Limonene-1,2-Epoxide Hydrolase from *Rhodococcus erythropolis* DCL14 Belongs to a Novel Class of Epoxide Hydrolases

MARIËT J. VAN DER WERF,* KARIN M. OVERKAMP, AND JAN A. M. DE BONT

Division of Industrial Microbiology, Department of Food Technology and Nutritional Sciences, Wageningen Agricultural University, 6700 EV Wageningen, The Netherlands

Received 20 May 1998/Accepted 23 July 1998

An epoxide hydrolase from *Rhodococcus erythropolis* DCL14 catalyzes the hydrolysis of limonene-1,2-epoxide to limonene-1,2-diol. The enzyme is induced when *R. erythropolis* is grown on monoterpenes, reflecting its role in the limonene degradation pathway of this microorganism. Limonene-1,2-epoxide hydrolase was purified to homogeneity. It is a monomeric cytoplasmic enzyme of 17 kDa, and its N-terminal amino acid sequence was determined. No cofactor was required for activity of this colorless enzyme. Maximal enzyme activity was measured at pH 7 and 50°C. None of the tested inhibitors or metal ions inhibited limonene-1,2-epoxide hydrolase activity. Limonene-1,2-epoxide hydrolase has a narrow substrate range. Of the compounds tested, only limonene-1,2-epoxide, 1-methylcyclohexene oxide, cyclohexene oxide, and indene oxide were substrates. This report shows that limonene-1,2-epoxide hydrolase belongs to a new class of epoxide hydrolases based on (i) its low molecular mass, (ii) the absence of any significant homology between the partial amino acid sequence of limonene-1,2-epoxide hydrolase and amino acid sequences of known epoxide hydrolases, (iii) its pH profile, and (iv) the inability of 2-bromo-4-nitroacetophenone, diethylpyrocarbonate, 4-fluorochothalocline, and 1,10-phenanthroline to inhibit limonene-1,2-epoxide hydrolase activity.

Epoxides are highly reactive compounds which readily react with numerous biological compounds, including proteins and nucleic acids. Consequently, epoxides are cytotoxic, mutagenic, and potentially carcinogenic, and there is considerable interest in biological degradation mechanisms for these compounds.

In bacteria, epoxides are formed during the metabolism of alkenes (23) and halohydrins (15, 26, 34, 49). Enzymes belonging to a large number of enzyme classes, including dehydrogenases (17), lyases (21), carboxylases (1, 43), glutathione transferases (6, 8), isomerases (24), and hydrolases (7, 19, 44), are involved in the microbial degradation of epoxides.

Epoxide hydrolases are enzymes catalyzing the addition of water to epoxides forming the corresponding diol. This group of enzymes has been extensively studied in mammals, while only limited information is available on bacterial epoxide hydrolases. Three functions for epoxide hydrolases are recognized (42). In bacteria, epoxide hydrolases are involved in the degradation of several hydrocarbons, including 1,3-dihalo-2-propanol (34), 2,3-dihalo-1-propanol (15, 26), epichlorohydrin (46), propylene oxide (16), 9,10-epoxy fatty acids (30, 36), trans-2,3-epoxysuccinate (2), and cyclohexene oxide (14). Other epoxide hydrolases, such as microsomal and cytosolic epoxide hydrolase from mammals (for reviews, see references 4, 8, and 44), are involved in the detoxification of epoxides formed due to the action of P-450-dependent monooxygenases (8). Epoxide hydrolases are also involved in biosynthesis of hormones, such as leukotrienes and juvenile hormone (40, 45), and plant cuticular elements (11). Remarkably, the bacterial and eukaryotic epoxide hydrolases described so far form a homogeneous group of enzymes belonging to the α/β-hydrolase fold superfamily (10, 38).

