Remodeling of energy metabolism and absence of electrophysiological changes in the heart of obese hyperleptinemic mice. New insights into the pleiotropic role of leptin

Rocio Guzmán-Ruiz1*, Nieves Gómez-Hurtado2, Marta Gil-Ortega1, Beatriz Somoza1, M. Carmen González3, Isabel Aránguez4, Miriam Martín-Ramos1, Carmen González-Martín1, Carmen Delgado1,5, Marisol Fernández-Alfonso6 and Mariano Ruiz-Gayo1*

1 Departamento de Ciencias Farmacéuticas y de la Salud, Facultad de Farmacia, Universidad CEU-San Pablo, Madrid, Spain
2 Departamento de Farmacología, Facultad de Medicina, Universidad Complutense, Madrid, Spain
3 Departamento de Fisiología, Facultad de Medicina, Universidad Autónoma, Madrid, Spain
4 Departamento de Bioquímica, Facultad de Farmacia, Universidad Complutense, Madrid, Spain
5 Centro de Investigaciones Biológicas, Consejo Superior de Investigaciones Científicas, Madrid, Spain
6 Instituto Pluridisciplinar-Departamento de Farmacología, Facultad de Farmacia, Universidad Complutense, Madrid, Spain

Dietary treatment with high-fat diets (HFD) triggers diabetes and hyperleptinemia, concomitantly with a partial state of leptin resistance that affects hepatic and adipose tissue but not the heart. In this context, characterized by widespread steatosis, cardiac lipid content remains unchanged. As previously reported, HFD-evoked hyperleptinemia could be a pivotal element contributing to increase fatty-acid (FA) metabolism in the heart and to prevent cardiac steatosis. This metabolic adaptation might theoretically reduce energy efficiency in cardiomyocytes and lead to cardiac electrophysiological remodeling. Therefore, the aim of the current study has been to investigate the impact of long-term HFD on cardiac metabolism and electrophysiological properties of the principal ionic currents responsible of the action potential duration in mouse cardiomyocytes. Male C57BL/6J mice were fed a control (10 kcal% from fat) or HFD (45 kcal% from fat) during 32 weeks. Quantification of enzymatic activities regulating mitochondrial uptake of pyruvate and FA showed an increase of both carnitine-palmitoyltransferase and citrate synthase activities together with a decrease of lactate dehydrogenase and pyruvate dehydrogenase activities. Increased expression of uncoupling protein-3, Mn-, and Cu/Zn-superoxide dismutases and catalase were also detected. Total glutathione/oxidized glutathione ratios were unaffected by HFD. These data suggest that HFD triggers adaptive mechanisms aimed at (i) facilitating FA catabolism, and (ii) preventing oxidative stress. All these changes did not affect the duration of action potentials in cardiomyocytes and only slightly modified electrocardiographic parameters.

Keywords: leptin, cardiac metabolism, UCP3, electrophysiology, obesity
and they can be ejected from mitochondria by means of the uncoupling protein-3 (UCP3), leading to a mitochondria-cytosol futile FA cycling (15). Otherwise, excessive FA oxidation has been shown to increase citrate production which can inhibit phosphofructokinase, a key enzyme recruiting fructose-6-phosphate into the glycolytic pathway, and also impair pyruvate oxidation (16–18). Finally, under these conditions, the rate of reactive oxygen species (ROS) production can be increased (19), and cause mitochondrial damage (20). In fact, optimizing energy metabolism by increasing glucose oxidation has been proposed to be a useful approach to prevent and treat cardiac dysfunction in obese people (15) as well as a valuable therapeutic intervention in failing hearts (3, 21, 22).

The aim of the current study was to further characterize the influence of long-term DIO on cardiac metabolism and the eventual association of metabolic remodeling and cardiac electrophysiological properties. For this purpose, we have analyzed in hearts of DIO mice: (i) enzymatic activities of lactate dehydrogenase, pyruvate dehydrogenase, carnitine-palmitoyltransferase (CPT), and citrate synthase (CS), as well as lactate content, (ii) mitochondrial uncoupling, (iii) redox status, (iv) electrophysiological properties of ionic currents responsible for the action potential duration (APD) in cardiomyocytes, and (v) electrocardiographic characteristics.

