The nuclear receptor PPARβ/δ programs muscle glucose metabolism in cooperation with AMPK and MEF2

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To identify new gene regulatory pathways controlling skeletal muscle energy metabolism, comparative studies were conducted on muscle-specific transgenic mouse lines expressing the nuclear receptors peroxisome proliferator-activated receptor α (PPARα; muscle creatine kinase [MCK]-PPARα) or PPARβ/δ (MCK-PPARβ/δ). MCK-PPARβ/δ mice are known to have enhanced exercise performance, whereas MCK-PPARα mice perform at low levels. Transcriptional profiling revealed that the lactate dehydrogenase b (Ldhb)/Ldha gene expression ratio is increased in MCK-PPARβ/δ muscle, an isoenzyme shift that diverts pyruvate into the mitochondrion for the final steps of glucose oxidation. PPARβ/δ gain- and loss-of-function studies in skeletal myotubes demonstrated that PPARβ/δ, but not PPARα, interacts with the exercise-inducible kinase AMP-activated protein kinase (AMPK) to synergistically activate Ldhb gene transcription by cooperating with myocyte enhancer factor 2A (MEF2A) in a PPARβ/δ ligand-independent manner. MCK-PPARβ/δ muscle was shown to have high glycogen stores, increased levels of GLUT4, and augmented capacity for mitochondrial pyruvate oxidation, suggesting a broad reprogramming of glucose utilization pathways. Lastly, exercise studies demonstrated that MCK-PPARβ/δ mice persistently oxidized glucose compared with nontransgenic controls, while exhibiting supranormal performance. These results identify a transcriptional regulatory mechanism that increases capacity for muscle glucose utilization in a pattern that resembles the effects of exercise training.

[Keywords: muscle; exercise; nuclear receptors; glucose metabolism; gene regulation]

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Muscle performance and resistance to fatigue are determined, in part, by the capacity to burn the chief fuels—fatty acids (FAs) and glucose—in order to generate the ATP needed for consistent mechanical function [Coggan 1991; Holloszy et al. 1998; Burke and Hawley 1999; Hawley 2002, Hargreaves 2004]. The capacity for muscle glucose utilization is an important determinant of muscle fitness. Endurance training enhances insulin-dependent and -independent muscle glucose uptake and utilization [Ivy and Holloszy 1981; Richter et al. 1982; Hayashi et al. 1997; Holloszy 2005], resulting in improvements in whole-body insulin sensitivity [DeFronzo et al. 1985; Treadway et al. 1989, Goodyear et al. 1995; Wojtaszewski et al. 1997, 2000a]. Evidence is emerging that the effects of exercise training on muscle glucose uptake involve events downstream from the exercise-stimulated kinase AMP-activated protein kinase (AMPK) and related cellular signaling pathways [Witzczak et al. 2008]. Delineation of the molecular regulatory pathways involved in the beneficial effects of exercise on muscle glucose metabolism could yield novel therapeutic targets aimed at the prevention or treatment of obesity-related insulin resistance and its consequences.

The capacity of muscle to burn FAs is determined, in part, at the level of gene expression. A transcriptional regulatory circuit involved in the control of skeletal muscle FA utilization has been delineated. The peroxisome proliferator-activated receptors (PPARs) α and β [also known as δ], members of the nuclear receptor superfamily, have been shown to regulate genes involved in muscle FA uptake and catabolism [Desvergne and Wahli 1999]. The activity of the PPARs is regulated at multiple levels, including availability of activating ligands [endogenous FA/lipid moieties] and coactivators such as PPARγ coactivator-1α [PGC-1α] [Desvergne and Wahli 1999; Vega et al. 2000; Wende et al. 2007; Madrazo and Kelly 2008; Schupp and Lazar 2010]. Most of the work relevant to the control of cellular fuel...
metabolism by PPARs has focused on PPARα. PPARα activates transcription of genes involved in numerous steps of cellular FA uptake and oxidation in muscle, liver, and heart (Gullick et al. 1994; Leone et al. 1999; Finck et al. 2002, 2005; Madrazo and Kelly 2008; Montagner et al. 2011). Thus, activation of PPARα serves to program the skeletal myocyte for high-capacity FA burning.

PPARβ shares many gene targets with PPARα, including those involved in cellular FA utilization [Desvergne and Wahli 1999; Gilde et al. 2003; Huss and Kelly 2004; Montagner et al. 2011]. Surprisingly, however, transgenic mice with skeletal muscle-specific forced expression of either PPARα or PPARβ exhibit remarkably different phenotypes [Luquet et al. 2003; Wang et al. 2004; Finck et al. 2005], suggesting that the two structurally related nuclear receptors regulate a subset of unique downstream genes and biological functions. Specifically, muscle-specific PPARα [muscle creatine kinase [MCK]-PPARα] transgenic mice exhibit myocyte triacylglyceride (TAG) accumulation, high muscle FA oxidation [FAO] rates, glucose intolerance, and mild insulin resistance [Finck et al. 2005]. In striking contrast, muscle-specific PPARβ mice [MCK-PPARβ] develop many features of an exercise-trained phenotype (“marathon mice”), including increased endurance, increased mitochondrial capacity, an oxidative fiber type shift, and enhanced insulin sensitivity [Luquet et al. 2003; Wang et al. 2004].

