Cyclin D1 Represses Gluconeogenesis via Inhibition of the Transcriptional Coactivator PGC1α

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Hepatic gluconeogenesis is crucial to maintain normal blood glucose during periods of nutrient deprivation. Gluconeogenesis is controlled at multiple levels by a variety of signal transduction and transcriptional pathways. However, dysregulation of these pathways leads to hyperglycemia and type 2 diabetes. While the effects of various signaling pathways on gluconeogenesis are well established, the downstream signaling events repressing gluconeogenic gene expression are not as well understood. The cell-cycle regulator cyclin D1 is expressed in the liver, despite the liver being a quiescent tissue. The most well-studied function of cyclin D1 is activation of cyclin-dependent kinase 4 (CDK4), promoting progression of the cell cycle. We show here a novel role for cyclin D1 as a regulator of gluconeogenic and oxidative phosphorylation (OxPhos) gene expression. In mice, fasting decreases liver cyclin D1 expression, while refeeding induces cyclin D1 expression. Inhibition of CDK4 enhances the gluconeogenic gene expression, whereas cyclin D1–mediated activation of CDK4 represses the gluconeogenic gene-expression program in vitro and in vivo. Importantly, we show that cyclin D1 represses gluconeogenesis and OxPhos in part via inhibition of peroxisome proliferator–activated receptor γ coactivator-1α (PGC1α) activity in a CDK4-dependent manner. Indeed, we demonstrate that PGC1α is a novel cyclin D1/CDK4 substrate. These studies reveal a novel role for cyclin D1 on metabolism via PGC1α and reveal a potential link between cell-cycle regulation and metabolic control of glucose homeostasis.

Hepatic gluconeogenesis is crucial for the body to maintain normal blood glucose levels during fasting or extended periods of nutrient deprivation. Gluconeogenesis can also be maladaptive, contributing to the hyperglycemia observed in type 2 diabetes. Therefore, understanding how gluconeogenesis is controlled has been intensely studied because of its role in hyperglycemia and type 2 diabetes.

Peroxisome proliferator–activated receptor γ coactivator-1 (PGC1) is a family of multifunctional transcriptional coactivators that have emerged as playing a central role in cellular and systemic metabolism (1,2). PGC1α is the founding member of this family and was initially shown to be a central regulator of brown fat thermogenesis and mitochondrial biogenesis (3,4). Subsequent studies showed that PGC1α plays a critical role in the regulation of gene-expression programs, driving oxidative phosphorylation (OxPhos) and hepatic gluconeogenesis (5–9). Given the ability of PGC1α to promote gluconeogenesis, identification of mechanisms that control PGC1α activity has received significant attention.
In the fed state, various growth factors and signal transduction pathways lead to repression of gluconeogenesis and increased glucose uptake into peripheral tissues (10,11). Although the initial signaling events of these growth factors in the liver have been well studied, many of the downstream events are not clear (12). Activation of signaling pathways by growth factors activate the D-type cyclins, of which cyclin D1 is the best studied. The classic function of cyclin D1 is regulation of the cell cycle (13,14). Cyclin D1 promotes the G1 to S phase transition of the cell cycle by binding and activating Cdk4 or Cdk6. However, cyclin D1 affects other cellular processes. Cyclin D1 plays a role in both activation and repression of gene expression (14–20). Indeed, a recent article described the presence of cyclin D1 on hundreds of promoters throughout the genome (21). Other recent studies have pointed to a role for cyclin D1 in metabolism (22–24). Early studies as well as microarray analysis show that cyclin D1 is expressed in human and murine liver (25–27). Most studies on cyclin D1 in the liver are in the context of pathogenic conditions such as regeneration, carcinogenesis, and liver damage. However, most hepatocytes are quiescent/in G0 in the absence of chemical or physical damage. This raises the question as to the role of cyclin D1 in the liver under normal physiological conditions. Despite previous studies showing that cyclin D1 represses mitochondrial function and biogenesis in the livers of mice (23), to date, no studies have directly examined the effect of cyclin D1 on liver metabolism. We describe a new role for cyclin D1 as a repressor of PGC1α and, in particular, the ability of cyclin D1 to inhibit metabolic gene-expression programs induced by PGC1α.