MATERIALS AND METHODS

EXPERIMENTAL DESIGN
Five-week old male C57BL/6J mice (Harlan, Spain) were housed (five per cage) under a 12 h light/12 h dark cycle, in a temperature-controlled room (22°C) with food and water ad libitum. Animals were divided in two groups with similar average BW and assigned either to a control or to a high-fat diet (HFD). Control diet (D12450B, 10 kcal% fat, 70 kcal% carbohydrates, and 20 kcal% protein; 3.85 kcal/g) and HFD (D12451, 45 kcal% fat, 35 kcal% carbohydrates, and 20 kcal% protein; 4.73 kcal/g) were supplied by Test Diet Limited BCM IPS Ltd (UK). After 32 week dietary treatment, groups of animals were separated for biochemical and functional studies. The investigation conforms to the Guide for the Care and Use of Laboratory Animals published by the US National Institute of Health (NIH publication No. 85–23, revised 1996). This study was approved by the ethics committee of the University CEU-San Pablo (SAF 2009-09714).

PLASMA BIOCHEMISTRY
Animals were killed by decapitation between 9 and 10 a.m., blood collected in chilled EDTA-coated polypropylene tubes and left ventricle dissected and frozen. Plasma samples were stored at −80°C until assay. For enzymatic activities, tissues were immediately processed and prepared for assay. Proteins, lactate, and glutathione were determined in frozen tissues. Plasma leptin (Linco Research, USA; 4.9% intra-assay variation, 3.3% inter-assay variation) and insulin (Mercodia, Denmark; 2.2% intra-assay variation, 4.9% inter-assay variation) were quantified by specific radioimmunoassay and ELISA, respectively.

CARDIAC TRIGLYCERIDES
Triglyceride content in heart was determined as previously described (8). Briefly, 20 mg of wet tissue were homogenized in a mixture containing 40 ml of 2 mM NaCl/20 mM EDTA/50 mM sodium phosphate buffer (pH 7.4) 40 ml of tert-butanol, and 20 ml of Triton X-100/methanol (1/1). Triglycerides were measured with a Sigma diagnostic kit (USA).

QUANTIFICATION OF PROTEINS BY WESTERN-BLOT
We quantified uncoupling protein-1, 2, and 3 (UCP1, UCP2, UCP3, respectively) Mn- and Cu/Zn-superoxide dismutase (Mn-SOD, Cu/Zn-SOD), and catalase. Western-blotting was performed as previously described (8, 9). Briefly, primary antibodies against UCP1, UCP3 (Affinity BioReagent, USA), UCP2 (Santa Cruz Biotechnology, USA), and Mn-SOD, Zn-SOD, and catalase (Sigma, USA) were applied overnight at 4°C at the appropriate dilution. After washing and incubation with appropriate IgG-peroxidase complexes, blots were incubated in commercial enhanced chemiluminescence reagents (ECL, Amersham Bioscence, UK) and exposed to autoradiographic film. Films were scanned using a GS-800 Calibrated Densitometer (Bio-Rad, Spain) and quantified using Quantity One software (Bio-Rad, Spain). Values were normalized with β-actin to account for variations in gel loading.

DETERMINATION OF ENZYMATIC ACTIVITIES
Lactate dehydrogenase, CPT, CS, and pyruvate dehydrogenase activities were measured by means of colorimetric methods as described in the literature (8, 9, 23, 24). Glucose-6-phosphate dehydrogenase (G6PD) activity was determined by measuring Δ absorbance at 340 nm (Versamax, Molecular Devices, USA) due to the conversion of NADP+ to NADPH by G6PD and 6-phosphogluconate-dehydrogenase (6PGD). For total activity (G6PD and 6PGD), samples (1 mg/ml protein) were incubated in 96-well microplates containing assay buffer (50 mM Tris pH = 8.1, 1 mM MgCl2, 0.2 mM glucose–6-phosphate, 0.2 mM 6-phosphogluconate, and 0.1 mM NADP+). For 6PGD activity determination, samples were incubated in absence of glucose–6-phosphate and then G6PD activity theoretically estimated by subtracting 6PGD activity from total activity. Results were expressed as Δ Abs/min/mg protein (25).

