ab
20	48 ab	12 ab
40	43 ab	7 abc
80	20 b	4 bc
160	1 c	0 c

Values are means of four replicates. Means followed by the same letter do not differ ($P < 0.05$) according to Duncan's multiple-range test.

sterilized exposed soil treated with ethoprop. All other procedures were similar to those in the first experiment.

The fourth experiment was conducted to determine the effect of ethoprop concentration on the ability of the biological agent(s) to reduce the nematicidal efficacy. Soil for this study was obtained from a site which had been used for evaluating nematicides, including ethoprop, for the past 10 years at the Irrigated Agriculture Research and Extension Center, Prosser, Washington. This loamy fine Hezel sand soil (81% sand, 17% silt, 2% clay; pH 6.7, 0.9% OM, and 13.6% moisture by weight at field capacity) was initially tested for enhanced biodegradation of ethoprop. Steam-sterilized and unsterilized soils were treated with ethoprop at 1.8, 3.6, and 7.2 μg/g soil. All other procedures were similar to those in the first experiment. Only significant ($P < 0.05$) treatment comparison will be discussed unless stated otherwise.

Results and Discussion

Hatching and survival: Four days of exposure to various concentrations of ethoprop did not influence the viability of eggs within egg masses of *M. chitwoodi* or *M. hapla*, and ED_{50} values to inhibit infection for both nematode species were beyond the ranges of concentrations tested (Table 1). However, when *M. chitwoodi* egg masses were exposed to ethoprop for 21 days, hatching was affected adversely by all concentrations and ED_{50} was 0.23 μg/ml. These results suggest that the gelatinous matrix restricts movement of ethoprop into the egg mass, but after prolonged exposure, ethoprop adversely affects egg viability even at low concentrations.

Eggs freed from the gelatinous matrix by NaOCl were more sensitive to ethoprop than were eggs within egg masses, and the ED_{50} to inhibit hatching was 46.0 μg/ml for *M. chitwoodi* and 83.3 μg/ml for *M. hapla* (Table 1). When the chemical solution was replaced with water, suppression of egg hatch was reversed in all treatments except the 80 and 160 μg/ml concentrations. These two concentrations of ethoprop not only affected the hatch of *M. chitwoodi*, but also the infectivity of hatched J2 (Table 2).

The J2 stage was the most sensitive life stage to ethoprop, with ED_{50} of 5.6 μg/ml for *M. chitwoodi* and 6.8 μg/ml for *M. hapla* (Table 1). These values were lower than ED_{50} values for free eggs (46.0 μg/ml) and egg masses (>160 μg/ml).

Meloidogyne chitwoodi and *M. hapla* were differentially sensitive to ethoprop (Table 1), and ED_{50} for free eggs hatching was lower for *M. chitwoodi* than for *M. hapla*.

TABLE 3. Effect of daily irrigation water on the downward movement of ethoprop 8 days after application measured by vertical migration of *Meloidogyne chitwoodi* second-stage juveniles (J2) and ethoprop residue in soil columns.

Column depth (cm)	*M. chitwoodi* detected in roots				Ethoprop residue† (μg/ml)
	0.45 cm water		0.90 cm water		
	E	U	E	U	
0–5	−	+	−	+	0.19
10	−	+	−	+	1.73
20	+	+	−	+	0.62
30	+	+	+	+	0.04
40‡	+	+	+	+	0.003

Detection (− = no, + = yes) of nematodes at each depth was based on examination of four tomato roots. The 0–5-cm ring contained soil treated with ethoprop (E) at 7.2 μg/g or untreated (U) and planted with a tomato seedling before assembling on top of the columns.

† Ethoprop residue at different depths was determined 8 days after assembling a treated ring on top of a soil column and irrigating daily with 0.90 cm water.

‡ A suspension of 2,500 J2 was added to this ring in all soil columns and incubated at 18 C for 8 days.

Soil samples taken after fumigation with metham sodium containing mixed populations of *M. chitwoodi* and *M. hapla* support these findings that *M. hapla* is more difficult to kill than *M. chitwoodi* (unpubl.). Nevertheless, ethoprop is effective in minimizing *M. hapla* damage on potato because it is less prolific (17). Similar differential sensitivity among other *Meloidogyne* species to ethoprop has been reported (10).

Mobility and residual effects of ethoprop: Meloidogyne chitwoodi J2 migrated upward 35 cm within 8 days and infected tomato roots in the untreated soil under both irrigation regimes (Table 3). The effective zones of nematode control below the ethoprop treated ring were 10 cm with the 0.45 cm/day irrigation regime and 20 cm with the 0.90 cm/day regime. Inactive J2 5–10 cm below the treated ring were recovered by wet-sieving soil, indicating that J2 migrated upward before a lethal concentration of ethoprop was leached from the treated soil profile.

