Modulation of Cytochrome P450 Gene Expression and Arachidonic Acid Metabolism during Isoproterenol-Induced Cardiac Hypertrophy in Rats

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ABSTRACT:

Several cytochrome P450 (P450) enzymes have been identified in the heart, and their levels have been reported to be altered during cardiac hypertrophy. Moreover, there is a strong correlation between P450-mediated arachidonic acid metabolites and the pathogenesis of cardiac hypertrophy. Therefore, we investigated the effect of isoproterenol-induced cardiac hypertrophy on the expression of several P450 genes and their associated P450-derived metabolites of arachidonic acid. Cardiac hypertrophy was induced by seven daily i.p. injections of 5 mg/kg isoproterenol. Then the heart, lung, liver, and kidney were harvested, and the expression of different genes was determined by real-time polymerase chain reaction. Heart microsomal protein from control or isoproterenol-treated rats was incubated with 50 μM arachidonic acid, and arachidonic acid metabolites were determined by liquid chromatography-electron spray ionization-mass spectrometry. Our results show that isoproterenol treatment significantly increased the heart/body weight ratio and the hypertrophic markers. In addition, there was a significant induction of CYP1A1, CYP1B1, CYP4A3, and soluble epoxide hydrolase and a significant inhibition of CYP2C11 and CYP2E1 in the hypertrophied hearts as compared with the control. CYP1A1, CYP2E1, and CYP4A3 gene expression was induced in the kidney, and CYP4A3 was induced in the liver of isoproterenol-treated rats. Isoproterenol treatment significantly reduced 5,6-, 8,9-, 11,12-, and 14,15-epoxyeicosatrienoic acid formation and significantly increased their corresponding 8,9-, and 14,15-dihydroxyeicosatetraenoic acid and the 20-hydroxyeicosatetraenoic acid metabolite. In conclusion, isoproterenol-induced cardiac hypertrophy alters arachidonic acid metabolism and its associated P450 enzymes, suggesting their role in the development and/or progression of cardiac hypertrophy.

Heart failure affects more than 5 million people in North America, with about half a million of new cases every year (Zordoky and El-Kadi, 2008). Cardiac hypertrophy and fibrosis precede heart failure; therefore, research into the molecular basis of hypertrophy can be considered as research into the initial steps of heart failure (Ritter and Neyses, 2003). Cardiac hypertrophy can be defined macroscopically as a thickening of the ventricular wall and/or septum leading to alterations in chamber size and geometry, collectively called remodeling (Braunwald and Bristow, 2000). Hypertrophy can be considered as a compensatory mechanism that transiently balances the biochemical stress and optimizes cardiac pump function; however, prolonged hypertrophy is a significant risk factor for heart failure (Carreno et al., 2006).

The role of cytochrome P450 (P450) enzymes in cardiovascular health and disease is well established (Elbekai and El-Kadi, 2006; Zordoky and El-Kadi, 2008). P450 is a family of mono-oxygenases that is involved in the oxidative metabolism of a wide range of xenobiotics and endogenous substances (Elbekai and El-Kadi, 2006). Many studies have examined the expression of P450 enzymes in the heart (Elbekai and El-Kadi, 2006). In vivo, P450 enzymes have been reported in explanted human hearts (Thum and Borlak, 2000b; Delozier et al., 2007) and in the left ventricle of Sprague Dawley (SD) rats and spontaneously hypertensive rats (SHRs) (Thum and Borlak, 2002). At the intracellular level, the gene expression and protein activity of many P450 enzymes have been reported in cultured cardiomyocytes (Thum and Borlak, 2000a) and in the rat cardiomyoblast H9c2 cell line (Zordoky and El-Kadi, 2007).