DETERMINATION OF SUPEROXIDE ANION, GLUTATHIONE AND LACTATE
Production of superoxide anion (O2−) was determined in fresh samples by measuring lucigenin-enhanced chemiluminescence (26, 27). Briefly, heart explants were incubated for 30 min at 37°C in oxygenated 20 mM HEPES buffer (pH = 7.4, 99 mM NaCl, 4.7 mM KCl, 1.9 mM CaCl2, 25 mM NaHCO3, 1 mM KH2PO4, 1.2 mM MgSO4, 11 mM glucose). Explants, NADPH (1 mM/l) and lucigenin (5 µM/l) were then added to tubes containing 1 ml buffer. O2− production was expressed in arbitrary units/mg protein. Lactate concentration was measured as described (8). For glutamate assay, samples were homogenized in phosphate buffer (pH = 6–7) containing EDTA (1 mM) and centrifuged for 15 min (10,000 g; 4°C). Supernatants were collected and kept at −20°C until assay. Both reduced (GSH) and oxidized glutathione (GSSG) concentrations were quantified by colorimetric assay by using a commercial kit (Cayman Chemical, USA) following the manufacturer instructions. Data were expressed as (total glutathione)/(GSSG).
**ISOLATION OF VENTRICULAR MYOCYTES**

Mice were heparinized (1 IU/kg i.p.) and anesthetized with pentobarbital (30 mg/kg). Hearts were rapidly dissected, mounted on a modified Langendorff apparatus and retrogradely perfused through the aorta (2–3 min, 36–37°C) with Ca\(^{2+}\)-free Tyrode solution (TS; 130 mM NaCl, 5.4 mM KCl, 0.4 mM Na\(_2\)PO\(_4\), 0.5 mM MgCl\(_2\), 25 mM HEPES, 5 mM NaHCO\(_3\), 22 mM glucose; pH adjusted to 7.4 with NaOH) containing 0.2 mM EGTA, then 3–4 min with TS containing 251 IU/ml type II collagenase (Worthington, USA), and 0.1 mM CaCl\(_2\). After perfusion, hearts were chopped in small pieces, and gently stirred in TS containing 1 mg/ml bovine serum albumin (BSA, Sigma, USA). Isolated cells were filtered, centrifuged and suspended in TS containing 2 mg/ml BSA and 0.5 mM CaCl\(_2\). After centrifugation, cells were suspended in a solution containing 1 mM CaCl\(_2\) and 2 mg/ml BSA and stored at room temperature until electrophysiological assays (within 5 h of the isolation).