RESULTS

Repression of Cyclin D1 Expression Correlates With Induction of a Program of Gluconeogenic Gene Expression

Previous studies and gene-expression analysis data show that cyclin D1 is expressed in a wide range of tissues, including the liver (26,27). We examined several tissues from mice for expression of cyclin D1 mRNA and protein. Cyclin D1 was expressed to varying degrees in a number of different tissues (Supplementary Fig. 1A and B). While spleen and intestine contained the highest levels of cyclin D1, almost no cyclin D1 was observed in pancreas or brain. The liver expressed abundant levels of cyclin D1 at both the protein and the RNA level. The presence of cyclin D1 in liver, despite the fact that hepatocytes, which comprise the bulk of the liver, are typically in G0, raises the question as to the normal physiological function of cyclin D1 in the liver.

Next we wanted to determine the role of cyclin D1 in a biologically relevant context. One of the functions of the liver is gluconeogenesis during fasting. Therefore, we examined a published microarray data set (National Center for Biotechnology Information Gene Expression Omnibus [NCBI GEO] accession GSE24504) for the expression of cyclin D1 in the liver during fasting, in which the gluconeogenic gene-expression program was induced (28). As the length of the fast progressed, cyclin D1 mRNA expression decreased (Fig. 1A). This prompted us to confirm and directly examine cyclin D1 expression, along with gluconeogenic genes induced during 6 and 24 h of fasting. As expected, glucose levels were reduced in fasted mice at 6 and 24 h (Supplementary Fig. 2A and B). Fasting significantly reduced the expression of liver cyclin D1 mRNA at both time points (Fig. 1B and Supplementary Fig. 2C). PEPCK is the rate-limiting step in gluconeogenesis that converts oxaloacetate into phosphoenolpyruvate. G6Pase controls the last step in gluconeogenesis, which converts glucose-6-phosphate to glucose. PGC1α induces the expression of PEPCK and G6Pase during fasting. The reduction in cyclin D1 mRNA during fasting correlated with a concomitant induction of mRNA for Pepck (and protein), G6pase, and Pgc1α at both 6 and 24 h (Fig. 1C and Supplementary Fig. 2D and E).

Previous studies demonstrate that loss of cyclin D1 led to an increased mitochondrial biogenesis. Since genes involved in OxPhos are induced during fasting/gluconeogenesis, we examined the expression of OxPhos genes and Pgc1α in liver RNA from cyclin D1 knockout mice (Cyclin d1−/−). Cytochrome C (Cyt-C), Cox4i, and Atpsynthase F1 (AtpsynF1) expressions were elevated in the livers of Cyclin d1−/− mice, demonstrating an increase in OxPhos gene expression in the absence of cyclin D1 (Fig. 1D–F). In addition, the expression of the Pgc1α was increased in livers from Cyclin d1−/− mice (Fig. 1G). As a whole, these studies suggest that cyclin D1 may be regulating metabolic function in the liver. These data prompted us to examine a published data set of livers from obese patients with and without diabetes (NCBI GEO accession GSE15653). PEPCK and G6Pase were significantly elevated in livers from diabetic patients compared with nondiabetic patients (Fig. 1H). In contrast, cyclin D1 levels were reduced in the livers of diabetic patients (Fig. 1I). This demonstrates a possible link between cyclin D1 and glucose homeostasis.

The gluconeogenic gene-expression program is repressed following refeeding. Therefore, we examined the effect of refeeding on cyclin D1 expression. While fasting reduced cyclin D1 expression (Fig. 1B), cyclin D1 expression was significantly induced following refeeding (Fig. 2A). This induction of cyclin D1 correlated with reduced Pepck, G6pase, Pgc1α, and OxPhos gene expression (Supplementary Fig. 3A–F).