The contrasting phenotypes of the MCK-PPAR lines, while clearly representing extremes due to genetic manipulation of transcription factor expression, afford a unique opportunity to identify new downstream gene regulatory mechanisms involved in the chronic control of muscle energy metabolism. Moreover, delineation of the mechanisms whereby PPARα and PPARβ regulate distinct gene targets is an important question for the nuclear receptor biology field. Therefore, we embarked on a study to compare and contrast the gene expression profiles and metabolic phenotypes of closely matched MCK-PPAR lines. Our results indicate that, in addition to controlling muscle FA metabolism, PPARβ activates a program involved in muscle glucose utilization in a pattern that is strikingly similar to the effects of exercise training. Specifically, PPARβ activates transcription of the gene encoding lactate dehydrogenase B [LDHB], which catalyzes a key nodal point for increasing capacity for glucose oxidation by diverting glucose and lactate into the formation of pyruvate for mitochondrial oxidation. This transcriptional regulatory mechanism, which is unique for PPARβ compared with PPARα, involves cooperation with the exercise-induced kinase AMPK and the muscle-enriched transcription factor myocyte enhancer factor 2 [MEF2].

Results

PPARβ and PPARα regulate distinct LDH isoenzyme shifts

MCK-PPARβ transgenic lines were established using the same strategy as that for MCK-PPARα lines generated previously [Finck et al. 2005]. Three independent MCK-PPARβ transgenic lines were established, with levels of expression ranging from high physiological [low expression [LE]] to supraphysiological [medium [ME] and high [HE]] expression [Supplemental Fig. 1A,B], corresponding to those of the MCK-PPARα lines [Finck et al. 2005]. The data shown here represent assessment of the HE lines unless indicated otherwise. Gene expression profiling studies conducted with RNA isolated from muscle of the MCK-PPARβ and MCK-PPARα mice, compared with corresponding nontransgenic littermate [NTG] controls, confirmed increased expression of many known PPAR target genes involved in cellular FAO in both lines [Supplemental Fig. 1C,D]. Exercise studies using a motorized treadmill confirmed that MCK-PPARβ mice ran longer distances than corresponding NTG controls using an endurance regimen [Supplemental Fig. 1E], consistent with the original observations by Wang et al. [2004] for the PPARα-VP16 transgenic line. In contrast, MCK-PPARα mice ran shorter distances than NTG controls [Supplemental Fig. 1E].

The gene expression profiling data sets were analyzed to identify metabolic genes that were differentially regulated in the two MCK lines. Comparative analysis identified several genes involved in glucose utilization pathways that were differentially regulated [Supplemental Fig. 2]. Of particular interest was the expression pattern of the Ldhb (or Ldh2) and Ldha (or Ldh1) genes. Ldhb gene expression was increased, whereas Ldha gene expression was modestly decreased, in MCK-PPARβ muscle, a pattern that was not observed for MCK-PPARα [Supplemental Fig. 2]. These results were of interest because the Ldhb isoenzyme favors the reaction that converts lactate to pyruvate, which in turn provides substrate for the mitochondrial TCA cycle (glucose oxidation), whereas LDHA favors the reverse reaction to produce lactate from pyruvate generated by glycolysis [Fig. 1A]. These findings were validated by quantitative RT–PCR (qRT–PCR) across several muscle types [Fig. 1B]. The expected LDH isoenzyme activity shifts were confirmed by activity gel studies [Fig. 1C, Supplemental Fig. 3; data not shown].

PPARβ cooperates with AMPK and MEF2 to activate Ldhb gene transcription

Evidence has emerged that AMPK serves as a transducer of exercise to increase muscle glucose uptake [Ren et al. 1994; Holmes et al. 1999; Ojuka et al. 2000; Holloszy 2005; Witzcak et al. 2008]. More recently, PPARβ and AMPKα were shown to cooperate in the transcriptional regulation of several known PPAR target genes, including uncoupling protein 3 [Ucp3] and lipoprotein lipase [Lpl] [Narkar et al. 2008]. To assess the requisite role of PPARβ in the control of Ldhb gene expression in the absence of overexpression and determine whether AMPK signaling is involved in this mechanism, PPARβ loss-of-function studies were conducted in wild-type mouse primary skeletal myotubes. The effects of PPARβ ligand (GW501516) and the AMPK activator AICAR were assessed alone and together in the presence or absence of PPARβ shRNA-mediated knockdown. Consistent with previous results [Narkar et al. 2008], GW501516 and AICAR cooperated to increase expression of the Ucp3 gene, a known PPAR target—an
To further define the mechanism involved in the activation of Ldhb gene expression by PPARβ, the Ldhb gene promoter region was screened for putative PPARβ occupation sites via chromatin immunoprecipitation (ChIP) in primary skeletal myotubes in which PPARβ was overexpressed. One region, defined by primers amplifying a segment spanning −1228 to −1078 base pairs (bp) upstream of the transcription start site, was specifically enriched by the PPARβ antibody (Supplemental Fig. 4). This region was notable for the lack of a consensus PPAR response element but the presence of a MEF2 recognition site (CTATTATAG), which is highly conserved in the rat (CTATTATAG) and human (ATAATTATAG) Ldhb gene promoter regions. Mef2a and Mef2c mRNA levels were modestly increased in MCK-PPARβ muscle, whereas Mef2a gene expression was decreased in MCK-PPARα muscle (Supplemental Fig. 5).

Additional ChIP experiments were conducted to determine whether PPARβ and MEF2 co-occupy the region of the Ldhb promoter containing the MEF2-binding site. Antibodies to both PPARβ and MEF2 precipitated this region of chromatin (Fig. 3A). As a control for specificity, ChIP experiments were also performed with a region of the Cpt1b gene promoter containing known PPAR and MEF2 response elements (Brandt et al. 1998; Baldan et al. 2004). Anti-PPARβ and anti-MEF2 immunoprecipitated the relevant region in the Cpt1b promoter. In contrast, anti-PPARβ, but not anti-MEF2, immunoprecipitated the PPAR-responsive region of the Ucp1 gene promoter, which lacks a MEF2 site (Fig. 3A). These results strongly suggest that MEF2 is involved in the PPARβ-mediated regulation of Ldhb gene transcription. Consistent with this conclusion, siRNA-mediated knockdown of Mef2a transcript in skeletal myotubes resulted in diminished expression of Ldhb and the known MEF2 target Sscl2a4 (GLUT4), but not Ldha (Fig. 3B).