**CELLULAR ELECTROPHYSIOLOGY**

Electrophysiological experiments were performed at room temperature (24–26°C) on Ca\(^{2+}\)-tolerant rod-shaped myocytes. Whole-cell configuration of the patch-clamp technique was employed to measure the L-type Ca\(^{2+}\) current (\(I_{\text{CaL}}\)), the transient outward K\(^{+}\) current (\(I_{\text{to}}\)), the ultra-rapid delayed rectifier K\(^{+}\) current (\(I_{\text{Kur}}\)), and the steady-state outward K\(^{+}\) current (\(I_{\text{Ks}}\)). The voltage-clamp circuit was provided by an Axopatch-200B amplifier controlled by a computer equipped with a pClamp 6.0 and interfaced to the amplifier with a Digidata 1322A (Axon Instruments, USA). Recording pipettes were made from 1.5 mm-OD soft-glass capillary tubing by using a microprocessor-based patch-clamp puller (P97/PC, Sutter Instruments, USA). Tip resistances after filling with the internal solution ranged between 0.9–2 MΩ. Whole-cell currents were expressed as current densities, which were calculated from the current amplitude normalized by the membrane capacitance. Membrane capacitance (\(C_m\)) was calculated according to the equation \(C_m = \tau_c I_0/\Delta E_m\) \([1 -(I_{\infty}/I_0)]\) (\(\tau_c\), time constant of the membrane capacitance; \(I_0\) maximum capacitance current value; \(\Delta E_m\), amplitude of the voltage step; \(I_{\infty}\), amplitude of the steady-state current). \(C_m\) was elicited by applying ±10 mV voltage steps from a holding potential of −60 mV. Current-voltage (\(I-V\)) relationship for the \(I_{\text{CaL}}\) was obtained from a holding potential of −50 mV and cell depolarization (300 ms, 0.2 Hz) in 10 mV increments from −40 to +60 mV. Current-voltage (\(I-V\)) relationship for \(I_{\text{to}}\) was constructed from 500 ms series test potentials varying from −70 to +50 mV in 10 mV increments, from a holding potential of −80 mV at a frequency rate of 0.1 Hz. The same protocol was applied but now preceded by a 100 ms inactivating pre-pulse (−40 mV) in order to suppress the transient portion of the \(I_{\text{to}}\) (28). The remaining current (\(I_{\text{Klow}}\)) is composed of both a \(I_{\text{Kur}}\) (4-aminopyridine-sensitive component) and a \(I_{\text{Ks}}\) (4-aminopyridine-resistant component). For \(I_{\text{Ks}}\) recording, an inactivating pre-pulse was applied in presence of 250 μM 4-AP. \(I_{\text{Ks}}\) was calculated by subtracting \(I_{\text{Kur}}\) from the current recorded in the absence 4-AP. Current densities were determined at the peak current. Action potentials (AP) were elicited at 10 s intervals by 1.5-fold excitation threshold current pulses of 2.5 ms in duration. After stabilization of the records, 10 successive APs were recorded. The parameters of the APs for each cell corresponded to the mean of these 10 APs. The APD was measured at 20, 50, and 90% repolarization. The solution for \(I_{\text{CaL}}\) recording contained (in mM): 140 NaCl, 1.1 MgCl\(_2\), 5.4 CaCl\(_2\), 10 glucose, 5 HEPES, 1.8 CaCl\(_2\); pH adjusted to 7.4 with NaOH. The intracellular recording pipette solution for whole-cell experiments contained (in mM): 100 CsCl, 20 triethylamine, 5 EGTA, 10 HEPES, 5 Na\(_2\)ATP, 0.4 Na\(_2\)GTP, 5 Na\(_2\)creatine phosphate, 0.06 CaCl\(_2\); pH adjusted to 7.4 with CsOH. The solution for \(K^+\) current recordings contained (in mM): 135 NaCl, 10 glucose, 10 HEPES, 1 MgCl\(_2\), 1 CaCl\(_2\), 5.4 KCl, 1 CoCl\(_2\), 1 BaCl\(_2\) (25 μM 4-AP), pH adjusted to 7.4 with NaOH. The solution for AP recordings contained (in mM): 140 NaCl, 4 KCl, 1.1 MgCl\(_2\), 1.8 CaCl\(_2\), 10 glucose, 10 HEPES, pH adjusted to 7.4 with NaOH. The intracellular recording pipette solution for both Ito and AP contained (in mM): 135 KCl, 4 MgCl\(_2\), 5 EGTA, 10 HEPES, 10 glucose, 5 Na\(_2\)ATP, 3-phosphocreatine, pH adjusted to 7.2 with KOH.

**SURFACE ELECTROCARDIOGRAM RECORDINGS**

Mice were anesthetized with sodium pentobarbital (60 mg/kg). Thirty two-gauge needle steel electrodes were inserted subcutaneously, according to a standard three-lead ECG scheme. Acquisition was performed during 8–10 min following anesthesia and data collected for 8–15 consecutive min at a rate of 4 kHz by using a ML136 Animal Bio amplifier. Parameters for each individual animal were calculated from 40 to 45 consecutive traces (LabChart 47.0 Pro, ADInstruments, UK). QTc interval was calculated using a modified Bazzett’s correction formula \[\text{QTc} = \text{QT} / (\text{RR} / 100)^{1/2}\] (29).

**STATISTICS**

All parameters were analyzed by a one-way ANOVA, followed by Newman-Keuls post hoc test. Statistical significance was set at \(p < 0.05\).