P450 enzymes are considered one of the major metabolic pathways for the metabolism of arachidonic acid (AA) in addition to cyclooxygenases and lipoxygenases. In the presence of NADPH and oxygen, P450 metabolizes AA to epoxyeicosatrienoic acid (EET) and hydroxyeicosatetraenoic acid (HETE) metabolites. EETs can be either incorporated into membrane phospholipid pools or efficiently hydrolyzed by soluble epoxide hydrolase (sEH) to biologically less active dihydroxyeicosatetraenoic acids (DHETs) or secreted into the extracel-

ABBREVIATIONS: P450, cytochrome P450; SD, Sprague Dawley; SHR, spontaneously hypertensive rat; AA, arachidonic acid; EET, epoxyeicosatrienoic acid; HETE, hydroxyeicosatetraenoic acid; sEH, soluble epoxide hydrolase; DHET, dihydroxyeicosatetraenoic acid; PCR, polymerase chain reaction; LC, liquid chromatography; ESI, electron spray ionization; MS, mass spectrometry; HPLC, high-performance liquid chromatography; ANP, atrial natriuretic peptide; BNP, brain natriuretic peptide; SHHF, spontaneously hypertensive heart failure.
lular space (Imig et al., 2002). P450 epoxigenases are known to metabolize AA to four regioisomeric metabolites, 5,6-, 8,9-, 11,12-, and 14,15-EET, and by P450 to HETEs are regarded as important mediators in hypertension, cardiovascular disease, and inflammation (Roman, 2002).

The expression of several P450 genes identified in the heart has been reported to be altered during cardiac hypertrophy; however, there is a great discrepancy among various reports on P450 alterations during cardiac hypertrophy, likely because of differences in the etiology of hypertrophy, disease severity, species in question, and other underlying conditions (Zordoky and El-Kadi, 2008). In the present study, we investigated the expression of multiple P450 genes in the heart, lung, liver, and kidney of male SD rats during isoproterenol-induced cardiac hypertrophy. Furthermore, we investigated the effect of isoproterenol-induced cardiac hypertrophy on the formation of AA metabolites and to determine whether the changes in P450 and sEH gene expression have led to changes in AA metabolism. Our findings provide the first evidence for the cardiac-specific changes in P450 and sEH metabolism during isoproterenol-induced cardiac hypertrophy.

Materials and Methods

Materials. TRizol reagent was purchased from Invitrogen (Carlsbad, CA). High-Capacity cDNA Reverse Transcription Kit, SYBR Green SuperMix, and 96-well optical reaction plates with optical adhesive films were purchased from Applied Biosystems (Foster City, CA). Real-time PCR primers were synthesized by Integrated DNA Technologies, Inc. (Coralville, IA) according to previously published sequences. AA, isoproterenol, and 4-hydroxybenzoic acid (HBA) were obtained from Sigma-Aldrich (St. Louis, MO). Acetonitrile and water (HPLC grade) were purchased from EM Science (Gibbstown, NJ). Real-time PCR primers were incubated in the incubation buffer (5 mM magnesium chloride hexahydrate).

RNA Extraction and cDNA Synthesis. Total RNA from the frozen tissues was isolated using TRIzol reagent (Invitrogen) according to the manufacturer’s instructions and quantified by measuring the absorbance at 260 nm. RNA quality was determined by measuring the 260/280 ratio. Thereafter, first strand cDNA synthesis was performed using the High-Capacity cDNA reverse transcription kit (Applied Biosystems) according to the manufacturer’s instructions. Briefly, 1.5 μg of total RNA from each sample was added to a mix of 2.0 μl of 10× reverse transcriptase buffer, 0.8 μl of 25× dNTP mix (100 mM), 2.0 μl of 10× reverse transcriptase random primers, 1.0 μl of MultiScribe reverse transcriptase, and 3.2 μl of nuclelease-free water. The final reaction mix was kept at 25°C for 10 min, heated to 37°C for 120 min, heated for 85°C for 5 s, and finally cooled to 4°C.