A series of coimmunoprecipitation (co-IP) studies were conducted to determine whether AMPK participated directly in the PPARβ/MEF2A interaction and define the PPARβ versus PPARs specificity in this response. The α subunit of the AMPK complex was chosen for these experiments, given that this subunit has recently been shown to be involved in AMPK signaling in skeletal muscle (Narkar et al. 2008). HEK293 cells were cotransfected with expression vectors for PPARβ, PPARα, Flag-MEF2A, and/or Myc-AMPKα2. Anti-Flag was found to coimmunoprecipitate PPARβ and endogenous AMPKα1 and AMPKα2 (Fig. 4A). Using AMPKα1 as the immunoprecipitation target, PPARβ, but not PPARα, was pulled down (Fig. 4B). To determine the in vivo relevance of these interactions, co-IP studies with anti-PPARβ or anti-PPARα antibodies were conducted with extracts of gastrocnemius muscle from the MCK-PPAR lines. PPARβ, but not PPARα, interacted with endogenous AMPKα2 (Fig. 4C).

Lastly, to determine whether endogenous AMPK localizes to the region of the Ldhb promoter containing the MEF2-binding site, additional ChIP experiments were conducted in primary skeletal myotubes. Anti-AMPKα2 precipitated this region of chromatin in response to activation of AMPK (AICAR) but not in response to PPARβ ligand (Fig. 4D). Taken together, these results suggest that...
PPARβ, but not PPARα, interacts directly with activated AMPK to occupy the Ldhb promoter in cooperation with MEF2A.

To explore functional correlates of the PPARβ/MEF2/AMPK interaction, a series of cell cotransfection studies were conducted using a MEF2 reporter containing three MEF2-responsive DNA elements multimerized upstream of a thymidine kinase (tk) promoter driving a luciferase reporter, [MEF2MEF2A]tk-Luc [Zhu and Gulick 2004]. In the presence of MEF2A, [MEF2MEF2A]tk-Luc was not activated by either PPARβ or AMPKα2 alone, but when expressed together, synergistic activation was observed (Fig. 5A). PPARβ and AMPKα2 also synergized on a Gal4-MEF2A reporter, indicating that PPARβ can coactivate MEF2 with AMPK without binding DNA (Fig. 5B). In contrast, and as predicted by the binding studies, PPARβ did not activate either reporter with PPARα (Fig. 5A,B). These findings indicate that PPARβ is capable of activating Ldhb gene transcription via MEF2A in cooperation with AMPK, likely in a ligand-independent manner.

**MCK-PPARβ muscle is reprogrammed for increased capacity for glucose uptake, storage, and oxidation**

The capacity of muscle to import, store, and utilize glucose is an important determinant of endurance and sprint exercise performance. Therefore, we next sought to determine whether a broad program of muscle glucose metabolism was activated in MCK-PPARβ muscle. Pre-exercise muscle glycogen levels were significantly increased in MCK-PPARβ, but not MCK-PPARα, mice [Fig. 6A,B]. Glycogen levels were depleted after run to exhaustion in both groups [Fig. 6B], indicating that the MCK-PPARβ mice used the high glycogen stores as fuel during exercise. Levels of total cellular GLUT4 protein and plasma PPARβ, but not MCK-PPARα, mice [Fig. 6A,B]. Glycogen levels were depleted after run to exhaustion in both groups [Fig. 6B], indicating that the MCK-PPARβ mice used the high glycogen stores as fuel during exercise. Levels of total cellular GLUT4 protein and plasma

![Diagram](image1)

**Figure 2.** PPARβ activates and cooperates with AMPKα to activate Ldhb gene expression via a ligand-independent mechanism. (A) Results of qRT–PCR analysis for primary mouse myotubes after infection with adenovirus expressing specific or scrambled shRNAs as indicated. Forty-eight hours post-infection, myotubes were treated for 24 h with DMSO (vehicle), 0.5 μM GW501516 (GW), 1 mM AICAR, or GW + AICAR as indicated in the key. Values represent mean (±SEM) arbitrary units (AU) normalized (=1.0) to the value of DMSO-treated scrambled shRNA (n = 3). (*) P < 0.05 versus the corresponding scrambled shRNA; (†) P < 0.05 versus DMSO control. (Inset) Western blot analysis confirms an increase in p-AMPK levels in myotubes treated with AICAR for 30 min. [B, left] Results of Western blot analysis performed on extracts of gastrocnemius muscle isolated from NTG or MCK-PPARβ mice using p-AMPKα (Thr 172), AMPKα, p-ACC (Ser 79), or total ACC antibodies. [Right] Quantification of the p-AMPK/AMPK and p-ACC/ACC signal ratios (n = 8 mice in each group) normalized (=1.0) to the NTG control. (C, left) Results of Western blot analysis performed on gastrocnemius muscle extracts using CaMKKα, LKB1, and S6RP (control) antibodies. [Right] Quantification of the Western blot signals corrected to S6RP (n ≥ 3 mice in each group). Values represent mean (±SEM) shown as arbitrary units (AU) normalized (=1.0) to the value of NTG control. (*) P < 0.05 compared with NTG.