*Corresponding author. Mailing address: Division of Industrial Microbiology, Department of Food Technology and Nutritional Sciences, Wageningen Agricultural University, P.O. Box 8129, 6700 EV Wageningen, The Netherlands. Phone: 31-317-484412. Fax: 31-317-484978. E-mail: mariert.vanderWerf@imb.ftms.wau.nl.

Materials and Methods

**Isolation of strain DCL14.** *R. erythropolis* DCL14 was isolated from an enrichment culture containing a sediment sample (10 g) from a ditch in Recuweit, The Netherlands, diluted in 30 ml of mineral salts medium (pH 7.0) (24) in the presence of 1 mM (−)-dihydrocarveol as the carbon and energy source. After incubation of this culture for 2 weeks on a shaker at 30°C and two successive transfers into fresh medium, samples of the enrichments were plated onto agar plates with mineral salts medium. These plates were incubated in a desiccator in which (+)-limonene was supplied via the gas phase. Colonies that developed were isolated and checked for purity by plating on yeast extract-glucose plates. *R. erythropolis* DCL14 (CIMW 0387B) is maintained at the Division of Industrial Microbiology, Wageningen, The Netherlands.

**Growth conditions.** *R. erythropolis* DCL14 was subcultured once a month and grown at 30°C on a yeast extract-glucose agar plate for 2 days, after which the plates were stored at room temperature. Cultures were grown in 5-liter Erlenmeyer flasks containing 1 liter of mineral salts medium with 0.01% (vol/vol) carbon source and fitted with rubber stoppers. The flasks were incubated at 30°C on a horizontal shaker oscillating at 1 Hz with an amplitude of 10 cm. After growth was observed, the concentration of the toxic substrates was increased with steps of 0.01% (vol/vol) until a total of 0.1% (vol/vol) carbon source had been added.

Cells for enzyme purification were grown fed-batch in a fermentor with a working volume of 2 liters at 28°C. (+)-Limonene was supplied via the gas phase by passing the airflow (300 ml/min) into the fermentor through a bubble column containing (+)-limonene. Every day, 1.5 liters of the culture was harvested, after which the working volume was increased to 2.0 liters. Cells were collected by centrifugation (4°C, 10 min at 16,000 × g) and washed with 50 mM potassium phosphate buffer (pH 7.0) and 0.1% (vol/vol) Tween 20. The pellet was resuspended in 7 ml of this buffer and stored at −20°C until used.

**Preparation of cell extract.** Two aliquots of frozen cell suspension (7 ml) were thawed and disrupted by sonication (20 min, 30% duty cycle, output control 23) with a Branson Sonifier 250. Cell debris was removed by centrifugation at 20,000 × g for 20 min. The supernatant was used as the cell extract. Protein was...
determined by the method of Bradford (12), with bovine serum albumin as the standard.

**Purification of limonene-1,2-epoxide hydrolase.** All purification steps were performed at 4°C and pH 7.0. If necessary, the pooled fractions were concentrated by ultrafiltration with an Amicon ultrafiltration unit using a membrane with a molecular weight cutoff of 10,000 under nitrogen at a pressure of 4 bar.

**Step 1: gel filtration.** The cell extract was applied onto a Sephacryl S300 (Pharmacia) column (2.5 by 98 cm) equilibrated with 10 mM potassium phosphate buffer (flow rate, 0.75 ml/min; collected fraction volume, 7.5 ml). Fractions containing limonene-1,2-epoxide hydrolase were pooled.

**Step 2: hydroxypatite.** The pooled fractions from the gel filtration step were applied to a hydroxypatite (Bio-Rad) column (5 by 6 cm) equilibrated with 10 mM potassium phosphate buffer (flow rate, 0.3 min/ml; collected fraction volume, 3 ml). The column was washed with 50 ml of the same buffer, and subsequently the enzyme was eluted with a 10 to 500 mM linear gradient of potassium phosphate (total volume, 400 ml). Limonene-1,2-epoxide hydrolase eluted at a potassium phosphate concentration of 100 mM. Active fractions were pooled.