Quantification by Real-Time PCR. Quantitative analysis of specific mRNA expression was performed by real-time PCR by subjecting the resulting cDNA to PCR amplification using 96-well optical reaction plates in the ABI Prism 7500 System (Applied Biosystems). The 25-μl reaction mix contained 0.1 μl of 10 μM forward primer and 0.1 μl of 10 μM reverse primer (40 nM final concentration of each primer), 12.5 μl of SYBR Green Universal Mastermix, 11.05 μl of nuclelease-free water, and 1.25 μl of cDNA sample. The primers used in the current study were chosen from previously published studies (Bleicher et al., 2001; Kalsotra et al., 2002; Wang et al., 2003; Hirase et al., 2005; Rollin et al., 2005; Baldwin et al., 2006) and are listed in Table 1. Assay controls were incorporated onto the same plate, namely, no-template controls to test for the contamination of any assay reagents. After sealing the plate with an optical adhesive cover, the thermocycling conditions were initiated at 95°C for 10 min, followed by 40 PCR cycles of denaturation at 95°C for 15 s and annealing/extension at 60°C for 1 min. Melting curve (dissociation stage) was performed by the end of each cycle to ascertain the specificity of the primers and the purity of the final PCR product.

Real-Time PCR Data Analysis. The real-time PCR data were analyzed using the relative gene expression (i.e., ΔΔCT) method, as described in Applied Biosystems User Bulletin No. 2 and explained further by Livak and Schmittgen (2001). Briefly, the data are presented as the -fold change in gene expression normalized to the endogenous reference gene (glyceraldehyde-3-phosphate dehydrogenase) and relative to a calibrator. The untreated control was used as the calibrator when the change of gene expression by isoproterenol was being studied, whereas the P450 of lowest expression was used as a calibrator to compare the expression of different P450 genes.

Microsomal Preparation and Incubation. Microsomal protein was prepared from the heart tissue as described previously (Barakat et al., 2001). Briefly, hearts were washed in ice-cold potassium chloride [1.15% (w/v)], cut into pieces, and homogenized separately in cold sucrose solution (1 g of tissue in 5 ml of 0.25 M sucrose). Microsomal protein from homogenized tissues was separated by differential ultracentrifugation. The final pellet was reconstituted in cold sucrose and stored at −80°C. Heart microsomal protein concentration was determined by the Lowry method using bovine serum albumin as a standard (Lowry et al., 1951). Heart microsomes (1 mg protein/ml) were incubated in the incubation buffer (5 mM magnesium chloride hexahydrate)

### Table 1

<table>
<thead>
<tr>
<th>Gene</th>
<th>Forward Primer</th>
<th>Reverse Primer</th>
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<tr>
<td>CYP1A1</td>
<td>CCAACGATGGTCGCCCTT</td>
<td>TGCCCACACCCAAAGAATAAGA</td>
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<tr>
<td>CYP1B1</td>
<td>GCTTCTTCTGGCAAGCAGA</td>
<td>GGAAGGAGGATATTGAGAGA</td>
</tr>
<tr>
<td>CYP2B1</td>
<td>ACCCTTCTGGCAGAGCATAAA</td>
<td>TGTTGCTAGCATGAGAAGTC</td>
</tr>
<tr>
<td>CYP2B2</td>
<td>CCATCCCTTGGATGATCCAA</td>
<td>AATTGGCCAGAATCTCCCAC</td>
</tr>
<tr>
<td>CYP2C11</td>
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<td>GTCGCTCAGCTGAGGAAAGA</td>
</tr>
<tr>
<td>CYP2E1</td>
<td>AAGGCTTCTTGTTGCTGAGA</td>
<td>AGAGAAGCTCAGTTGAGAA</td>
</tr>
<tr>
<td>CYP2J3</td>
<td>TGTTGCTACGGCAATTCCAC</td>
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</tr>
<tr>
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</tr>
<tr>
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</tr>
<tr>
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<td>CTGGCTCAGCTGAGGAAAGA</td>
<td>AGGTGCTGAGGATATAAG</td>
</tr>
<tr>
<td>CYP4F4</td>
<td>CAGGCTTAGCAAGGATATAAGC</td>
<td>AGGTGCTGAGGATATAAG</td>
</tr>
<tr>
<td>CYP4F5</td>
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<td>AGGTGCTGAGGATATAAG</td>
</tr>
<tr>
<td>sEH</td>
<td>CAGGCTTAGCAAGGATATAAGC</td>
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</tr>
<tr>
<td>ANP</td>
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</tr>
<tr>
<td>BNP</td>
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</tr>
<tr>
<td>GAPDH</td>
<td>CAGGCTTAGCAAGGATATAAGC</td>
<td>AGGTGCTGAGGATATAAG</td>
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Primers sequences used for real-time PCR reactions
dissolved in 0.5 M potassium phosphate buffer, pH 7.4) at 37°C in a shaking water bath (50 rpm). A pre-equilibration period of 5 min was performed. The reaction was initiated by the addition of 1 mM NADPH. AA was added to a final concentration of 50 μM and incubated for 30 min. The reaction was terminated by the addition of 600 μl of ice-cold acetonitrile followed by the internal standard, 4-hydroxybenzophenone. AA metabolites were extracted twice by 1 ml of ethyl acetate and dried using a speed vacuum (Thermo Fisher Scientific). The concentrations of these eicosanoids in the samples were calculated by comparing the ratios of peak heights with their corresponding standards.