The distribution of ethoprop residue in columns receiving 0.90 cm water daily indicated that most of the ethoprop was leached from the 0–5-cm zone of incorporation and deposited at depths of 10 and 20 cm (Table 3). The concentration of 0.62 μg/g detected at 20 cm effectively controlled *M. chitwoodi*. Ethoprop concentration below 20 cm declined and did not restrict J2 migration. These results agree with others (19) that downward movement of ethoprop is restricted. A maximum downward movement of 20–30 cm following the

TABLE 5. Efficacy of ethoprop on *Meloidogyne chitwoodi* in Quincy sand previously exposed to ethoprop, unexposed soil, and sterilized exposed soil.

Treatment	Ethoprop (1.8 μg/g)	*M. chitwoodi*/ tomato root system (no.)
Unexposed	−	422 a
Unexposed	+	4 b
Exposed	−	248 a
Exposed	+	248 a
Sterilized exposed†	+	4 b

Values are means of five replicates. Means followed by the same letter do not differ ($P < 0.05$) according to Duncan's multiple-range test. Soil was infested with 2,000 second-stage juveniles 1 week after ethoprop treatments and bioassayed 1 week later with tomato seedlings.

† Sterilized at 120 C and 103.4 KPa for 2 hours.

TABLE 4. Residual effect of ethoprop on vertical migration of *Meloidogyne chitwoodi* second-stage juveniles (J2) in soil columns 1–8 weeks after treatment.

Column depth (cm)	*M. chitwoodi* J2 in tomato roots											
	1		3		4		5		6		8	
	E	U	E	U	E	U	E	U	E	U	E	U
0–5	−	+	−	+	−	+	−	+	+	+	+	+
10	−	+	−	+	−	+	−	+	+	+	+	+
15†	−	+	−	+	−	+	+	+	+	+	+	+
20	−	+	+	+	+	+	+	+	+	+	+	+

Detection (− = no, + = yes) of nematodes at each depth was based on examination of four tomato roots. Soil in the 0–5-cm ring was treated with ethoprop (E) at 7.2 μg/g or untreated (U) and irrigated weekly with 0.9 cm water for 1–8 weeks before being assembled on top of 3-ring columns. Thereafter, 0.45 cm water was added daily for 1 week before each ring was bioassayed.

† A suspension of 2,000 J2 was added to this ring.

TABLE 6. Efficacy of different concentrations of ethoprop on *Meloidogyne chitwoodi* in Hezel sand previously exposed to ethoprop and sterilized exposed soil.

Ethoprop (μg/g)	*M. chitwoodi*/tomato root system (no.)
Exposed	
0.0	311 a
1.8	141 ab
3.6	128 b
7.2	76 b
Sterilized exposed†	
1.8	0 d
3.6	4 c
7.2	0 d

Values are means of five replicates. Means followed by the same letter do not differ ($P < 0.05$) according to Duncan's multiple-range test. Soil was infested with 2,000 second-stage juveniles 1 week after ethoprop treatments and bioassayed 1 week later with tomato seedlings.

† Soil was sterilized at 120 C and 103.4 KPa for 2 hours.

addition of 7.5 cm water has been reported (1,7).

Soil treated with ethoprop for 1, 3, and 4 weeks created nematistatic zones of 15, 10, and 10 cm, respectively, below the treated soil (Table 4). The nematistatic zone was reduced to 5 cm in 5-week-old ethoprop-treated soil, and thereafter *M. chitwoodi* migrated into the top 5 cm and infected tomato roots (Table 4). *Meloidogyne chitwoodi* migrated freely in untreated soils throughout the experiment. The residual life of ethoprop determined in this test is shorter than previously reported (2). The difference may be related to the experimental procedure or nematode species or both. Bunt (2) obtained significant reduction in number of *Pratylenchus penetrans* (Cobb) Chitwood & Oteifa per root system 16 weeks after drenching ethoprop at 3 μg/ml on an established corn plot. Thus, the entire population of *P. penetrans* was exposed to the initial concentration of the chemical. In our study, freshly hatched *Meloidogyne* J2 continuously were added into the soil columns while residual ethoprop concentrations continued to diminish.