The inverse correlation between gluconeogenic/OxPhos gene expression and cyclin D1 gene expression suggests a role for cyclin D1 in regulating the gluconeogenic and OxPhos gene-expression programs. Cyclin D1 functions primarily by activating the cyclin-dependent kinase 4 (CDK4). However, cyclin D1 also has CDK4-independent effects. Therefore, we used a pharmacological loss of function approach to determine the role of cyclin D1 on gluconeogenic and OxPhos gene expression and, in particular, whether these effects
are CDK4 dependent. For these studies, we used a potent and specific inhibitor of cyclin D-CDK4/6 kinases, PD0332991 (29). C57Bl/6J mice were fasted overnight, administered PD0332991 or PBS control, and refed for 4 h. After 4 h, glucose levels in refed control mice increased to a normal glucose concentration (198 mg/dL) (Fig. 2B). In fed mice treated with PD0332991, plasma glucose levels increased to 240 mg/dL, a level indicating hyperglycemia. While refeeding reduced the expression of gluconeogenic and OxPhos gene expression (Supplementary Fig. 3A–F), the presence of PD0332991, Pck1, G6pase, Pgc1α, and OxPhos gene expression were significantly higher (Fig. 2C–F). These data suggest that inhibition of cyclin D1/CDK4 via administration of PD0332991 either promotes gluconeogenic gene expression or prevents the repression of gluconeogenic gene expression, resulting in increased plasma glucose.

Next we used a gain-of-function approach to directly determine whether cyclin D1 is involved in the gluconeogenesis and OxPhos gene-expression programs. We used a transgenic Cyclin d1 mouse strain in which overexpression of cyclin D1 was targeted to the liver using the −596 to +21 fragment of the FABP1 promoter (Cyclin d1LiTG) (30). Cyclin d1LiTG mice were either fed or subjected to an overnight fast, and the expression of gluconeogenic genes Pck1, G6pase, and Pgc1α examined. Pck1 and G6pase were induced approximately fourfold and Pgc1α induced ~2.5-fold in the wild-type mice during fasting (Fig. 3A). However, in Cyclin d1LiTG mice, fasting-induced Pck1, G6pase, and Pgc1α gene expression was significantly reduced along with OxPhos gene expression (Supplementary Fig. 4A–C). Blood glucose levels were not altered in fed Cyclin d1LiTG mice. However, fasting glucose levels were reduced in Cyclin d1LiTG mice, underscoring a physiological

Figure 1—A reduction in cyclin D1 is associated with increased gluconeogenic and OxPhos gene expression. A: A published microarray data set of livers from fasted mice was reanalyzed for expression of cyclin D1 as described in RESEARCH DESIGN AND METHODS. Mice were fed or fasted overnight, livers were removed, and RT-PCR was performed for (B) Cyclin d1 or (C) G6pase, Pck1, and Pgc1α; n = 4–6 ± SD. OxPhos and PGC1α gene expression are increased in the absence of cyclin d1. RNA was isolated from the livers of Cyclin d1+/+ and Cyclin d1−/− mice. cDNA was synthesized and RT-PCR was performed for (D) Cyt-C, (E) Cox4i, (F) AtpsynF1, or (G) Pgc1α; n = 4 ± SD. Analysis of a published microarray data set from livers of obese nondiabetic and obese diabetic patients for (H) PEPCK, G6Pase, and (I) cyclin D1. *P < 0.05; **P < 0.0001; ***P < 0.01.
effect of the decrease in gluconeogenic gene expression (Fig. 3B). We also wanted to determine if acute expression of cyclin D1 affected the fasting-induced gluconeogenic response. Adenoviral-mediated expression of cyclin D1 in the liver of mice via tail vein injection significantly repressed fasting-induced Pepck, G6pase, and Pgc1α gene expression (Fig. 3C). Similar to the cyclin D1 liver transgenic mice, adenoviral cyclin D1 expression blunted fasting glucose (Fig. 3D).

The pharmacological and genetic data show that cyclin D1 represses gluconeogenic and OxPhos gene expression. This raises the question as to how cyclin D1 represses these programs of gene expression. PGC1α is a key regulator of gluconeogenesis and OxPhos. Therefore, we hypothesized that the reduction in gluconeogenic and OxPhos gene expression by cyclin D1 is a result of either cyclin D1–mediated reduction of PGC1α expression or PGC1α transcriptional activity. We examined whether cyclin D1 reduces the activity of PGC1α by using a full-length PGC1α cDNA fused to a heterologous Gal4 DNA-binding domain (Gal4-PGC1α). Gal4-PGC1α potently coactivated a UAS-TATA luciferase construct as expected (Fig. 4A). Increasing amounts of cyclin D1 repressed Gal4-PGC1α activity in a dose-dependent manner, demonstrating that cyclin D1 reduces the activity of PGC1α.