![Diagram](image2)

**Figure 3.** PPARβ and MEF2 bind to the Ldhb promoter. (A) The results of ChIP assays performed on primary mouse myotubes following infection with Ad-PPARβ. Schematic shows PCR primer set location (−1228 and −1078) and the putative MEF2-binding site relative to the Ldhb promoter transcription start site (= +1). [Left] Results of representative gel analysis showing relative binding (PCR results) to Ldhb, Cpt1b, and Ucp1 promoters and control (L32) promoter. Antibodies (or IgG control) are shown at the top. [Right] Graphs display mean SYBR Green-based quantification of ChIP normalized to IgG control (n = 3). (⁎) P < 0.05 versus IgG control. (B) Results of qRT–PCR analysis for primary mouse myotubes after transfection with Mef2α siRNAs or scrambled control siRNAs as indicated. Values represent mean (±SEM) shown as arbitrary units (AU) normalized (=1.0) to the value of control siRNAs (n = 4). (⁎) P < 0.05 versus control.
membrane-associated GLUT4 (Fig. 6C,D) were significantly increased in MCK-PPARβ muscle. In contrast, and as shown previously (Finck et al. 2005), GLUT4 levels were reduced in MCK-PPARα muscle (Fig. 6C). Consistent with these results, MCK-PPARα mice developed marked hyperglycemia during exercise, whereas blood glucose levels declined with exercise in MCK-PPARβ mice (Supplemental Table 1). Thus, PPARβ, but not PPARα, programs muscle for increased glucose uptake and storage.

A series of studies were next conducted to determine whether PPARβ-driven activation of Ldhb expression resulted in increased capacity for mitochondrial pyruvate oxidation in MCK-PPARβ muscle. As has been shown for an independent muscle-specific PPARβ-VP16 line (Wang et al. 2004), electron microscopy revealed an increase in mitochondrial volume density and mitochondrial DNA levels in MCK-PPARβ soleus muscle compared with NTG controls (Supplemental Fig. 6A,B). A similar increase in muscle mitochondrial volume density was also present in MCK-PPARα muscle, together with myocyte lipid droplet accumulation (Supplemental Fig. 6A,B). Respiration rates were determined in mitochondria isolated from the hindlimb of the MCK-PPAR lines and corresponding NTG controls using pyruvate as a substrate. Pyruvate-driven state 3 [maximal ADP-stimulated respiration] rates were significantly higher in MCK-PPARβ mitochondria compared with controls, an effect that was not seen with the MCK-PPARα mice (Fig. 7A). In contrast, and consistent with the increased expression of genes involved in FAO in both lines, palmitoyl carnitine-driven state 3 respiration rates were increased in both MCK-PPARβ and MCK-PPARα mice (Supplemental Fig. 6C).

To determine the effect of increasing the LDHB/LDHA ratio on cellular pyruvate oxidation, oxygen consumption rates (OCR) were measured in skeletal myotubes following knockdown of Ldhb expression. As predicted, increasing the LDHB/LDHA ratio stimulated OCRs under basal conditions and in the presence of the uncoupler FCCP (Fig. 7B). Importantly, these effects were dependent on the addition of pyruvate to the medium (Supplemental Fig. 7).

To assess the physiological impact of increased capacity for muscle glucose uptake and oxidation in MCK-PPARβ mice, additional exercise testing was conducted. Sprint exercise performance depends, in part, on high capacity for glucose utilization. Therefore, we hypothesized that the MCK-PPARβ mice would perform better than wild-type controls and MCK-PPARα mice on a high-intensity (“wind sprint”) exercise regimen protocol. Indeed, MCK-PPARβ mice ran longer distances than corresponding NTG controls on this protocol (Fig. 7C). In contrast, MCK-PPARα mice ran shorter distances than NTG controls. Strikingly,
despite running longer, levels of blood lactate following exercise were lower in MCK-PPAR\textsubscript{b} mice compared with NTG littermate controls, consistent with increased diversion of pyruvate into mitochondrial oxidation (Table 1). In stark contrast, lactate levels increased post-exercise in MCK-PPAR\textsubscript{a} mice (Table 1).

To further evaluate the muscle fuel utilization preference of MCK-PPAR\textsubscript{b} mice during exercise, respiratory exchange ratio (RER) and oxygen utilization (VO\textsubscript{2}) were measured during a run-to-exhaustion exercise protocol. Consistent with a shift to muscle glucose oxidation, the RER increased to \(\sim 1.0\) with exercise in both MCK-PPAR\textsubscript{b} and the NTG control group, indicative of a switch to carbohydrates as the chief fuel (Supplemental Fig. 8A). Despite exercising significantly longer [and consuming more oxygen] compared with the control group, the MCK-PPAR\textsubscript{b} mice maintained RER at \(\sim 1.0\) during the entire exercise period (Supplemental Fig. 8A). Interestingly, whereas the MCK-PPAR\textsubscript{b} mice consumed more...
oxygen during the exercise period (as reflected by an increase in $\Delta VO_{2\text{max}}$), the maximal peak oxygen consumption [$VO_{2\text{max}}$] was not different between the groups (Supplemental Fig. 8B,C). Taken together, these results suggest that PPAR$\beta$ activates a gene regulatory program that increases the coupling of muscle glycolysis to glucose oxidation, allowing for greater ATP generation per mole of glucose burned.

**Discussion**

The capacity to burn fuel is a key determinant of muscle fitness. Exercise training triggers an adaptive metabolic response in muscle, leading to increased fuel burning capacity and the flexibility to switch between the chief substrates: FAs and glucose [Holloszy and Coyle 1984]. In contrast, obesity and chronic disease, conditions that reduce physical activity, lead to a state in which the capacity of muscle to burn glucose is constrained [Mujika and Padilla 2001]. Delineation of the gene regulatory mechanisms involved in the adaptive and maladaptive programming of muscle fuel metabolism could unveil new therapeutic targets aimed at common metabolic diseases such as obesity-related insulin resistance and diabetes. Here, we show that the nuclear receptor PPAR$\beta$, but not PPAR$\alpha$, is capable of increasing capacity for muscle glucose oxidation by activating transcription of the $Ldhb$ gene through a unique mechanism, in cooperation with AMPK and the transcription factor MEF2.