Our data indicate that ethoprop applied at 7.2 μg/g soil will provide a 10–20-cm nematistatic zone for up to 5 weeks with 3.6 cm and 7.2 cm water applied in 8 days. This period of control would not be adequate to prevent severe losses on potato by *M. chitwoodi*. Potato growers in the Pacific Northwest apply a maximum of 2.5 cm water after ethoprop application. Ethoprop 6EC applied as a pre-emergence broadcast, followed by 2.5 cm water failed to control *M. chitwoodi* on potatoes in the field, whereas mechanical incorporation of the same formulation frequently provided control (15,17). In addition, the efficacy of ethoprop may also be determined by the nematode life stages present at the time of application.

TABLE 7. Effect of incubation period on efficacy of ethoprop on *Meloidogyne chitwoodi* in Quincy sand previously exposed to ethoprop.

Incubation period (weeks)	Ethoprop (1.8 μg/g)	*M. chitwoodi*/tomato root system (no.)
0	−	246 a
1	−	337 a
0	+	63 b
1	+	248 a

Values are means of five replicates. Means followed by the same letter do not differ ($P < 0.05$) according to Duncan's multiple-range test. Soil was infested with 2,000 second-stage juveniles simultaneously or 1 week after ethoprop treatments and bioassayed 1 week later with tomato seedlings.

Enhanced biodegradation of ethoprop in field soil with previous ethoprop history: The effi-

TABLE 8. Effect of long-term incubation on efficacy of ethoprop to control *Meloidogyne chitwoodi* in Quincy sand exposed to ethoprop.

Last exposure (months)	Incubation period (months)	Ethoprop (1.8 μg/g)	*M. chitwoodi*/ tomato root system (no.)
9	0	−	248 a
		+	248 a
17	8	−	252 a
		+	64 b
18	9	−	128 a
		+	10 c
19	10	−	197 a
		+	7 c
Sterilized†		+	4 c

Values are means of five replicates. Means followed by the same letter do not differ ($P < 0.05$) according to Duncan's multiple-range test. Soil was collected 9 months after the last application of ethoprop and used immediately, or incubated at 15–18 C. The treatment soils were infested with 2,000 second-stage juveniles 1 week after ethoprop addition and bioassayed with tomato 1 week later.

† Average of four trials. In each trial, soil was sterilized at 120 C and 103.4 KPa for 2 hours.

cacy of ethoprop at 1.8 μg/g soil on *M. chitwoodi* was reduced in exposed soil compared to ethoprop applied to unexposed soil (Table 5). Numbers of *M. chitwoodi* recovered from the exposed soil treated with ethoprop were not different from those in treatments that did not receive ethoprop. Steam sterilizing the exposed soil restored the efficacy of ethoprop, similar to the unexposed soil treated with ethoprop. Increasing the concentration of ethoprop to 7.2 μg/g in the exposed soil reduced the numbers of *M. chitwoodi,* but numbers were higher than those in the steamed exposed soil (Table 6). The number of nematodes that survived was inversely proportional ($r = -0.65$) to the ethoprop concentration. Ethoprop at 13.2 kg/ha (assuming soil bulk density = 1.3 g/cm^3) incorporated 15 cm deep, as recommended for potato, results in ca. 6 μg/g soil, which is less than 7.2 μg/g soil biodegraded in these experiments. Thus, enhanced biodegradation of ethoprop under commercial conditions is a concern.

Ethoprop applied to unsterilized exposed soil was not as effective in controlling *M. chitwoodi* when the nematode was added 1 week after ethoprop as it was when added simultaneously (Table 7). This rapid detoxification of ethoprop suggests that degradation is dependent on agents, most likely bacteria (13,14), that are mitigated by steam sterilization. Efficacy of ethoprop declined over time. Seventeen months after the last application of ethoprop, the number of *M. chitwoodi* that survived in the ethoprop-treated exposed soil was less than in the untreated soil and greater than in ethoprop-treated sterilized soil (Table 8). After 18 months no difference was observed between the ethoprop-treated exposed soil and the ethoprop-treated sterilized soil. The declined biodegradation property of soil with ethoprop history over a period of time might be responsible for the variable behavior of ethoprop in degradation studies reported by others (14).

Our data suggest that ethoprop should not be applied more than once every 2 years. Since potatoes generally are cropped once every 4 years, enhanced biodegradation of ethoprop does not appear to pose a serious problem under current potato production systems in the Pacific Northwest.

Literature Cited

1. Brodie, B. B. 1971. Differential vertical movement of nonvolatile nematicides in soil. Journal of Nematology 3:292–295.

2. Bunt, J. A. 1979. Effect and mode of action of the nematicide ethoprophos. Mededelingen van de Rijksfakulteit der Landbouwwetenschappen te. Gent 44:357–366.