PGC1α promotes gluconeogenic gene expression by coactivating PEPCK and G6Pase (7). Therefore, we investigated whether cyclin D1 can repress PGC1α-mediated coactivation of PEPCK and G6Pase using promoter luciferase reporter constructs. As expected, PGC1α strongly coactivated PEPCK and G6Pase promoters (Fig. 4B and C). Cyclin D1 on its own had no effect activation of the PEPCK and G6Pase promoter activity in the absence of PGC1α. However, cyclin D1 reduced PGC1α-mediated activation of both promoters by at least 50%.

Although our experiments with the CDK4 inhibitor PD0332991 supports the role for CDK4 mediating the effects of cyclin D1, we wanted to further determine if CDK4 activity is responsible for the repression of PGC1α by cyclin D1. We carried out promoter luciferase studies using a mutant of cyclin D1 with a lysine to glutamic acid mutation at amino acid 112 (cyclin D1K112E) (31,32). This mutant can bind CDK4 and CDK6, but it is unable to activate their kinase activity. Wild-type cyclin D1 repressed PGC1α activation of the PEPCK and G6Pase promoters (Supplementary Fig. 5A and B). However, not only did the mutant cyclin D1 not reduce PGC1α-mediated activation of the PEPCK and G6Pase promoters, but it actually increased the activity of PGC1α. This is in line with the ability of the cyclin D1K112E mutant to reduce endogenous CDK4 activity and further suggests that CDK4 kinase activity is required for the activity of cyclin D1 to repress PGC1α.

Figure 2—Inhibition of cyclin D1/CDK4 promotes gluconeogenesis. A: Cyclin D1 is increased following refeeding of mice subjected to an overnight fast. B–H: Wild-type C57Bl/6J mice were fasted overnight then treated with vehicle control or 75 mg/kg PD0332991 for 4 h. B: Blood glucose was measured after 4 h. C–H: Mice were killed, livers were removed, RNA was isolated, and RT-PCR was performed as indicated. n = 4–6 ± SD. *P < 0.05.
The data above also show that cyclin D1 suppresses OxPhos gene expression. The ability of PGC1α to promote the expression of OxPhos genes is one of its most well-studied functions. Therefore, we wanted to determine whether cyclin D1 represses PGC1α transcriptional activity on the OxPhos gene promoters that it controls. We used the 5′ upstream promoter region of the cytochrome C (Cyt-C) gene linked to a luciferase reporter construct (33). PGC1α significantly increased Cyt-C promoter activity, whereas cyclin D1 on its own had little effect on the Cyt-C promoter (Fig. 4D). However, the ability of PGC1α to activate the Cyt-C promoter was reduced significantly in the presence of cyclin D1. Next we used a multimerized NRF1 response element (NRF1×4) to determine whether cyclin D1 affected the ability of PGC1α to coactivate endogenous NRF1 (34). NRF1 is coactivated by PGC1α and is one of the main transcription factors driving OxPhos gene expression (35). PGC1α alone increased NRF1×4 activity approximately threefold (Fig. 4E). Cyclin D1 by itself had little effect on the activity of the NRF1×4 luciferase construct, indicating that in this system, cyclin D1 is not directly suppressing NRF1 activity. In contrast, PGC1α-induced NRF1×4 luciferase activity was reduced by cyclin D1.

Several different cyclins regulate the cell cycle, so we wanted to determine whether the effect of cyclin D1 was specific for D-type cyclins or also other cell cycle–related cyclins. Cyclin E controls the entry of cells from G1 into S phase. Cells were transfected with the cytochrome C or NRF1×4 luciferase constructs along with PGC1α in the absence and presence of cyclin D1 or cyclin E. While cyclin D1 repressed PGC1α activity, cyclin E had no effect on PGC1α activity on either luciferase promoter (Supplementary Fig. 6A and B). These data demonstrate the specificity and ability of cyclin D1 to repress PGC1α activity.

Results from the cyclin D1 knockout studies above show that loss of cyclin D1 is associated with increased Pgc1α expression. PGC1α is part of an autoregulatory loop whereby PGC1α induces its own expression (36). We used a 10-Kb fragment of the PGC1α promoter linked to luciferase (PGC1α-10Kb luciferase) to examine the role of cyclin D1 on induction of PGC1α. Expression of PGC1α
cDNA increased the PGC1α promoter activity several fold (Fig. 4F), supporting the autoregulatory role of PGC1α. Cyclin D1 on its own had little effect on basal promoter activity. However, cyclin D1 significantly blocked the ability of PGC1α to activate its own promoter in a dose-dependent manner, which shows that cyclin D1 decreases PGC1α expression, in part, by inhibiting PGC1α activity.