Separation of Different AA Metabolites by LC-ESI-MS. Extracted AA metabolites were analyzed using LC-ESI-MS (Waters Micromass ZQ 4000 spectrometer; Waters, Milford, MA) method as described previously (Nithipatkikom et al., 2001). The mass spectrometer was operated in negative ionization mode with single-ion recorder acquisition. The nebulizer gas was obtained from an in-house high-purity nitrogen source. The temperature of the source was set at 150°C, and the voltages of the capillary and the cone were 3.51 kV and 25 V, respectively. The samples (10 μl) were separated on reverse phase C18 column (Kromasil, 250 × 3.2 mm) using linear gradient mobile phase system water/acetonitrile with 0.005% acetic acid as mobile phase at a flow rate of 0.2 ml/min. The mobile phase system started at 60% acetonitrile, linearly increased to 80% acetonitrile in 30 min, increased to 100% acetonitrile in 5 min, and held for 5 min. 4-Hydroxybenzophenone was used as an internal standard.

Statistical Analysis. Data are presented as mean ± S.E.M. Control and treatment measurements were compared using Student’s t test. Comparative gene expression across tissues was analyzed using a one-way analysis of variance followed by a Student-Newman-Keuls post hoc comparison. A result was considered statistically significant where p < 0.05.

Results

Expression of P450 Genes and sEH in the Heart, Lung, Kidney, and Liver. To examine the constitutive expression of various P450 genes and sEH in the heart, lung, liver, and kidney tissues of male SD rats, total RNA was isolated from different tissues, and different genes were determined by real-time PCR. The data were analyzed using the relative gene expression method. The data were normalized to the endogenous reference gene (glyceraldehyde-3-phosphate dehydrogenase) and relative to a calibrator. The gene of lowest expression in each tissue was used as a calibrator as described under Materials and Methods. Results are presented as mean ± S.E.M. (n = 6).

FIG. 1. Constitutive expression of P450 genes and sEH in different tissues. Total RNA was isolated from different tissues, and the relative expression of P450 genes and sEH in the heart (A), lung (B), liver (C), and kidney (D) was determined by reverse transcription followed by real-time PCR. The data were analyzed using the relative gene expression method. The data were normalized to the endogenous reference gene (glyceraldehyde-3-phosphate dehydrogenase) and relative to a calibrator. The gene of lowest expression in each tissue was used as a calibrator as described under Materials and Methods. Results are presented as mean ± S.E.M. (n = 6).
moderate to low expressed genes, about 290-, 65-, 17-, and 11-fold higher than the calibrator, respectively (Fig. 1B).