**RESULTS**

**PLASMA BIOCHEMISTRY AND CARDIAC WEIGHT AND LIPID CONTENT**

As summarized in Table 1, HFD animals displayed a 40% increase BW, compared to control animals (\(p < 0.001\)). Heart weight was increased approx. about 7% in HFD mice (\(p < 0.05\)), although cardiac TGs were not different between control and HFD groups. Plasma insulin (\(p < 0.001\)) and leptin (\(p < 0.05\)) were also increased by HFD treatment.

**ENZYMATIC ACTIVITIES INVOLVED IN MITOCHONDRIAL UPTAKE OF FATTY-ACIDS AND PYRUVATE APPEAR TO BE UP- AND DOWN-REGULATED, RESPECTIVELY, IN HFD MICE**

We observed that both lactate dehydrogenase \([F_{(1,21)} = 5.717; p < 0.05; \text{Figure 1A}]\) and pyruvate dehydrogenase activities \([F_{(1,8)} = 12.479; p < 0.001; \text{Figure 1B}]\) were reduced in HFD mice. This suggests that the contribution of lactate and pyruvate to cardiac energetics would be reduced in HFD mice. Moreover, as previously reported (9), 1-way ANOVA revealed an increase of total CPT activity in HFD mice \([F_{(1,18)} = 15.417; p < 0.05; \text{Figure 1C}]\), suggesting that mitochondrial uptake of FA is up-regulated by HFD treatment.
Table 1 | Effect of 32-week HFD on body and cardiac weight, cardiac triglyceride content, and insulin and leptin plasma concentration

|                  | BW (g)  | Heart weight (mg) | Cardiac triglycerides (mg/g tissue) | Plasma insulin (µg/l) | Plasma leptin (ng/ml) |
|------------------|---------|-------------------|------------------------------------|----------------------|----------------------|
| Control          | 33.9±0.6| 160.4±3.1         | 33.5±3.6                           | 1.9±0.2              | 12.8±1.9             |
| HFD              | 47.1±1.1***| 175.2±6.1*        | 27.8±2.6                           | 6.0±0.7***           | 30.2±4.5*            |

*p < 0.05; ***p < 0.001 (Newman–Keuls’s test).

CITRATE SYNTHASE ACTIVITY AND MITOCHONDRIAL UNCOUPLING
The ability of cardiac mitochondria to incorporate acetyl-CoA to the tricarboxylic acid cycle can be estimated by measuring CS activity, which accounts for the condensation of acetyl-CoA and oxaloacetic acid to yield citric acid. Figure 1D illustrates the effect of HFD on CS activity, which was significantly increased in HFD mice [1-ANOVA F(1,10) = 5,349; p < 0.05]. This up-regulation corresponds to almost 30% increase of CS activity. We determined the amount of UCP1, UCP2 (data not shown), and UCP3 and only UCP3 (Figure 2) appeared to be up-regulated in HFD mice [1-ANOVA F(1,12) = 5,327; p < 0.05].

REDOX STATUS OF CARDIAC TISSUE AFTER HFD TREATMENT
Cu/Zn-SOD, Mn-SOD, and catalase were increased in the HFD group [1-ANOVA F(1,11) = 5.593, p < 0.05; 1-ANOVA F(1,10) = 5.017, p < 0.05; 1-ANOVA F(1,10) = 5.141, p < 0.05 respectively; see Figures 3A–C. (Total glutathione)/(Oxidized glutathione) ratios were not different between control (7.69 ± 1.21) and HFD animals (7.76 ± 1.69). O$_2^-$ production appeared to be slightly increased (p = 0.08) in HFD mice (Figure 3D). Finally, we observed a reduction of glucose-6-phosphate dehydrogenase activity (Figure 3E). This enzyme is a main source of NADPH, a co-factor necessary for maintaining redox equilibrium.