The promoter luciferase data strongly suggest that effects of cyclin D1 are mediated in part via effects on PGC1α expression. PGC1α induced PEPCk in both cell lines (Fig. 5A and B). PD0332991 significantly enhanced the PGC1α-mediated induction of PEPCk and G6Pase in both cell lines. G6Pase was induced robustly by PGC1α in both cell lines. This induction was further potentiated by PD0332991, further suggesting that PGC1α is the target of cyclin D1/CDK4. We also examined whether inhibition of CDK4 by PD0332991 could increase PGC1α-mediated induction of OxPhos gene expression. PGC1α induced Cyt-c and AtpsynF1 in the liver-derived cancer cell lines (Supplementary Fig. 7A and B). In the presence of PD0332991, the induction by PGC1α was significantly increased.

Next we examined the effect of cyclin D1 on PGC1α activity in primary hepatocytes by ectopically expressing cyclin D1 and PGC1α adenovirus. Pepck and G6pase expression were induced robustly by PGC1α (Fig. 5C). The Pepck and G6pase induction were blunted 40 and 80% in the presence of cyclin D1, respectively. In order to confirm that the effect was mediated via CDK4, we examined the effect of cyclin D1 on PGC1α in the presence of the CDK4 inhibitor PD0332991. PD0332991 increased PGC1α-mediated induction of Pepck and G6pase ~50–60% (Fig. 5D). Moreover, repression of PGC1α by cyclin D1 was significantly reduced by PD0332991.

We also used Cdk4+/+ and Cdk4−/− mouse embryonic fibroblasts (MEFs) to further confirm the role of CDK4 since even specific inhibitors can have off-target effects. For these studies, we focused on OxPhos genes since MEFs express little to no PEPCk or G6Pase (Supplementary Fig. 8). Cells were infected with retrovirus expressing a control vector or PGC1α (Fig. 5E). PGC1α increased Cyt-c and AtpsynF1 ~25 and 50% in CDK4+/+ MEFs. While there was a trend toward increased Cox4i and Cox5b induction, it was not significant. However, in CDK4−/− MEFs, PGC1α significantly induced all four OxPhos genes (Fig. 5F–H). This demonstrates genetically that CDK4 is regulating the activity of PGC1α. Since MEFs do not express appreciable amounts of gluconeogenic genes, we also knocked
down CDK4 in Huh7 cells (Supplementary Fig. 9). While PGC1α induced PEPCK and G6Pase expression by itself, knockdown of CDK4 resulted in increased induction of PEPCK and G6Pase (Fig. 6E and F). These inhibitor and genetic loss of function data strongly suggest that CDK4 is, in part, responsible for the effects of cyclin D1 on PGC1α.

Next we wanted to determine whether cyclin D1/CDK4 are regulating gene expression via PGC1α in vivo. We again used the CDK4 inhibitor PD0332991 in PGC1α liver-specific knockout (Pgc1αLKO) mice. Pgc1αLKO and wild-type (Pgc1αWT) mice were fasted overnight and then administered vehicle control or PD0332991 by intraperitoneal injection and refed for 4 h. Previously (Fig.

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**Figure 5**—Pharmacological inhibition of CDK4 increases PGC1α transcriptional activity. A: HepG2 or B: Huh7 cells were infected with GFP control or PGC1α adenovirus and then treated with 1 μmol/L PD0332991 for 20 h. C: Primary hepatocytes were infected with GFP control, PGC1α, cyclin D1, or cyclin D1 and PGC1α adenovirus. D: Primary hepatocytes were infected with adenoviral PGC1α in the absence and presence of cyclin D1 and treated with PD0332991 for 20 h. Following treatment, RNA was isolated, cDNA was synthesized, and RT-PCR was performed for Pepck or G6pase. n = 3 ± SD. *P < 0.05; **P < 0.005.
2), we saw that inhibition of CDK4 in wild-type mice blocked the inhibitory effects of refeeding on gluconeogenic and OxPhos gene expression. In Pgc1aLKO mice, we did not observe a significant increase in Pepck or G6pase expression (Fig. 7A and B) (P > 0.05). Similarly, we did not observe an increase in OxPhos gene expression (Fig. 7C–E) (P > 0.05). These data demonstrate that the effects of cyclin D1 on gluconeogenic and OxPhos gene expression are, in large part, mediated via PGC1α.