**Step 3: anion-exchange chromatography.** The pooled fractions from the hydroxypatite step were applied onto a DEAE-Phosphor CL-6B (Pharmacia) column (2.5 by 31 cm) equilibrated with 25 mM potassium phosphate buffer. The column was washed with 100 ml of the same buffer (flow rate, 0.75 ml/min; collected fraction volume, 7.5 ml), and the enzyme was eluted with a 0 to 1 M linear gradient of NaCl in the same buffer (total volume, 1 liter). Limonene-1,2-epoxide hydrolase eluted at an NaCl concentration of 260 mM. Fractions exhibiting limonene-1,2-epoxide hydrolase activity were pooled and concentrated.

**Determination of molecular weight.** The molecular weight of the denatured protein was determined by sodium dodecyl sulfate-polyacrylamide gel electrophoresis (SDS-PAGE). An SDS–20% polyacrylamide gel was prepared by the method of Laemmli (28). Proteins were stained with Coomassie brilliant blue G. A Pharmacia low-molecular-weight calibration kit containing phosphorylase b (94,000), bovine serum albumin (67,000), ovalbumin (43,000), carbonic anhydrase (30,000), soybean trypsin inhibitor (20,100), and -lactalbumin (14,400) was used for the estimation of the molecular weight.

**RESULTS**

**Induction of limonene-1,2-epoxide hydrolase activity in R. erythropolis.** R. erythropolis DCL14 was grown on different carbon sources, and the limonene-1,2-epoxide hydrolase activity was determined (Table 1). Growth on monoterpenes resulted in a 10- to 100-fold increase in limonene-1,2-epoxide hydrolase activity. Notably, growth on the (+)-isomer(s) of the terpenes resulted in a limonene-1,2-epoxide hydrolase activity higher than that found after growth of the cells on the (−)-isomer(s) of the same compound (Table 1). Cells for further experiments were grown on (+)-limonene.

**Purification of limonene-1,2-epoxide hydrolase.** The limonene-1,2-epoxide hydrolase activity was present in the 100,000 × g supernatant of cell extract. Storage of cell extract at room temperature for 1 month did not result in any apparent loss of limonene-1,2-epoxide hydrolase activity. The purification scheme for limonene-1,2-epoxide hydrolase is presented in Table 2. Limonene-1,2-epoxide hydrolase was purified 53-fold, with an overall yield of 60%. From Table 2, it

<table>
<thead>
<tr>
<th>Growth substrate</th>
<th>Sp act* (nmol · min⁻¹ · mg [of protein]⁻¹)</th>
</tr>
</thead>
<tbody>
<tr>
<td>(+)-Limonene</td>
<td>795</td>
</tr>
<tr>
<td>(−)-Limonene</td>
<td>185</td>
</tr>
<tr>
<td>(+)-Limonene-1,2-epoxide</td>
<td>705</td>
</tr>
<tr>
<td>(−)-Limonene-1,2-epoxide</td>
<td>315</td>
</tr>
<tr>
<td>(+)-Limonene-1,2-diol</td>
<td>1,450</td>
</tr>
<tr>
<td>(−)-Limonene-1,2-diol</td>
<td>300</td>
</tr>
<tr>
<td>(+)-Carvone</td>
<td>185</td>
</tr>
<tr>
<td>(−)-Carvone</td>
<td>150</td>
</tr>
<tr>
<td>Succinate</td>
<td>15</td>
</tr>
<tr>
<td>Ethanol</td>
<td>15</td>
</tr>
</tbody>
</table>

*Activity with (+)-limonene-1,2-epoxide.