CELLULAR ELECTROPHYSIOLOGY
To analyze the influence of HFD in the cellular electrophysiological properties of ventricular myocytes, patch-clamp experiments were carried-out. Figure 4A shows that mean APD values, measured at 20, 50, and 90% repolarization, were identical in control and HFD ventricular myocytes. Figure 4B shows current density-voltage relationship for I_{Ca,L} obtained in myocytes isolated from control and HFD mice. The maximum
value of $I_{CaL}$ density was $-7.8 \pm 0.7 \text{pA/pF}$ at $0 \text{mV}$ ($n=10$) for control and $-7.0 \pm 0.7 \text{pA/pF}$ at $0 \text{mV}$ ($n=9$) for HFD mice. Figure 4C illustrates current density-voltage relationships for $K^+$ currents ($I_{\text{tot}_{\text{total}}}$, $I_{\text{kur}}$, and $I_{\text{ss}}$). Mean density values and voltage-dependence were similar between control and HFD myocytes. $I_{\text{tot}_{\text{total}}}$ at $+60 \text{mV}$ was $23.4 \pm 4.6 \text{pA/pF}$ ($n=10$) for control and $22.0 \pm 3.1 \text{pA/pF}$ ($n=9$) for HFD. $I_{\text{kur}}$ at $+60 \text{mV}$ was $15.1 \pm 4.2 \text{pA/pF}$ ($n=9$) for control and $15.3 \pm 3.6 \text{pA/pF}$ ($n=7$) for HFD. $I_{\text{ss}}$ at $+60 \text{mV}$ was $10.5 \pm 0.9 \text{pA/pF}$ ($n=9$) for control and $10.1 \pm 0.9$ ($n=7$) for HFD. Taken together these data indicate that HFD did not induce changes in the cellular electrophysiological properties of ventricular myocytes.

**DISCUSSION**

This study shows that long-term HFD activates metabolic pathways in cardiac tissue that would (i) promote $\beta$-oxidation to the detriment of carbohydrate catabolism, and (ii) prevent cardiac steatosis. This is striking inasmuch as under our experimental conditions both adipose and hepatic tissues undergo pathological alterations of energy metabolism leading to obesity and hepatic steatosis. The interest of our animal model (long-term treatment with a relatively mild, 45% kcal from fat, HFD from the early adolescence) deals with the fact that HFD mice display diabetes,
obesity, and hyperleptinemia together with a generalized state of leptin resistance that does not involve cardiac tissue (10). This is a very relevant issue because the above mentioned study evidenced that ectopic deposition of lipids in the liver of obese mice is coincident with the development of leptin resistance in this organ. In fact, many authors have reported that HFD causes cardiac steatosis in genetically engineered models of obesity dealing with either generalized leptin resistance or aleptinemia (15, 30–32). Thus, the current data would further support the concept that a particular responsiveness of cardiac tissue to hyperleptinemia might account for the adaptive metabolic remodeling triggered by HFD. Otherwise, although a systematic analysis of heart morphology has not been carried-out, cardiac hypertrophy seems to be absent (cardiac weight appears to be only slightly increased in HFD mice, but appears to be decreased when comparison is made between cardiac weight/BW ratios). Moreover, we have observed that cardiac electrophysiology remains mostly unaffected suggesting that 45% HFD treatment are less detrimental for the heart than for other tissues such as the liver (10) or, even, the central nervous system (33, 34).

In our previous study we had characterized the influence of HFD on the kinetic characteristics of cardiac CPT and we had found that DIO reduces the inhibitory effect of malony-CoA on CPT activity by a mechanism involving the Akt signaling pathway (9). This former study strongly supports the concept that cardiac metabolism adapts to HFD by increasing mitochondrial uptake and further oxidation of FA. In the current study we have evaluated enzymatic activities involved in glucose metabolism and we have identified a decrease of both lactate dehydrogenase and pyruvate dehydrogenase activities together with a reduction of lactate concentration that suggests a low glycolytic rate as well as a small contribution of pyruvate to mitochondrial energy supply. In addition, the elevated CS activity detected in HFD mice suggests an increased rate of β-oxidation (18). Moreover, increased citrate production would be coherent with an elevated rate of pyruvate to mitochondrial energy supply. In addition, the elevated CS activity detected in HFD mice suggests an increased rate of β-oxidation (18). Otherwise, the activity of glucose-6-phosphate dehydrogenase appears to be reduced in HFD mice, suggesting that NADPH levels might be reduced by the dietary treatment. The
eventual consequences of a fall of NADPH levels are difficult to be
drawn as this co-factor is pivotal to maintain the pool of reduced
 glutathione for reductive process but it is also the substrate of
 NADPH-oxidase, a source of $\cdot \mathrm{O}_2^-$ and ROS production. In fact, $\cdot \mathrm{O}_2^-$
 levels appears to be slightly increased in HFD hearts while (total
 glutathione)/(oxidized glutathione) ratios appear to be unchanged
 by HFD. Otherwise we have detected an up-regulation of antiox-
didant enzymes. Taken together, all these data would indicate that
 cardiac tissue of HFD mice remain mostly unaffected in terms of
 oxidative stress.