On the contrary to the heart and lung, all the examined genes were found to be constitutively expressed in the liver but at different levels. CYP1B1 was the lowest expressed gene and was considered as a calibrator. CYP2E1 and CYP2C11 were the most highly expressed genes, about 13,000- and 11,000-fold higher than the calibrator, respectively (Fig. 1C). sEH, CYP4A2, and CYP4A3 were also highly expressed, about 5000-, 4600-, and 2000-fold higher than the calibrator, respectively (Fig. 1C). CYP4F4, CYP2J3, CYP2B2, and CYP4A1 were moderately expressed genes, about 480-, 800-, 750-, and 350-fold higher than the calibrator, respectively (Fig. 1C). The constitutive expression of CYP2B1, CYP1A1, and CYP4F5 was low, about 35-, 10-, and 10-fold higher than the calibrator, respectively (Fig. 1C).

Similar to the liver, all the examined genes were found to be constitutively expressed at different levels in the kidney. CYP2B2 was the lowest expressed gene and was considered as the calibrator (Fig. 1D). CYP4A2 and CYP4A3 were the most highly expressed genes, about 50,000- and 24,000-fold higher than the calibrator, respectively. sEH and CYP2E1 were highly expressed genes, about 6000-fold higher than the calibrator (Fig. 1D). CYP2C11, CYP4A1, CYP4F4, CYP2J3, CYP1A1, and CYP1B1 were high to moderate expressed genes, about 750-, 360-, 210-, 200-, 150-, and 60-fold higher than the calibrator, respectively (Fig. 1D). The constitutive expression of CYP4F4 and CYP2B1 was very low, about 2- and 1.5-fold higher than the calibrator, respectively (Fig. 1D).

**Effect of Isoproterenol Treatment on Hypertrophic Markers and the Heart/Body Weight Ratio.** To investigate whether isoproterenol treatment caused cardiac hypertrophy in the treated SD rats, we measured the cardiac gene expression of the hypertrophic markers, atrial natriuretic peptide (ANP) and brain natriuretic peptide (BNP) relative to control rats. Our results showed that isoproterenol treatment caused statistically significant induction of both hypertrophic markers, ANP and BNP, by 10- and 3-fold, respectively (Fig. 2A). In addition, isoproterenol treatment caused a statistically significant increase in the heart/body weight ratio by about 30% (Fig. 2B).

**Effect of Isoproterenol Treatment on P450 Gene Expression.** To examine the effect of isoproterenol-induced cardiac hypertrophy on the expression of several P450 genes and sEH in different tissues, total RNA was extracted from the heart, lung, liver, and kidney tissues of both control and isoproterenol-treated rats. Thereafter, the expression of different genes was measured using reverse transcription followed by real-time PCR as described under Materials and Methods.

Figure 3 shows the relative expression of CYP2C11 and CYP2E1 in the different tissues and the changes in their expression during isoproterenol-induced cardiac hypertrophy. CYP2C11 is highly expressed in the liver, moderately expressed in the kidney, and of low expression in the heart and lung (Fig. 5A). Similarly, CYP2E1 is highly expressed in the liver, moderately expressed in the kidney and lung, and of low expression in the heart (Fig. 5B). Isoproterenol-induced cardiac hypertrophy caused a significant inhibition of CYP2C11 gene expression in the heart by 50% (Fig. 5C). Nevertheless, CYP2C11 gene expression did not change significantly in the other tissues (Fig. 5C). Similar to CYP2C11, isoproterenol treatment caused a significant inhibition of CYP2E1 gene expression in the heart by 80% (Fig. 5D). However, unlike CYP2C11, isoproterenol treatment caused a paradoxical induction of CYP2E1 in the kidney (Fig. 5D). There was no change in CYP2E1 gene expression in the liver or lung during isoproterenol-induced cardiac hypertrophy (Fig. 5D).

With regard to the CYP2B subfamily, CYP2B1 was highly expressed in the lung, about 100,000-fold higher than the heart (Fig. 4A). The liver expresses CYP2B1 to a moderate degree, whereas the constitutive expression of CYP2B1 in the heart and kidney was low (Fig. 4A). Unlike CYP2B1, CYP2B2 was highly expressed in the liver, about 10,000-fold higher than its expression in the other organs (Fig. 4B). Isoproterenol treatment did not cause any statistically significant change of CYP2B1 or CYP2B2 gene expression in all the examined tissues (Fig. 4, C and D).