**TABLE 2. Purification of limonene-1,2-epoxide hydrolase from (+)-limonene-grown cells of R. erythropolis DCL14**

<table>
<thead>
<tr>
<th>Purification step</th>
<th>Total protein (mg)</th>
<th>Sp act* (nmol · min⁻¹ · mg [of protein]⁻¹)</th>
<th>Purification (fold)</th>
<th>Yield (%)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Cell extract</td>
<td>248</td>
<td>1,590</td>
<td>1</td>
<td>100</td>
</tr>
<tr>
<td>Gel filtration</td>
<td>17.8</td>
<td>21,800</td>
<td>13.6</td>
<td>98</td>
</tr>
<tr>
<td>Hydroxypatite</td>
<td>3.94</td>
<td>72,900</td>
<td>45.5</td>
<td>72</td>
</tr>
<tr>
<td>Anion exchange</td>
<td>2.75</td>
<td>85,100</td>
<td>53.2</td>
<td>59</td>
</tr>
</tbody>
</table>

*Activity with (+)-limonene-1,2-epoxide.
can be calculated that limonene-1,2-epoxide hydrolase represents 2% of the total soluble cellular protein of (+)-limonene-grown cells. The absorption spectrum of this colorless protein gave no indication of the presence of a prosthetic group. The results of the ICP-MS analysis indicate that limonene-1,2-epoxide hydrolase does not contain a metal ion as a cofactor. Limonene-1,2-epoxide hydrolase could be stored at 220°C for 6 months without loss of activity.

SDS-PAGE revealed one distinct band, corresponding to a protein with a molecular mass of 17 kDa (Fig. 2). Activity determinations with this protein electroeluted from a nondeaturing polyacrylamide gel revealed that the stained protein band indeed represented limonene-1,2-epoxide hydrolase (not shown). Gel filtration revealed a molecular mass of about 15 kDa, indicating that the native enzyme is a monomer.

N-terminal amino acid sequence. The N-terminal amino acid sequence of limonene-1,2-epoxide hydrolase was determined to be Thr-Ser-Lys-Ile-Glu-Gln-Pro-Arg-Trp-Ala-Ser-Lys-Asp-Ser-Ala-Ala-Gly-Ala-Ala-Ser-Thr-Pro-Asp-Glu-Lys-Ile-Val-Leu-Glu-Phe-Met-Asp-Ala-Leu-Thr-Ser-Asp-Ala-Ala-Lys-Leu-Ile-Glu-Tyr-Phe-Ala-Glu-Asp-Thr. Comparison of this amino acid sequence with entries in the databases by using the BLAST search program revealed no substantial homology with any other protein. The highest similarity (38%) was with checkpoint protein RAD24 from Saccharomyces cerevisiae (SWISS-PROT accession no. P32641).

Temperature and pH optimum and temperature stability of the purified enzyme. The enzyme has a very broad pH optimum, peaking at around pH 7 (Fig. 3). Phosphate buffer seemed to slightly inhibit limonene-1,2-epoxide hydrolase activity. Below pH 5 the activity could no longer be measured accurately, as chemical hydrolysis of the substrate overtook the biological hydrolysis rate. The temperature optimum of the enzyme was 50°C (Fig. 4). From the data presented in Fig. 4, we calculated an activation energy of 51.8 kJ/mol for limonene-1,2-epoxide hydrolase. The temperature stability of limonene-1,2-epoxide hydrolase is shown in Fig. 5. At temperatures over 45°C, rapid inactivation of the enzyme was observed.

Inhibitors and metal ions. We tested a variety of enzyme inhibitors for the ability to inhibit limonene-1,2-epoxide hydrolase activity: EDTA, 1,10-phenanthroline, a,a’-dipyridyl, nitritolriacetate, SDS, iodoacetamide, iodoacetate, p-chloromercuribenzoate, dithiothreitol, cysteine, 2-mercaptoethanol, glutathione and phenylhydrazine, hydroxylamine, potassium cyanate, N-ethylmaleimide, semicarbazide, diethylpyrocarbonate, 2-bromo-4-nitroacetophenone, and 2-bromo-4-methylacetophenone (all at 1 mM). None of these compounds inhibited limonene-1,2-epoxide hydrolase activity, nor did 4-fluorochalcone oxide and 4-phenylchalcone oxide, which are competitive inhibitors of mammalian soluble epoxide hydrolase (33).