Finally we have also explored the effect of HFD on the amount
of the uncoupling proteins UCP1, UCP2, and UCP3. In our study
UCP3 is the only UCP that appears to be over-expressed in HFD
mice. In a recent study Boudina et al. (35) have reported that
UCP3 is up-regulated by HFD. These authors propose that UCP3
could be a negative regulator of cardiac efficiency in using energy
derived from FA oxidation, although UCP3 lacks of effect on FA
oxidation rates. Otherwise, UCP3 might act as a proton leak and
might also facilitate the translocation of FA carboxylate anions
out of the mitochondrial matrix. Because carboxylates re-enter
the mitochondria as neutral (protonated) species (36), the up-
regulation of UCP3 might generate a futile cycling of FA that
would contribute to reduce ATP synthesis. In this context, our
results would suggest that HFD hearts might display poor energy
efficiency. In fact ATP levels appear to be decreased in HFD hearts
(0.25 ± 0.03 in HFD vs. 0.40 ± 0.05 nmol/mg protein in control
animals; $p < 0.05$; results not shown), although this data is of dif-
cult interpretation in absence of AMP and/or creatinphosphate
values. From our data we cannot draw the mechanism that account
for UCP3 up-regulation, but we hypothesize that hyperleptinemia
might play a role. Interestingly, the former study by Boudina et al.
(35) has evidenced that the energy-wasting function attributed to
UCP3 is not perceptible in aleptinemic mice, and leptin has been
shown to induce UCP3 in different tissues (37, 38). Our data sug-
gest that UCP3 up-regulation would be integral to the adaptive
response to HFD/hyperleptinemia, that would contribute to spare
ectopic lipid deposit even at expense of a loss of energy efficiency.

Although functional studies are necessary to characterize car-
diac function in DIO mice, our data suggest that HFD trigger an
efficient metabolic adaptation, able to avoid ectopic accumulation
of triglycerides and to limit lipotoxic damage (35), which include
the preservation of cardiac electrophysiological properties of the
major ionic currents, as already reported to occur in DIO rats (39).
K$^+$-current remodeling has been observed in metabolic cardiomy-
opathy induced by PPAR$\alpha$ overexpression, supporting the concept
that myocardial metabolism and cardiac function are closely linked
(40). The shortening of PR intervals detected in the current study is
compatible with the existence of accessory atrio-ventricular path-
ways and suggests that, in spite of a normal behavior of ventricular
K$^+$-channels, conduction abnormalities might be incipient. To our
knowledge, this is the first study showing that a dietary treatment
is able to evoke this kind of conduction alteration. Interestingly,
this kind of accessory pathways is present at birth in humans (41)
and normally disappears few weeks after birth. Nevertheless, they
appear to be preserved in adult hearts lacking normal AMPK activ-
ity (42). We have previously reported that the active form of AMPK
is less abundant in DIO than in control hearts (9) and we speculate
now that a HFD treatment from early adolescence might induce de
novo development of cardiac accessory pathways or, alternatively,
to delay the natural degeneration of perinatal atrio-ventricular

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**FIGURE 5 | Electrocardiogram recordings and interval values obtained in control and HFD mice.** Typical surface ECG recording obtained in control (A) and HFD mice (B). Intervals duration and heart rate appear summarized in table (C). *$p < 0.05$, compared to the control group. HR, heart rate.
connections. It has to be noted that ECG recordings were performed in anesthetized animals and, therefore, eventual effects of the anesthetics cannot be discarded.

In conclusion, all these results indicate that metabolic remodeling evoked by HFD appears to be insufficient to affect cardiomycyte electrical properties, but could trigger incipient functional disorders.

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