Figure 5 shows the relative expression of CYP2C11 and CYP2E1 in the different tissues and the changes in their expression during isoproterenol-induced cardiac hypertrophy. CYP2C11 is highly expressed in the liver, moderately expressed in the kidney, and of low expression in the lung and heart (Fig. 5A). Similarly, CYP2E1 is highly expressed in the liver, moderately expressed in the kidney and lung, and of low expression in the heart (Fig. 5B). Isoproterenol-induced cardiac hypertrophy caused a significant inhibition of CYP2C11 gene expression in the heart by 50% (Fig. 5C). Nevertheless, CYP2C11 gene expression did not change significantly in the other tissues (Fig. 5C). Similar to CYP2C11, isoproterenol treatment caused a significant inhibition of CYP2E1 gene expression in the heart by 80% (Fig. 5D). However, unlike CYP2C11, isoproterenol treatment caused a paradoxical induction of CYP2E1 in the kidney (Fig. 5D). There was no change in CYP2E1 gene expression in the liver or lung during isoproterenol-induced cardiac hypertrophy (Fig. 5D).

With regard to CYP2J3, it was found to be highly expressed in the liver, moderately expressed in the kidney and lung, and of low expression in the heart (Fig. 6A). CYP4A1 is also highly expressed in the liver and kidney, of low expression in heart, but not constitutively expressed in the lung (Fig. 6B). Isoproterenol-induced cardiac hypertrophy did not cause any changes in the gene expression of either CYP2J3 or CYP4A1 in all the examined tissues (Fig. 6, C and D).
Figure 7 shows the relative gene expression of CYP4A2 and CYP4A3 enzymes. CYP4A2 was constitutively expressed in the liver and kidney but not in the heart or lung (Fig. 7A); however, CYP4A3 was constitutively expressed in the heart, liver, and kidney but not in the lung (Fig. 7B). Isoproterenol-induced cardiac hypertrophy did not cause any changes in the gene expression of CYP4A2 in the liver and kidney (Fig. 7C); nevertheless, it caused a significant induction of CYP4A3 in the heart, liver, and kidney by 2.4-, 1.6-, and 1.7-fold, respectively (Fig. 7D).

With regard to CYP4F subfamily, CYP4F4 and CYP4F5 were found to be expressed in all the examined tissues. CYP4F4 was highly expressed in the liver compared with the other examined tissues (Fig. 8A). On the other hand, CYP4F5 was found to be highly expressed in the lung, moderately expressed in the heart and kidney, and of low expression in the liver (Fig. 8B). The gene expression of CYP4F4 and CYP4F5 was not altered during isoproterenol-induced cardiac hypertrophy in all the examined tissues (Fig. 8, C and D).

Effect of Isoproterenol Treatment of sEH Gene Expression. sEH was found to be highly expressed in the liver, moderately
expressed in the kidney, and of low expression in the lung and heart (Fig. 9A). Isoproterenol treatment caused a significant induction of sEH gene expression in the heart by 1.8-fold (Fig. 9B). However, the gene expression of sEH was not significantly altered in the other tissues (Fig. 9B).

Separation of AA Metabolites Using the LC-ESI-MS Selected Ion Chromatogram. LC-ESI-MS has been used in this study for the separation and quantification of P450-derived metabolites of AA. The mass spectrometer was operated in negative ionization mode with single-ion recorder acquisition, where the most abundant ion corresponds to the \( m/z = [M-1]^- \). All four regioisomeric epoxygenoic acids (5,6-, 8,9-, 11,12-, and 14,15-EET) exhibited the most abundant ions, corresponding to \( m/z = 319 \) ion, whereas their corresponding DHET has \( m/z = 337 \) ion, and 20-HETE has \( m/z = 319 \) ion. The fact of having functional groups at different positions in the eicosanoid structure allowed successful separation that was achieved through the use of the reverse phase C18 HPLC column and linear gradient mobile phase. Using authentic standards, 14,15-, 11,12-, 8,9-, and 5,6-EET were found to be eluted at 26.33, 28.56, 29.38, and 30.09 min, respectively, whereas that of 20-HETE was eluted at 15.07 min.
The elution patterns of 14,15-, 11,12-, 8,9-, and 5,6-DHET were at 11.35, 12.58, 13.59 and 14.73 min, respectively.