We found that PPAR$\beta$ activates a program that increases the coupling of glycolysis to glucose oxidation in muscle. This response involves PPAR$\beta$-mediated induction of $Ldhb$ gene transcription and repression of $Ldha$ gene expression, resulting in an increase in the LDHB/LDHA isoenzyme ratio. The LDHB isoenzyme functions to convert lactate to pyruvate, LDHA favors the opposite reaction to produce lactate. Notably, exercise training is known to increase muscle LDHB/LDHA ratio, increasing capacity to fully catabolize glucose for maximal ATP production [Hittel et al. 2005]. Several lines of evidence presented here support the conclusion that PPAR$\beta$, but not PPAR$\alpha$, drives a gene regulatory program that increases capacity for muscle glucose oxidation. First, activity of LDH isoenzymes containing LDHB is selectively activated in muscle extracts prepared from MCK-PPAR$\beta$, but not MCK-PPAR$\alpha$, mice. Second, capacity for glucose uptake, glycogen storage, and mitochondrial pyruvate oxidation is increased in MCK-PPAR$\beta$ muscle compared with MCK-PPAR$\alpha$ muscle. Third, the MCK-PPAR$\beta$ mice maintain a high RER throughout a bout of exercise to exhaustion. Interestingly, despite running longer and thus using more oxygen during exercise, the MCK-PPAR$\beta$ mice did not exhibit an increased absolute VO$_{2\text{max}}$ compared with NTG controls. This interesting observation may reflect the persistent use of glucose (rather than fat) as an oxidative substrate, despite longer periods of exercise. Lastly, high-intensity exercise performance is increased and post-exercise blood lactate levels are decreased in MCK-PPAR$\beta$ mice compared with wild-type controls. In contrast, circulating lactate levels were abnormally increased in MCK-PPAR$\alpha$ mice following exercise.

We found that PPAR$\beta$ activates $Ldhb$ gene transcription via a novel mechanism for nuclear receptors, involving cooperation with AMPK and the transcription factor MEF2A. The observed role of MEF2 is of interest given its known role in the regulation of muscle energy metabolism [Michael et al. 2001; Naya et al. 2002]. Participation by AMPK in this mechanism is consistent with the observation that this kinase is activated by exercise [Wojtaszewski et al. 2000b] and has been shown to cooperate with PPAR$\beta$ [Narkar et al. 2008]. Moreover, emerging evidence indicates that activation of AMPK and related kinases is involved in muscle glucose uptake via a mechanism that is independent of insulin signaling, similar to the effects of exercise on glucose utilization [Witzczak et al. 2008]. We speculate that AMPK is a key trigger for the PPAR$\beta$-mediated activation of $Ldhb$ gene transcription. The mechanism whereby AMPK is activated in MCK-PPAR$\beta$ muscle was not fully delineated in this study. Among the known upstream activators of AMPK—LKB1 and CaMKK—we found only the latter to be induced in MCK-PPAR$\beta$ muscle. Specifically, CaMKK$\alpha$ protein levels were increased in MCK-PPAR$\beta$ muscle [CaMKK$\beta$ could not be detected, and therefore regulation could not be assessed].

The mechanisms whereby related nuclear receptors with similar DNA-binding domains regulate transcription of distinct target genes are poorly understood. Our results demonstrate that the $Ldhb$ gene is activated by PPAR$\beta$, but not by PPAR$\alpha$, via a trans-activation mechanism that involves interaction with the transcription factor MEF2A in the absence of a classical PPAR DNA recognition element. This unique mechanism appears to be PPAR ligand-independent, but requires AMPK. The observed cooperativity with AMPK is consistent with recent results by Narkar et al. (2008) demonstrating that AMPK interacts with PPAR$\beta$ to activate PPAR targets in muscle. However, in this previous study, AMPK/PPAR$\beta$ cooperativity was enhanced by ligand, in contrast to our results with the $Ldhb$ gene. It is tempting to speculate that in certain circumstances, the effects of AMPK or other signaling pathways can substitute for ligand in the

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**Table 1. Blood lactate levels in MCK-PPAR$\beta$ and MCK-PPAR$\alpha$ mice**

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<tr>
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<th>NTG</th>
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<tr>
<td>Run distance [m]</td>
<td>2086 ± 201</td>
<td>3559 ± 186$^a$</td>
<td>1883 ± 63</td>
<td>1451 ± 124$^a$</td>
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<tr>
<td>Baseline lactate levels [mmol/L]</td>
<td>3.9 ± 0.2</td>
<td>5.0 ± 1.1</td>
<td>2.9 ± 0.5</td>
<td>2.1 ± 0.2</td>
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<tr>
<td>Post-exercise lactate levels [mmol/L]</td>
<td>5.2 ± 0.7</td>
<td>3.9 ± 0.6$^a$</td>
<td>5.0 ± 1.1</td>
<td>9.0 ± 1.5$^a$</td>
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Values represent the mean (±SEM); n = 8–11 mice in each group. $^aP < 0.05$ versus NTG.
activation of a subset of PPARβ targets. Our results also suggest that the PPARβ versus PPARα specificity for activation of Ldhb gene transcription relates to specific interactions with AMPKα2 on the target promoter. The precise mechanisms involved in the activation of transcription via the MEF2A/PPARβ/AMPKα complex were not fully determined in this study. Recently, AMPK was shown to phosphorylate histone H2B, suggesting one possible mechanism [Bungard et al. 2010]. It is also possible that the cooperative interaction releases MEF2 from suppressive effects of repressors such as histone deacetylases [McGee et al. 2008]. Consistent with this latter notion, we showed that activation of AMPK in C2C12 myoblasts does indeed shift HDAC5 from nucleus to cytoplasm (data not shown). Future studies aimed at assessing changes in protein acetylation in the local vicinity of the PPARβ/AMPKα/MEF2A complex will likely require genome-wide chromatin surveys.