Our in vitro and in vivo pharmacological and genetic loss of function data strongly suggest that cyclin D1/CDK4 mediate their effects through PGC1α. Although most kinases have multiple substrates, only a handful of substrates have been identified for CDK4 (37). Therefore, we carried out in CDK4/cyclin D1 in vitro kinase assays on purified full-length PGC1α. PGC1α was effectively phosphorylated by cyclin D1/CDK4 (Fig. 8A). We then performed in silico analysis to determine the potential phosphorylation sites on PGC1α. Two highly conserved sites were identified in the transcriptional repression domain, threonine 298 and serine 312 (Fig. 8B). Next we mutated the potential sites to alanine (PGC1αAA) and

![Figure 6](https://example.com/figure6.png)
conducted reporter luciferase experiments using the PEPCK, G6Pase, and Cyt-C luciferase promoters (Fig. 8C–E). As expected, cyclin D1 reduced the activity of PGC1α WT on all three promoters. However, cyclin D1 had little to no effect on the activity of the PGC1α AA mutant. These data identify PGC1α as a novel CDK4 substrate and further confirm that cyclin D1/CDK4 represses PGC1α.

**DISCUSSION**

Research on cyclin D1 has focused mainly on its ability to regulate the cell cycle and cancer. Cyclin D1 is expressed in the liver, a tissue that is relatively quiescent, raising the question as to the role of cyclin D1 in the liver. We describe here a novel metabolic role for cyclin D1, whereby cyclin D1 represses gluconeogenesis (Fig. 8F). Fasting reduces the expression of cyclin D1, leading to reduced CDK4 activity and hence increased PGC1α activity. By contrast, feeding induces cyclin D1, causing the activation of CDK4, phosphorylation of PGC1α, and repression of gluconeogenic gene expression.

PGC1α plays a key role in gluconeogenesis by coactivating various transcription factors, stimulating the expression of genes involved in gluconeogenesis such as PEPCK and G6Pase. The ability of PGC1α to induce gene-expression programs of OxPhos and gluconeogenesis is well established. It is metabolically advantageous for the liver to induce both of these programs of gene expression. The liver will use OxPhos to generate ATP so that it can generate glucose for the rest of the body via gluconeogenesis and maintain its own bioenergetics. The repression of gluconeogenesis and OxPhos gene expression by cyclin D1 in our studies was lost in mice lacking liver PGC1α, strongly suggesting that the repressive effect of cyclin D1 is via PGC1α. Therefore, our data demonstrate another potential mechanism whereby feeding induces cyclin D1, which leads to inactivation of PGC1α and its ability to promote gluconeogenesis and oxidative metabolism.

In addition to its effects on the cell cycle, cyclin D1 controls transcription by interacting with transcription factors, coactivators, and corepressors, promoting or repressing gene expression depending on the setting (14–20). The ability of cyclin D1 to regulate transcription was highlighted in a recent study that showed cyclin D1 bound to the promoters of hundreds of genes (21). A role for cyclin D1 in metabolism was suggested several years ago in studies showing that loss of cyclin D1 increased mitochondrial biogenesis (22,23). Furthermore a recent study showed that cyclin D1 inhibited liver lipogenesis, in part, by inhibiting HNF4 and ChREBP in a CDK4-dependent and CDK4-independent manner (24). Interestingly, in our studies, cyclin D1 repressed coactivation of HNF4 by PGC1α but failed to repress HNF4 on its own (Fig. 4B and C, column 3). We also showed that cyclin D1 does not repress gene expression or promoter activity controlled by other transcription factors, such as NRF1, in the absence of PGC1α. Therefore, PGC1α may be required for the repression of HNF4 observed in previous studies, or alternatively, the target of cyclin