The metal salts CuSO4, CoSO4, BaCl2, NiCl2, CaCl2, CdCl2, HgCl2, AgNO3, ZnCl2, MgSO4, MnSO4, Fe(II)SO4, and Fe(III)3 (SO4)2 (all at 1 mM) and NaCl, KCl, NH4Cl, CsCl, and LiCl (all at 10 mM) did not affect limonene-1,2-epoxide hydrolase activity.

Substrate specificity. Limonene-1,2-epoxide hydrolase has a rather narrow substrate specificity. Of the compounds tested, only limonene-1,2-epoxide, 1-methylcyclohexene oxide, cyclohexene oxide, and indene oxide were substrates for limonene-1,2-epoxide hydrolase (Table 3). The enzyme showed the highest activity with (+)-limonene-1,2-epoxide.

DISCUSSION

This report describes the purification and characterization of a novel epoxide hydrolase from R. erythropolis DCL14. Limonene-1,2-epoxide hydrolase is induced when R. erythropolis DCL14 is grown on monoterpenes (Table 1), reflecting its function in the limonene degradation pathway of the bacte-
Epoxide hydrolases form a remarkably homogeneous group of enzymes. They belong to the α/β-hydrolase fold superfamily (10, 38), based on the observation that they show low but significant sequence similarity with haloalkane dehalogenase from *Xanthobacter autotrophicus* GJ10, of which the three-dimensional structure has been solved (49). Epoxide hydrolases do not contain a prosthetic group. Their catalytic activity depends on a catalytic triad consisting of Asp, His, and Asp(Glu) residues (5, 38). On nucleophilic attack by the catalytic Asp the epoxide ring opens, resulting in the formation of a covalent hydroxy ester intermediate (3). This covalent intermediate was recently visualized in elegant studies by Müller et al. (32). Subsequently, a proton is abstracted from a water molecule by the His-Asp(Glu) pair, resulting in hydrolysis of the ester bond and in release of a corresponding diol.

In bacteria, only soluble epoxide hydrolases have been found (25, 35), while in eukaryotes, α/β-hydrolase folded epoxide hydrolases are either soluble or membrane bound (9, 11, 44). Bacterial epoxide hydrolases have a subunit mass of 35 to 40 kDa, and both homodimeric and monomeric enzymes have been described (25, 31, 35). Plant epoxide hydrolases have similar subunit masses (32 to 40 kDa [11, 27, 41]) and the only homodimeric enzyme is a homodimer (11). Soluble epoxide hydrolases from mammals are homodimers and are much larger than bacterial epoxide hydrolases (subunit mass of 55 to 62 kDa [44]). Mammalian membrane-bound epoxide hydrolases are thought to be dodecamers with a subunit mass of 46 to 53 kDa (8).

Two epoxide hydrolases which do not belong to the α/β-hydrolase fold superfamily have been described (10, 44): leukotriene A₄ hydrolase and cholesterol-epoxide hydrolase.

Leukotriene A₄ hydrolase is an enzyme involved in the production of the hormone derivatives of arachidonic acid. This cytosolic enzyme has been purified from several mammalian sources and is a monomer of 68 to 70 kDa (44). The amino acid sequence of leukotriene A₄ hydrolase revealed that this enzyme belongs to the metallohydrolase superfamily (29). It contains 1 mol of catalytic Zn²⁺ per mol of enzyme (22). Remarkably, leukotriene A₄ hydrolase is also a highly efficient aminopeptidase (37).