In conclusion, our results suggest a model in which activation of AMPK, such as occurs with exercise, triggers the assembly of a transcriptional activation complex containing AMPK and PPARβ tethered to MEF2A to regulate transcription of the Ldhb gene and likely a larger subset of metabolic gene targets. Further delineation of the mechanisms unveiled here could lead to strategies to develop selective PPAR-based metabolic modulators with pathway, or possibly even target, specificity.

Materials and methods

Generation of MCK-PPARβ transgenic mice

A DNA construct containing a 1.0-kb PPARβ cDNA was cloned downstream from the skeletal muscle MCK promoter (kind gift of E.N. Olson, University of Texas Southwestern). Transgenic mice were generated by microinjection of the MCK-PPARβ construct into fertilized one-cell C57BL/6 × CBA/J F1 embryos. Three independent lines were generated, exhibiting different levels of transgenic expression as measured by Northern and Western blot (Supplemental Fig. 1A). Unless specifically indicated, the results described here were generated using the high-expressing MCK-PPARβ line (HE), compared with the previously generated high-expressing MCK-PPARα line [Finck et al. 2005]. The animal data represent studies with MCK-PPAR mouse lines (hybrid strain, B6/CBA), with the following specific exceptions: MCK-PPARα mouse lines (hybrid strain, B6/CBA), with the following specific exceptions: MCK-PPARα mice in a C57BL/6 pure strain were used for immunoblotting (Figs. 2C, 4C, 6C), qRT–PCR (Fig. 1B, Supplemental Fig. 5), and LDH isoenzymes (Fig. 1C; Supplemental Fig. 3) experiments. NTG controls were used in all cases. Of note, the majority of the phenotypic characterization of these mice was performed in the hybrid strain [B6/CBA] for both MCK-PPARα and MCK-PPARβ. In addition, several gene expression readouts, including the regulation of Ldhb and Ldhb compared with corresponding nontransgenic controls, were similar in pure B6 backgrounds compared with the hybrid strain for both MCK-PPARα and MCK-PPARβ lines [data not shown].

Animal studies

Male and female MCK-PPAR mice and NTG controls (−25–30 g of body weight; 8–16 wk of age) were used for all studies. Animal studies were conducted in strict accordance with the NIH guidelines for humane treatment of animals.

Mitochondrial respiration studies

Skeletal muscle mitochondria were isolated from hindlimbs of MCK-PPAR mice and NTG controls as previously described [Zechner et al. 2010]. Respiration rates of the mitochondrial isolate containing 0.5 mg of protein were determined at 30°C using an optical probe (Oxygen FOXY Probe, Ocean Optics) in a 2-mL sealed, continuously stirred respiration chamber, as previously described [Leone et al. 2005]. Respiration was determined using pyruvate and palmitoyl carnitine as substrates. Following measurement of basal respiration, state 3 respiration was determined by exposing mitochondria to 1 mM ADP. Uncoupled respiration was evaluated following the addition of oligomycin (1 μg/mL) to inhibit ATP synthase. The solubility of oxygen in the respiration buffer at 30°C was taken as 230 nmol of oxygen per milliliter. Respiration rates were expressed as nanomoles of oxygen per minute per milligram of mitochondrial protein.

Exercise studies

Mice were acclimated (run for 9 min at 10 meters [m]/min followed by 1 min at 20 m/min) to the treadmill for two consecutive days prior to the experimental protocol.

Low-intensity exercise (endurance) Fed mice were run for 1 h at 10 m/min, followed by an increase of 2 m/min every 15 min until exhaustion (defined as remaining on the shock grid for five consecutive seconds).

High-intensity exercise (wind sprints) Fed mice were run for 1 min, alternating with rest for 2 min. Running intervals started at 10 m/min and increased 5 m/min each interval until a speed of 50 m/min was reached. Then, speed was increased 5 m/min every sixth interval until exhaustion.

Tail blood was taken before and after exercise and measured for glucose (B-GLUCOSE, Hemacue AB) and lactate (Lactate Pro Arkray). Tissue was dissected and immediately processed for glycogen analyses.

Peak VO2 and RER were determined as described previously [Calvo et al. 2008]. Briefly, 3-mo-old male mice were placed in an enclosed treadmill attached to the Comprehensive Laboratory Animal Monitoring System (CLAMS; Columbus Instruments) for 30 min at a 0° incline and 0 m/min. The mice were then challenged with 1.5-min intervals of increasing speed at a 15° incline. The increasing speeds used in the protocol were 10, 14, 18, 22, 26, 28, 30, 32, 34, 36, 38, 40, 42, 44, and 46 m/min. Measurements were collected before the exercise challenge, throughout the challenge, and following failure.

Glycogen measurements

Mouse gastrocnemius tissue was pulverized under liquid nitrogen and homogenized in a 0.3 M perchloric acid solution. The muscle extract was then assayed with and without amyloglucosidase digestion (Sigma Aldrich) in 50 mM sodium acetate (pH 5.5) and 0.02% BSA. Resulting changes in absorption at 340 nM were measured following the addition of oligomycin (1 μg/mL) to inhibit ATP synthase. The solubility of glucose in the respiration buffer at 30°C was taken as 230 nmol of oxygen per milliliter. Measurements were collected before the exercise challenge, throughout the challenge, and following failure.