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*Figure 7—Cyclin D1 mediates its effects on gene expression in vivo via PGC1α. Pgc1α liver-specific knockout mice were fasted overnight and then treated with vehicle control or 75 mg/kg PD0332991 for 4 h. Livers were harvested, RNA was isolated, and RT-PCR was performed as indicated for Pepck, G6pase, Cyt-c, Cox4i, AtpsynF1. n = 4–5 ± SD. NS, not statistically significant.*
D1-mediated repression may be both gene specific and cell-type dependent. Cell-cycle control by cyclin D1 is mostly dependent upon CDK4. However, its regulation of transcription has been shown to be both CDK4 dependent and independent. Our studies strongly suggest a CDK4-dependent effect. A mutant of cyclin D1 that can bind but fails to activate CDK4 did not repress PGC1α and actually...
increased its activity. This is most likely due to the fact that the mutant of cyclin D1 interacts with but does not activate CDK4, thereby reducing the available active CDK4 (31,32). Finally, only a handful of CDK4 substrates have been identified to date (13,37,38). Our data strongly suggest that PGC1α is a substrate for CDK4. In support of this analysis, recently, a screening assay suggested that PGC1α is a potential CDK4 substrate (37). Although we cannot completely rule out CDK4-independent effects, our data strongly suggest that CDK4 mediates the effects of cyclin D1 on PGC1α.

These studies have implications beyond the response to fasting and gluconeogenesis. The Warburg effect describes a metabolic alteration where cancer cells display increased glycolytic metabolism and reduced OxPhos with concomitant production of ATP and lactic acid even in the presence of oxygen. In recent years, this phenomenon has been shown to be a prominent feature in cancer. Several years ago, it was shown that PGC1α expression is reduced in different cancers, including the liver, compared with normal adjacent tissues (39). Since cyclin D1 overexpression represents an early lesion in cancer (40), it is tempting to speculate that downregulation of PGC1α activity by cyclin D1 is a potential mechanism responsible, in part, for the Warburg effect and the downregulation of OxPhos observed in cancer. Indeed, the cyclin D1 liver transgenic mice used in these studies develop hepatocellular carcinoma in ~15 months (30).

Cyclin D1 is most well-known for regulating the cell cycle, raising the question as to why cyclin D1 is expressed in a quiescent tissue like the liver. The ability of a cell-cycle regulator to regulate glucose homeostasis would be advantageous for a cell. Progression through the cell cycle and cell division requires abundant amounts of nutrients. In contrast, gluconeogenesis is a state of nutrient deprivation. Therefore, it makes sense that a molecule that controls cell-cycle progression would also play a role in glucose homeostasis. Therefore, cyclin D1 activation represents a mechanism that links the cell cycle with nutrient homeostasis and sensing via its interaction with PGC1α and explains the presence of a cell-cycle regulator in a quiescent tissue. Finally, our data show that livers from obese mice express less cyclin D1 mRNA than their nondiabetic counterparts, although caution should be taken given the procancer role of cyclin hence as a therapeutic target.

RESEARCH DESIGN AND METHODS

Cell Culture and Treatments
HepG2 and Cos7 cells were obtained from ATCC. HuH7 cells originated from the Japanese Collection of Research Bioresources. All cells were grown in DMEM supplemented with 10% FBS and penicillin/streptomycin. PD0332991 was obtained from Selleckchem. Cells were seeded into six-well plates and treated 24 h later as indicated in the figure legends.

Microarray Analysis of Published Data Set
Normalized data and probe annotations for GSE15653 and GSE24504 were downloaded from the NCBI GEO website. Data were used for reanalysis. Time points (0, 12, 24, and 48 h) were used as factor in one-way ANOVA analysis, and significant probes were selected by false discovery rate ≤0.1.