Effect of Isoproterenol Treatment on AA Metabolism. To investigate the effect of isoproterenol treatment on the formation of AA metabolites, heart microsomes of either control or isoproterenol-treated rats were incubated with 50 μM AA for 30 min. Thereafter, AA metabolites were determined using LC-ESI-MS. Our results clearly show that 5,6-EET was the major metabolite produced in both control and isoproterenol-treated rats, followed by 8,9-, 14,15-, and 11,12-EET, respectively. In comparison with control animals, in microsomes of hypertrophied hearts, the formation of 5,6-, 8,9-, 11,12-, and 14,15-EET was significantly lower, by 34.20, 44.34, 50.84, and 44.17%, respectively (Fig. 10A).

We also measured levels of enzymatic hydroxylation of EET products, DHETs. As shown in Fig. 10B, the formation of 8,9- and 14,15-DHET was significantly higher, by 122.69 and 83.68%, respectively, compared with control. On the other hand, 5,6-and 11,12-DHET were not significantly altered.

To determine the effect of isoproterenol treatment on P450 α-hydroxylase activity, we determined the formation of 20-HETE in mi-
crosomes from control and hypertrophied rats. Isoproterenol treatment significantly increased the 20-HETE formation by 192% in comparison with the control group (Fig. 10C).

**Discussion**

Although cardiac hypertrophy is an important condition that usually precedes heart failure (Carreno et al., 2006), there are limited reports of altered expression of P450 genes during cardiac hypertrophy. The expression of P450 has been studied in SHRs as compared with SD rats (Thum and Borlak, 2002) and the expression of sEH has been studied in spontaneously hypertensive heart failure (SHHF) rats as compared with normal strains (Monti et al., 2008). Nevertheless, there are no data about the effect of experimentally induced cardiac hypertrophy on the expression of P450 and sEH genes and their associated P450-derived metabolites of AA. To the best of our knowledge, this work demonstrates the tissue-specific expression of many P450 genes and sEH in male SD rats.

Tissue-specific expression of P450 genes in the rat has been a subject of discrepancies (Zordoky and El-Kadi, 2007; Zordoky and El-Kadi, 2008). Several factors may have led to these discrepancies. The first factor is the use of a conventional PCR technique in most of these studies, which may be insensitive because of the low level of P450 expression, especially in extrahepatic tissues. Secondly, most of the previous studies focused on only one tissue without giving comparative information regarding the other tissues. Therefore, it was necessary to examine the expression of multiple P450 genes simultaneously in different organs by a sensitive technique, such as the real-time PCR technique. In the current study, we showed that CYP1A1, CYP1B1, CYP2B1, CYP2B2, CYP2C11, CYP2E1, CYP2J3, CYP4F4, CYP4F5, and sEH genes are constitutively expressed in all the examined tissues, whereas CYP4A1 and CYP4A3 genes are expressed in the heart, kidney, and liver but not in the lung, and the CYP4A2 gene was expressed in the liver and kidney only. The lung has the highest constitutive expression of CYP1A1, CYP1B1, CYP2B1, and CYP4F5 genes, and the liver and kidney have the highest expression of CYP4A2 and CYP4A3, whereas the liver has
the highest expression level of all other P450 genes and sEH. Generally, the heart has the lowest level of constitutive P450 expression except for CYP1B1 and CYP4F5, which are expressed in the heart at higher levels than those in the liver.