Histologic analyses and electron microscopy

Mouse gastrocnemius was collected, immersed in Tissue-Tek O.C.T. Compound [Sakura Finetek USA, Inc.], and snap-frozen using Cryotool II [Richard-Allan Scientific] in a cryomold for sectioning. Sections were stained with Periodic Acid Schiff [PAS]
to detect glycogen accumulation. Electron microscopy was performed as previously described (Zechnier et al. 2010).

**Gene expression array studies**

Total RNA isolated from gastrocnemius muscle of 6-wk-old MCK-PPARα [HE] or MCK-PPARβ [HE] and NTG littermate mice was used for gene expression array studies as previously described [Huss et al. 2004]. The Alvin Siteman Cancer Center's Multiplexed Gene Analysis Core at Washington University School of Medicine performed hybridization to Affymetrix mouse MOE430A chips. Affymetrix MAS 5.0 software was used for initial analysis and background normalization. Probe sets called “absent” by MAS 5.0 in both NTG and MCK-PPAR were excluded. Two independent samples were analyzed. Signal intensity ratios were averaged from both samples and calculated as MCK-PPAR/NTG. A gene with a calculated fold change ≥1.5 was considered an up-regulated gene target in the MCK-PPAR transgenic, and a gene with a fold change of ≤0.5 was considered a down-regulated gene target. For pathway analysis, the filtered data sets were uploaded into GenMAPP software to review the biopaths using the Gene Ontology database. The gene array data discussed in this study have been deposited in NCBI's Gene Expression Omnibus (GEO) and are accessible through GEO series accession numbers GSE5777 [MCK-PPARα] and GSE29055 [MCK-PPARβ].

**RNA and genomic DNA analyses**

Total RNA was isolated from mouse skeletal muscle using the RNAzol method [Tel-Test]. Northern blot analysis was performed as previously described [Wende et al. 2005]. Real-time qRT-PCR was performed using the Stratagene MX3005P detection system and reagents supplied by Stratagene. Specific oligonucleotide primers for target gene sequences are listed in Supplemental Table 2. Arbitrary units of target mRNA were corrected to expression of 36b4.

Genomic/mitochondrial DNA was isolated using the RNAzol method, followed by back extraction with 4 M guanidine thiocyanate, 50 mM sodium citrate, and 1 M Tris and an alcohol precipitation. Mitochondrial DNA content was determined by SYBR Green analysis [Stratagene]. The levels of NADH dehydrogenase subunit 1 [mitochondrial DNA] were normalized to the levels of Lpl [genomic DNA]. The primer sequences are noted in Supplemental Table 2.

**Antibodies and Western immunoblotting studies**

Antibodies directed against PPARβ [K-20], PPARα [H-98], insulin receptor β subunit [InsRβ], and CaMKKα [R-73] were purchased from Santa Cruz Biotechnology. Anti-GLUT4 antibody was a gift of M. Mueckler (Washington University); antibodies directed against p-AMPKα (Thr172), AMPKα, p-ACC (Ser79), ACC, LKB1, and S6 ribosomal protein (S6R) were purchased from Cell Signaling Technology; anti-GAPDH antibody was purchased from Abcam; and anti-AMPKα2 antibody was purchased from R&D Systems.

Western immunoblotting studies were performed with whole gastrocnemius muscle lysates as previously described [Cresci et al. 1996]. Detection was performed by measuring the chemiluminescent signal as assayed by SuperSignal Ultra [Pierce]. Band intensities were quantified using the ChemiDoc (Bio-Rad) or FluorChemQ [Alpha Innotech].

**LDH isoenzyme analysis**

LDH isoenzyme patterns were determined as previously described (Salplachta and Necas 2000). Briefly, mouse gastrocnemius was homogenized in a solution of 0.9% NaCl and 5 mM Tris-HCl [pH 7.4], and the lysates were centrifuged for 30 min at 15,000g to remove the cellular debris. One-hundred micromicrograms of protein was loaded onto a 6% nondenaturing polyacrylamide gel. Following electrophoresis, the gel was placed in 10 mL of staining solution containing 0.1 M sodium lactate, 1.5 M NaCl, 0.1 M Tris-HCl [pH 8.6], 10 mM NaCl, 5 mM MgCl2, 0.03 mg/mL phenazinemethosulphate (PMS), and 0.25 mg/mL nitroblue tetrazolium [NBT]. Protein extracted from mouse heart served as a positive control.

**Preparation of subcellular membrane fractions from skeletal muscle**

Subcellular membrane fractions were prepared using a modification of the Hirshman-modified Grimditch fractionation technique [Hirshman et al. 1990]. Briefly, frozen mouse gastrocnemius was powdered in liquid nitrogen and homogenized in homogenization buffer [HB] [20 mM Tris, 1 mM EDTA, 255 mM sucrose at pH 7.4] at 4°C using a 1-mL dounce homogenizer. Homogenates were centrifuged at 16,800g for 20 min at 4°C. Intracellular membranes [IMs] were isolated from supernatants, and plasma membranes were isolated from the pellets. Pellets containing plasma membranes were resuspended in HB, followed by dounce homogenization, and were centrifuged at 16,800g for 20 min at 4°C. Pellets were resuspended in HB, layered on a 1.12 M sucrose cushion, and centrifuged in a TL-100 [Beckman Coulter] at 44,000 rpm for 20 min at 4°C. Plasma membranes at the interface between the sucrose cushion and buffer were collected, resuspended in HB, and centrifuged in a TL-100.3 [Beckman Coulter] at 50,000 rpm for 10 min at 4°C. Plasma membrane pellets were resuspended in SDS lysis buffer.