Animal Experiments
For all experiments, male mice were used between 8–12 weeks of age. Wild-type C57BL/6J, cyclin D1 liver-specific transgenic (Cyclin d1LKO), PGC1α floxed, and albumin Cre mice were obtained from The Jackson Laboratory and previously described (30). Cyclin d1-/- and Cyclin d1LKO mice have been described previously (41,42). Mice flanked by LoxP sites at the PGC1α allele (PGC1αB/R) were bred to Alb-Cre mice to generate liver-specific PGC1α knockout mice (Pgc1αLKO). We did not observe a difference in weight of cyclin D1Tg or PGC1 liver knockout mice compared with control mice at these time points, similar to previous studies (30,43). For fasting experiments, mice were either fed or fasted for 6 h or overnight (16 h) as previously described (44). For refeeding experiments, mice were refed for 4 h. For experiments with PD0332991, mice were administered 75 mg/kg of PD0332991 or vehicle following overnight fasting and then refed for 4 h. Following the described time points, mice were killed, and the livers removed for RNA analysis. Blood glucose was measured following fasting, as well as following refeeding in control and PD0332991-treated mice as previously described (44). Cdk4LKO and Cdk4LKO MEFs were generated by the Scicinski Laboratory (45). In brief, Cdk4LKO and Cdk4LKO day 13.5 embryos were harvested, internal organs were removed, and remaining tissues were minced and digested. Tissue was resuspended by pipetting in DMEM supplemented with 10% FBS and plated out in T75 flasks. Once the cells reached confluence, they were trypsinized and passaged at 1:4. Cells were cultured in DMEM plus 10% FBS. For most of the experiments, cells from passages 1 to 2 were used.

RNA Isolation and Measurement of Gene Expression
RNA from cells and tissue was isolated by using TRIzol reagent (Invitrogen), and RNA was reverse transcribed as previously described (46). RT-PCR was performed using human- or mouse-specific primers (Supplementary Table 1).

Transfections
Gal4-PGC1α, UAS-TATA luciferase, G6Pase luciferase, PEPCK luciferase, PGC1α-10Kb promoter luciferase, Cyt-C luciferase, NRF1 × 4 luciferase, PGC1α, cyclin D1, and cyclin D1LKO have been described previously and were obtained from Addgene (Cambridge, MA) (33,34,36,44). pRL-SV40 renilla luciferase was used as a transfection control (Promega). Cos7 cells were transfected with DNA as indicated in the figure legends by using SuperFect or Attractene for 3 h. Cell extracts were prepared and
luciferase activity was measured as previously described (47). Mutation of T298 and S312 of PGC1α was accomplished using site-directed mutagenesis. The resulting product was sequenced for fidelity and subcloned into pcDNA3.1.

Adenovirus Experiments
GFP and PGC1α adenovirus were prepared as previously described (47). HepG2, Huh7, Cdk4+/– and Cdk4–/– MEFs were infected with GFP- or PGC1α-expressing adenovirus. Media were changed after 6 h. For MEFs, RNA was harvested as described above 24 h later, and RT-PCR was performed as indicated. HepG2 and Huh7 cells were treated with 1 μmol/L PD0332991 overnight, RNA was isolated, and RT-PCR was performed.

Primary Hepatocyte Isolation
Mouse primary hepatocytes were prepared by perfusing C57Bl/6J mice with liver digest medium (Invitrogen) followed by filtration through a 70 μm mesh and Percoll gradient centrifugation. Cells were seeded at a density of 4 × 10^5 cells/well in six-well plates in DMEM containing 10% FBS, 2 mmol/L sodium pyruvate, 2% penicillin/streptomycin, 1 μmol/L dexamethasone, and 0.1 μmol/L insulin. After 4 h, cells were switched to 0.2% BSA, 2 mmol/L sodium pyruvate, 2% penicillin/streptomycin, 0.1 μmol/L dexamethasone, and 1 nmol/L insulin (maintenance medium). Cells were infected with adenovirus as indicated and RNA harvested for gene expression.

In Vitro Kinase Reactions
Glutathione S-transferase (GST) substrate proteins were expressed in Escherichia coli and purified as previously described (37). Briefly, GST fusion proteins were eluted from the glutathione resin in 100 mmol/L HEPES, 100 mmol/L NaCl, 0.2% Tween 20, 20 mmol/L glutathione, and 2.5 mmol/L 1,4-dithiotreitol. Then 10 μL of the eluate was mixed with 10 μL of complementation buffer (100 mmol/L HEPES, 20 mmol/L MgCl2, 2 mmol/L EGTA; pH 7.4). Samples were supplemented with 10 μCi γ-32P-ATP (PerkinElmer, Waltham, MA) and 40 μmol/L ATP followed by addition of 0.5 μL recombinant cyclin D1-CDK4 (ProQnase, Freiburg, Germany). Reactions were run at 30°C for 20 min and terminated by adding 10 μL of Laemmli buffer. Samples were loaded on SDS gels and transferred to nitrocellulose membranes, followed by staining with Amido black 10B protein stain. Stained membranes were dried and exposed to X-ray film.

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