3. Byrd, D. W., Jr., T. Kirkpatrick, and K. R. Barker. 1983. An improved technique for clearing and staining plant tissue for detection of nematodes. Journal of Nematology 15:142–143.

4. Charchar, J. M. 1987. Effect of temperature on the life cycle of *Meloidogyne chitwoodi* races 1 and 2 and *M. hapla* on Russet Burbank potato. Ph.D. dissertation, Washington State University, Pullman.

5. Finney, D. J. 1952. Probit analysis, a statistical treatment of the sigmoid response curve. Cambridge, England: Cambridge University Press.

6. Hussey, R. S., and K. R. Barker. 1973. A comparison of methods of collecting inocula of *Meloidogyne* spp., including a new technique. Plant Disease 57:1025–1028.

7. Huvar, A. J. 1969. The persistence, movement and loss of porophos (Mocap) in the soil. Pesticide development report, Mobil Chemical Company, Industrial Chemical Division, Research and Development Department, Ashland, VA.

8. Mojtahedi, H., G. S. Santo, J. N. Pinkerton, and J. H. Wilson. 1988. Effect of ethoprop on *Meloidogyne chitwoodi* and *M. hapla.* Journal of Nematology 20:649 (Abstr.).

9. Mojtahedi, H., R. E. Ingham, G. S. Santo, J. N. Pinkerton, G. L. Reed, and J. H. Wilson. 1989. Seasonal migration of *Meloidogyne chitwoodi* and its role in potato production. Journal of Nematology 23:162–169.

10. Nordmeyer, D., J. R. Rich, and D. W. Dickson. 1982. Effect of ethoprop, carbofuran and aldicarb on flue-cured tobacco infected with three species of *Meloidogyne.* Nematropica 12:199–204.

11. Pinkerton, J. N., G. S. Santo, R. P. Ponti, and J. H. Wilson. 1986. Control of *Meloidogyne chitwoodi* in commercially grown Russet Burbank potatoes. Plant Disease 70:860–863.

12. Pinkerton, J. N., H. Mojtahedi, G. S. Santo, and J. H. O'Bannon. 1987. Vertical migration of *Meloidogyne chitwoodi* and *M. hapla* under controlled temperature. Journal of Nematology 19:152–157.

13. Racke, K. D., and J. R. Coats. 1987. Enhanced degradation of isofenphos by soil microorganisms. Journal of Agricultural and Food Chemistry 35:94–99.

14. Racke, K. D., and J. R. Coats. 1990. Enhanced biodegradation of insecticides in midwestern corn soils. Pp. 68–81 *in* K. D. Rake and J. R. Coats, eds. En-

hanced biodegradation of pesticides in the environment. American Chemical Society Symposium Series 426. Washington DC.

15. Santo, G. S., J. H. O'Bannon, A. P. Nyczepir, and R. P. Ponti. 1981. Ecology and control of root-knot nematodes on potato. Proceedings of the 20th Annual Washington State Potato Conference; 3–5 February; Washington Potato Commission, Moses Lake, WA. Pp. 135–139.

16. Santo, G. S., R. P. Ponti, and J. H. Wilson. 1985. Control of *Meloidogyne chitwoodi* on potato with soil fumigants alone and in combination with nonfumigants, 1983. American Phytopathological Society Fungicide and Nematicide Tests 40:107.

17. Santo, G. S., H. Mojtahedi, and J. H. Wilson. 1988. Biology and control of root-knot nematodes on potatoes, 1987. Proceedings of the 27th Annual Washington State Potato Conference; 2–4 February; Washington Potato Commission, Moses Lake, WA. Pp. 67–70.

18. Santo, G. S., H. Mojtahedi, J. H. Wilson, and R. E. Ingham. 1989. Population dynamics and control of the Columbia root-knot nematode on potato. Proceedings of the 28th Annual Washington State Potato Conference; 31 January–2 February; Washington Potato Commission, Moses Lake, WA. Pp. 111–116.

19. Smelt, J. H., M. Liestra, and S. Voerman. 1977. Movement and rate of decomposition of ethoprophos in soil columns under field conditions. Pesticide Science 8:147–151.

20. Smelt, J. H., S. J. H. Crum, W. Teunissen, and M. Liestra. 1987. Accelerated transformation of aldicarb, oxamyl and ethoprophos after repeated soil treatments. Crop Protection 6:295–303.

21. Vrain, T. C. 1977. Technique for the collection of larvae of *Meloidogyne* spp. and a comparison of eggs and larvae as inocula. Journal of Nematology 9:249–251.