To induce cardiac hypertrophy experimentally, we used a regimen of seven daily i.p. injections of 5 mg/kg isoproterenol, which is known to induce cardiac hypertrophy without heart failure or blood pressure elevation (Kravalova et al., 2008). In the current study, isoproterenol treatment caused cardiac hypertrophy in SD rats as manifested by significant induction of the hypertrophic markers, ANP and BNP, and the increase of the heart/body weight ratio. In agreement with our results, isoproterenol infusion of 4 mg/kg/day for 7 days caused a significant increase of the heart to body weight and a significant induction of ANP (Masson et al., 1998). Similarly, BNP is known to be increased in various models of cardiac hypertrophy (Maggi et al., 1998); however, there is limited information about the effect of isoproterenol-induced cardiac hypertrophy on BNP gene expression.

CYP1A1 and CYP1B1 are found to be highly expressed in the heart at the constitutive level; therefore, their role in AA metabolism in the heart cannot be ignored. CYP1A1 has been shown to be involved in \( \omega \)-terminal HETE synthesis, whereas CYP1B1 can metabolize AA to both midchain HETEs and EETs (Choudhary et al., 2004). In accordance, it was very important to investigate the effect of isoproterenol-induced cardiac hypertrophy on the gene expression of CYP1A1 and CYP1B1 in the heart and in other tissues. In the current study, we found that CYP1A1 gene expression is induced in the heart and kidney in isoproterenol-treated rats but not in the lung or liver, whereas CYP1B1 gene expression was induced only in the hypertrophied heart. In agreement with our results, expression of CYP1A1 and CYP1B1 was significantly increased in left ventricular tissues of SHR as compared with normotensive SD rats (Thum and Borlak, 2002).

On the contrary to CYP1A1 and CYP1B1, CYP2C11 gene expression was significantly lower in the hypertrophied heart than the control hearts; however, its gene expression was not altered in the other tissues. CYP2C11 is an important epoxygenase enzyme that is involved in AA metabolism and EET synthesis (Ng et al., 2007). Unlike CYP2C11, CYP2E1 metabolizes AA to 18- and 19-HETEs (Laethem et al., 1993). Isoproterenol-induced cardiac hypertrophy caused a significant inhibition of CYP2E1 gene expression in the heart and a significant induction of its expression in kidney. On the contrary to this finding, CYP2E1 has been reported to be higher in SHR than in normotensive SD rats (Thum and Borlak, 2002).

Despite previous reports that showed increased expression of CYP2B1/2 and CYP2J3 in SHR as compared with normotensive SD rats (Thum and Borlak, 2002), our results showed that CYP2B1, CYP2B2, and CYP2J3 gene expression were not altered during isoproterenol-induced cardiac hypertrophy. The discrepancy between our results and that of Thum and Borlak is likely because of the strain differences. CYP4A3 was significantly induced in the heart, liver, and kidney in isoproterenol-treated rats as compared with the control. The premise of this observation emerges from the fact that CYP4A3 is an important enzyme involved in AA metabolism to 20-HETE (Wang et al., 1996, 1999; Roman, 2002; Choudhary et al., 2004; Elbekai and El-Kadi, 2006). Therefore, the increase in 20-HETE formation in the present work could be attributed to the increased expression of CYP1A1, CYP1B1, and CYP4A3.

In conclusion, isoproterenol-induced cardiac hypertrophy caused significant changes of several P450 and sEH gene expression, which is mostly specific to the heart. The overall balance of these changes has led to higher production of 20-HETE and lower production of EETs in the hypertrophied hearts. 20-HETE is known to be involved in many cardiovascular diseases; however, there is indirect evidence about its role in the development of cardiac hypertrophy (Lee et al., 2004; Chabova et al., 2007). On the other hand, EETs are reported to have cardioprotective effects mainly by inhibiting the activation of nuclear factor \( \kappa B \) (Campbell, 2000; Hirotani et al., 2002; Xu et al., 2006). Increased expression of CYP1A1, CYP2E1, and CYP4A3 in the kidney during isoproterenol-induced cardiac hypertrophy could also lead to altered P450-mediated AA metabolites, which play a critical role in the kidney function (Elbekai and El-Kadi, 2006). Therefore, the cardiac and renal P450 may play an important role in the development and/or progression of cardiac hypertrophy. However, more studies are needed to explore the mechanisms by which cardiac hypertrophy modulates P450 gene expression.

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