Cholesterol-epoxide hydrolase is an enzyme involved in the conversion of epoxides arising from cholesterol during lipid peroxidation in mammals. It is a membrane-bound protein which has not yet been purified to homogeneity (32). The molecular mass of this enzyme is unknown. In contrast to the α/β-epoxide hydrolases, there is no evidence that a covalent intermediate is formed in the course of epoxide hydrolysis, suggesting a completely different reaction mechanism (32).

Limonene-1,2-epoxide hydrolase from *R. erythropolis* DCL14 clearly belongs to a separate class of epoxide hydrolases. It is a cytoplasmic enzyme with an unprecedented low molecular mass of 17 kDa (Fig. 2). This low mass distinctly sets this enzyme apart from the α/β-hydrolase fold class of epoxide hydrolases. It is estimated that a minimal molecular mass of 25 kDa is necessary to accommodate the reaction mechanism as used by the α/β-hydrolase folded epoxide hydrolases (2a).

The N-terminal amino acid sequence of limonene-1,2-epoxide hydrolase does not show homology with known amino acid sequences of α/β-hydrolase folded epoxide hydrolases (10, 38). At first sight, α/β-hydrolase folded epoxide hydrolases do not show any obvious sequence similarity (10). However, several regions which are highly conserved have been identified in the amino acid sequences of α/β-hydrolase folded epoxide hydrolases (10). There was also no homology between the partial amino acid sequence of limonene-1,2-epoxide hydrolase and the amino acid sequence of leukotriene A₄ hydrolase (29). Remarkably, no substantial homology of limonene-1,2-epoxide hydrolase with any other protein present in the databases was detected.

The imidazole-modifying compounds 2-bromo-4'-nitroacetophenone and diethylpyrocarbonate did not affect limonene-1,2-epoxide hydrolase activity. This suggests that unlike in the α/β-hydrolase folded epoxide hydrolases, no catalytic histidine is involved in limonene-1,2-epoxide hydrolase (5, 18). The involvement of histidine in the mechanism of α/β-folded epoxide hydrolases has also been deduced from the slope of the pH optimum of this enzyme (9). Limonene-1,2-epoxide hydrolase behaved differently in this respect, and the activity remained constant at pH values of >8 (Fig. 3). Also 4-fluoro- and 4-phenylchalcone oxide, compounds which selectively inhibit soluble α/β-hydrolase folded epoxide hydrolases (33), and 1,10-phenanthrol, an inhibitor of leukotriene A₄ hydrolase activity (22), did not inhibit limonene-1,2-epoxide hydrolase activity. These results with inhibitors indicate that limonene-1,2-epoxide hydrolase uses a reaction mechanism that is completely different from those used by known epoxide hydrolases.

In general, epoxides are hydrolyzed chemically under both acidic and basic conditions (13). However, limonene-1,2-epoxide is much more stable under basic than under neutral conditions (unpublished results). An overall base-catalyzed reaction mechanism, as used by the α/β-folded epoxide hydrolases, would therefore be unfavorable for the hydrolysis of limonene-1,2-epoxide. In this respect, it seems understandable that *R. erythropolis* DCL14 has evolved another type of epoxide hydrolase, in view of its specialized function in the limonene

---

**TABLE 3. Substrate specificity of limonene-1,2-epoxide hydrolase**

<table>
<thead>
<tr>
<th>Substrate</th>
<th>Relative activity a</th>
</tr>
</thead>
<tbody>
<tr>
<td>(+)-Limonene-1,2-epoxide</td>
<td>100</td>
</tr>
<tr>
<td>(-)-Limonene-1,2-epoxide</td>
<td>27</td>
</tr>
<tr>
<td>1-Methylcyclohexene oxide</td>
<td>47</td>
</tr>
<tr>
<td>Cyclohexene oxide</td>
<td>4.0</td>
</tr>
<tr>
<td>Indene</td>
<td>57</td>
</tr>
</tbody>
</table>

a Limonene-1,2-epoxide hydrolase showed less than 0.25% of the (+)-limonene-1,2-epoxide hydrolysis rate with (-)-α-pinene oxide, cyclopentene oxide, cyclooctene oxide, cyclododecene oxide, cyclohexene sulfide, 3-oxo-cyclohexene oxide, 1-oxaspir[2.5]pacte, styrene oxide, trans-β-methylstyrene oxide, cis-stilbene oxide, epichlorohydrin, cis-2,3-epoxybutane, and 1,2-epoxyhexane.