**Primary muscle cell culture**

Primary myoblasts [satellite cells] were isolated from wild-type mice as previously described [Rando and Blau 1994]. Briefly, mice were killed by CO2 inhalation, followed by cervical dislocation. Hindlimb muscles from both legs were removed. Minced tissue was digested in a collagenase/dispase/CaCl2 solution for 1.5 h at 37°C in a shaking bath. Dulbecco's modified Eagle's medium [DMEM] supplemented with 10% fetal bovine serum [FBS] [PAM] was added, and samples were triturated gently before loading onto a Netwell filter [70 μm, BD]. Cell suspension was pelleted at 1000 rpm for 5 min. Cells were resuspended in PPM and plated on an uncoated plate for differential plating. Cell suspension (nonadherent) was centrifuged at 1000 rpm for 5 min, and the pellet was resuspended in growth medium [GM] [Ham's F-10 medium supplemented with 20% FBS and 2.5 μg/mL bFGF]. Satellite cells were plated on collagen-coated flasks for expansion. Cells were fed daily with GM. For differentiation, plates were washed with PBS, refed with 2% horse-serum/DMEM differentiation medium, and refed daily.

**Adenoviral infection**

The adenoviral expression vector for PPARβ shRNA was a generous gift from Dr. Zhidan Wu [Novartis]. Primary muscle cells were infected with an adenovirus overexpressing GFP shRNA or PPARβ shRNA as previously described [Kleiner et al. 2009] and harvested 72 h post-infection. Cells were treated with DMSO, GW501516 [0.5 μM], AICAR [1 mM], or GW + AICAR for 24 h prior to harvest.

**RNAi experiments**

siRNAs (ON-TARGET plus SMARTpool, Dharmacon) targeting mouse Ldhα and Mef2a were transfected into primary myoblasts
at a final concentration of 20 nM using HiPerFect transfection reagent (Qiagen) according to the manufacturer’s instructions. Cells were then differentiated for 3 d prior to harvest.

**Oxygen consumption measurements**

Cellular OCRs were measured using the XF24 analyzer (Seahorse Bioscience, Inc.) per the manufacturer’s protocol. The basal OCR was measured in XF Assay medium supplemented with or without 10 mM sodium pyruvate [as indicated], following administration of 2 μM oligomycin (to inhibit ATP synthase) or the addition of the uncoupler FCCP [2 μM]. Immediately after measurement, total protein levels were measured with the Micro BCA protein assay kit (Thermo Scientific) for data correction.

**Cell transfection and luciferase reporter assays**

pCMX, pCMX-PPARβ, pBOS, and pBOS-PPARs vectors have been described previously (Robinson et al. 1998; Burkart et al. 2007). [MEF2a]tk-Luc, pG5Luc (Zhu and Gulick 2004), pCMX-Gal4-MEF2A (Zhu and Gulick 2004), and pCDNA3.1-Flag-MEF2A were generously provided by Dr. Tod Gulick (Sanford-Burnham Medical Research Institute). pCMV-myc-hAMPKα2 was kindly provided by Dr. Ronald Evans (Salk Institute). HEK293 cells were cultured at 37°C and 5% CO2 in DMEM supplemented with 10% FBS. Transient transfections in HEK293 cells were performed using FuGENE6 (Roche) as per the manufacturer’s protocol. Briefly, 600 ng of reporter was cotransfected with 100 ng of negative control luciferase vectors and 25 ng of CMV promoter-driven Renilla luciferase to control for transfection efficiency. Forty-eight hours after cotransfection, luciferase assay was performed using Dual-Glo (Promega) according to the manufacturer’s recommendations. All transfection data are presented as the mean ± standard error of the mean (SEM) for at least three separate transfection experiments done in triplicate.

**Immunoprecipitation**

Whole gastrocnemius muscle lysates from MCK-PPAR mice or lysate from HEK293 cells 48 h post-transfection were used for co-immunoprecipitation studies. HEK293 cells were collected and lysed. For AMPK ChIPs, myotubes were first fixed (power 1, 50% duty, 15 bursts). One microgram of M2 anti-Flag (Sigma) or anti-MYC (Millipore) antibodies were incubated with extract and protein G-conjugated agarose beads, and the immunoprecipitated proteins were analyzed by immunoblotting.

**ChIP assays**

ChIP assays were performed as previously described (Wende et al. 2005; Yang et al. 2009). Briefly, primary myotubes were cross-linked with 1% formaldehyde [10 min], and cells were collected and lysed. For AMPK ChIPs, myotubes were first fixed for 30 min with 0.5 mM ethylene glycol-bis(succinimidylsuccinate) (EGS) [Pierce]. Chromatin fragmentation was performed by sonication using a Bioruptor (Diagenode). Proteins were immunoprecipitated by using anti-PPARβ [K-20, Santa Cruz Biotechnology], anti-MEF2 [H-300, Santa Cruz Biotechnology], anti-AMPKα2 [R&D Systems], or IgG control [Sigma]. Following reversal of cross-linking, DNA was isolated [QIAquick PCR purification kit, Qiagen]. qPCR products were assessed and measured using the Stratagene MX3005P detection system. Quantitative analysis was performed by the standard curve method and normalized to IgG control. Specific oligonucleotide primers for target regions are listed in Supplemental Table 3.

**Statistical analyses**

Data were analyzed by Student’s t-test (two-tailed) or one-way ANOVA coupled to a Fisher’s least-significant difference (LSD) post-hoc test when more than two groups were being compared. Data represent the mean ± SEM, with a statistically significant difference defined as a value of P < 0.05.

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The nuclear receptor PPARβ/δ programs muscle glucose metabolism in cooperation with AMPK and MEF2

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