b Relative to 100% activity with (+)-limonene-1,2-epoxide.
degradation pathway (reference 46 and unpublished results). The specialized function of limonene-1,2-epoxide hydrolase in limonene degradation is also reflected by the narrow substrate range of this enzyme (Table 3), in contrast to the broad substrate specificity of other epoxide hydrolases (35, 38, 44).

In conclusion, based on (i) the molecular mass, (ii) the partial amino acid sequence, (iii) the pH profile, and (iv) the effect of different inhibitors on the activity of limonene-1,2-epoxide hydrolase, limonene-1,2-epoxide hydrolase from R. erythropolis DCL14 clearly belongs to a separate class of epoxide hydrolases.

ACKNOWLEDGMENTS

This work was supported by grant BIO4-CT95-0049 from the European Community.

We thank Martin de Wit for technical assistance. David Leak (Department of Biochemistry, Imperial College, London, England) is acknowledged for sending us a sample of 1-methylecyclohexene oxide. We thank Michael Arand (Institute of Toxicology, University of Mainz, Mainz, Germany) for carefully reading the manuscript and sending us a sample of 4-fluorochalcone oxide.

REFERENCES


6. Acknowledgments

This work was supported by grant BIO4-CT95-0049 from the European Community.

We thank Martin de Wit for technical assistance. David Leak (Department of Biochemistry, Imperial College, London, England) is acknowledged for sending us a sample of 1-methylecyclohexene oxide. We thank Michael Arand (Institute of Toxicology, University of Mainz, Mainz, Germany) for carefully reading the manuscript and sending us a sample of 4-fluorochalcone oxide.

ACKNOWLEDGMENTS

This work was supported by grant BIO4-CT95-0049 from the European Community.

We thank Martin de Wit for technical assistance. David Leak (Department of Biochemistry, Imperial College, London, England) is acknowledged for sending us a sample of 1-methylecyclohexene oxide. We thank Michael Arand (Institute of Toxicology, University of Mainz, Mainz, Germany) for carefully reading the manuscript and sending us a sample of 4-fluorochalcone oxide.

REFERENCES


6. Acknowledgments

This work was supported by grant BIO4-CT95-0049 from the European Community.

We thank Martin de Wit for technical assistance. David Leak (Department of Biochemistry, Imperial College, London, England) is acknowledged for sending us a sample of 1-methylecyclohexene oxide. We thank Michael Arand (Institute of Toxicology, University of Mainz, Mainz, Germany) for carefully reading the manuscript and sending us a sample of 4-fluorochalcone oxide.

ACKNOWLEDGMENTS

This work was supported by grant BIO4-CT95-0049 from the European Community.

We thank Martin de Wit for technical assistance. David Leak (Department of Biochemistry, Imperial College, London, England) is acknowledged for sending us a sample of 1-methylecyclohexene oxide. We thank Michael Arand (Institute of Toxicology, University of Mainz, Mainz, Germany) for carefully reading the manuscript and sending us a sample of 4-fluorochalcone oxide.

ACKNOWLEDGMENTS

This work was supported by grant BIO4-CT95-0049 from the European Community.

We thank Martin de Wit for technical assistance. David Leak (Department of Biochemistry, Imperial College, London, England) is acknowledged for sending us a sample of 1-methylecyclohexene oxide. We thank Michael Arand (Institute of Toxicology, University of Mainz, Mainz, Germany) for carefully reading the manuscript and sending us a sample of 4-fluorochalcone oxide.